Hort Innovation

Final Report

RD&E program for control, eradication and preparedness for vegetable leafminer

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Delivery partner:

Cesar Australia, the University of Melbourne, Northern Australia Quarantine Strategy, Plant Health Australia, AUSVEG

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RD&E program for control, eradication and preparedness for vegetable leafminer MT16004

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Contents

Contents 3
Summary 4
Keywords 5
Introduction 6
Methodology 8
Outputs 13
Outcomes 15
Monitoring and evaluation 19
Recommendations 28
Refereed scientific publications 34
References 39
Intellectual property, commercialisation and confidentiality 41
Acknowledgements 42
Appendices 43

Summary

The Hort Innovation-funded project MT16004 – RD&E Program for control, eradication and preparedness for vegetable leafminer (2017-2020) was developed in recognition of the extensive impact that exotic *Liriomyza* spp. pests, including the vegetable leafminer (VLM: *Liriomyza sativae*), the serpentine leafminer (SLM: *Liriomyza huidobrensis*) and the American serpentine leafminer (ASLM: *Liriomyza trifolii*), could have on the vegetable, nursery, melon and potato industries were they to enter Australian production areas. Primary focus was placed on VLM, which has been present in mainland Far North Queensland since 2015, where phytosanitary measures are in place to prevent the further spread of the pest on mainland Australia.

Project partners included Cesar Australia, the University of Melbourne, Plant Health Australia, the Department of Agriculture, Water and the Environment's Northern Australia Quarantine Strategy (NAQS) and AUSVEG. The wide skillset of project partners was paramount to the project's success. Key stakeholders included Australian vegetable, nursery, melon, and potato growers, agronomists and consultants, supply chain, industry bodies and associations, researchers, state government, and federal government.

The project successfully achieved its major objectives, specifically to:

- Investigate biological and chemical control options for VLM, SLM, and ASLM;
- Develop management guidelines for VLM, SLM, and ASLM;
- Develop leafminer surveillance information and toolkits for industry and government;
- Update existing contingency plans for VLM, SLM, and ASLM to ensure industry preparedness;
- Complete incursion pathway analysis for the Torres Strait populations of VLM, using overseas VLM samples and next-generation molecular sequencing tools;
- Model the potential spread and establishment risk of VLM, SLM, and ASLM to inform control and extension efforts, and to validate incursion pathway analysis;
- Increase community awareness about VLM on Cape York Peninsula and the Torres Strait Islands;
- Develop information packages for industry that provide scientifically robust methods for monitoring, eradication, and management;
- Support postgraduate students to build knowledge regarding biocontrol agents within Australia; and
- Communicate findings and increase industry understanding/awareness of VLM, SLM, and ASLM.

Key outputs included the development of molecular diagnostic tests; emergency use permits; a review of chemical options; a review of biological control agents and mass rearing prospects; a Cape York Peninsula scoping study report; surveillance guidelines; contingency plans; predictive modelling tools; incursion pathway analysis; industry preparedness plans, pest management plans, C&E resources; and scientific publications. Moreover, outputs have included the completion of one postgraduate student's M. Phil. thesis investigating Australian parasitoids of agromyzids and how they might be leveraged for biocontrol, and will ultimately include the completion of a second postgraduate student's PhD thesis investigating leafminer and parasitoid genetics, including the potential to improve mass rearing prospects for parasitoids via endosymbiont bacteria.

Extension efforts have been broad ranging. The project team have engaged with vegetable, potato, melon and nursery growers across all horticultural producing states and territories in Australia, and a total of 19 workshops, eight webinars, three podcasts, and two videos were delivered. The project team have also engaged closely with state and federal governments to increase uptake and understanding of preparedness research activities.

Key outcomes include an improved understanding of the ecology of Australian VLM populations; the creation of technically justified and cost-effective surveillance and diagnostic techniques to support diverse surveillance goals; improved understanding of establishment, spread and impact potential of *Liriomyza* spp., resulting in improved awareness activities and regional preparedness; a list of biocontrol agents present in southeastern Australia with a focus on parasitoids; increased awareness of *Liriomyza* pests within the vegetable, nursery, potato and melon industries and within government; improved capacity for rapid industry response to incursions via ready to go resources including contingency plans, chemical permits, host lists and regionally focused preparedness plans; and increased awareness of VLM within the Seisia community of Cape York Peninsula.

The project has developed several recommendations, including research and extension needs deemed important should *Liriomyza* pests establish within production areas in Australia. In light of the recent incursion of SLM in NSW and QLD in November 2020, specific research priorities relevant to this incursion are also provided.

Keywords

Liriomyza; agromyzid; exotic pest; leafminer; vegetable; nursery; melon; potato; surveillance; parasitoid

Introduction

With a horticulture industry valued at $14.4 billion (in 2018/19) (Hort Innovation 2020), Australia prides itself on producing clean, green, and safe produce. However, biosecurity incursions pose a significant threat to the future of Australia's horticulture industries.

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. Nearly all agromyzid species are very host-specific but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world (Spencer 1973, 1990). This includes the vegetable leafminer (VLM: *Liriomyza sativae*), the serpentine leafminer (SLM: *Liriomyza huidobrensis*) and the American serpentine leafminer (ASLM: *Liriomyza trifolii*), all of which are pests of vegetable and ornamental crops and thus pose a significant biosecurity threat to Australia. As highly polyphagous insects capable of infesting a broad range of crops from a variety of horticultural industries, *Liriomyza* spp. represent a significant threat to the viability of Australia's vegetable (valued at $4.7b in 2018/19) (Hort Innovation 2020), nursery (valued at $2.4b in 2018/19) (Hort Innovation 2020), melon (valued at $181m in 2018/19) (Hort Innovation 2020), and potato (valued at $753 million in 2018/19) (Hort Innovation 2020) industries.

The majority of *Liriomyza* spp. damage occurs during larval feeding under the leaf surface, which curtails photosynthetic ability and reduces marketability of crops. *Liriomyza* spp. are also prone to evolving insecticide resistance, making control and eradication difficult. With an increasing focus on international export, current reliance on interstate trade, and increasing costs of production, keeping Australian vegetable, nursery, melon, and potato production areas free of trade sensitive plant pests is important.

Following the detection of VLM in the Torres Strait Islands (TSI) in 2008 and Papua New Guinea in 2011 (Blacket et al. 2015), it was recorded at Seisia, QLD, in the Northern Peninsula Area (NPA) in 2015 (International Plant Protection Convention 2017). Eradication of VLM from the TSI or NPA was not considered technically feasible, and phytosanitary measures are in place to prevent the further spread of the pest on mainland Australia (International Plant Protection Convention 2017).

The Hort Innovation-funded project MT16004 – RD&E Program for control, eradication and preparedness for vegetable leafminer (2017-2020) was developed to prepare industry for the arrival of VLM into production regions. A project variation made in 2019 allowed RD&E activities to extend to SLM and ASLM, with the purpose of further safeguarding Australia's horticulture industries (Figure 1). In the final month of the project, an incursion of SLM was detected in NSW and QLD, and eradication was subsequently deemed not technically feasible (AUSVEG 2020).

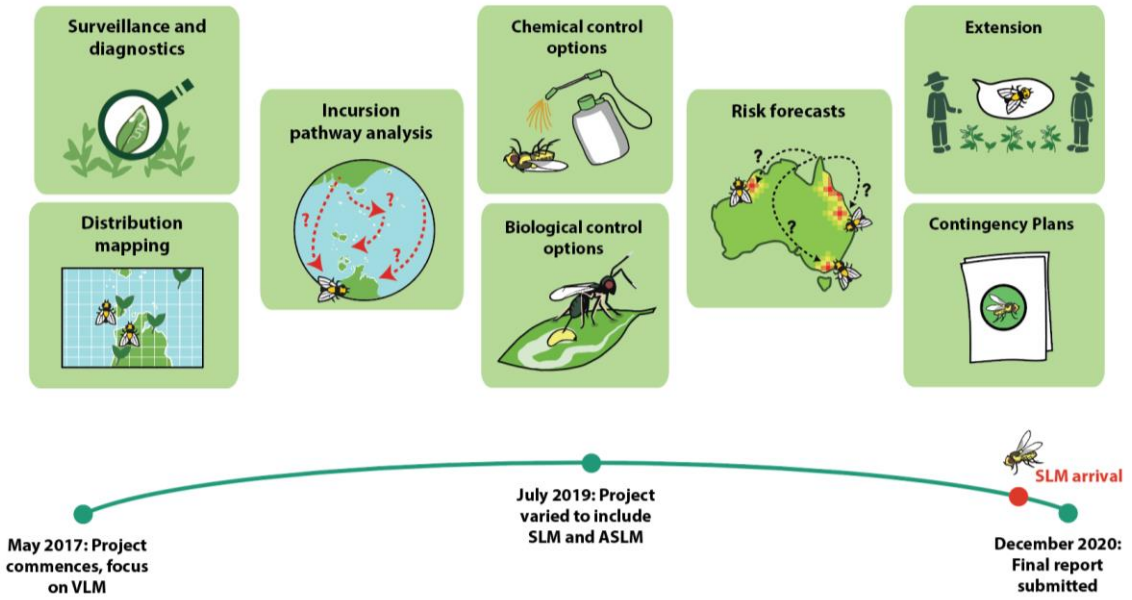


Figure 1. Timeline of project MT16004, including main focus areas and the recent incursion of SLM into NSW and QLD. Image credit: Dr. Elia Pirtle, Cesar Australia

This project brought together a strong team of project partners from Cesar Australia (project lead), Plant Health Australia, the University of Melbourne, the Department of Agriculture, Water and the Environment's Northern Australia Quarantine Strategy, and AUSVEG, with capabilities in pest surveillance, diagnostics, modelling, pest control, incursion response, and industry engagement. This mixture of capabilities was complementary in nature for the purposes of undertaking this project.

Between 2017 and 2020, robust scientific research provided by project partners resulted in the development of several outputs that strengthen industry's ability to prepare for *Liriomyza* spp. Intrinsic to the success of the project was the strong information flow between project partners as outputs were achieved, and these achievements are outlined in this report.

Methodology

To ensure comprehensive preparedness against *Liriomyza* spp., the project involved several components, including (1) the development of surveillance tools (including risk forecasting tools and pathway analyses); (2) the development of management advice (considering chemical and biological control options); (3) an extension and communication campaign; and (4) the development of contingency plans (Figure 2).

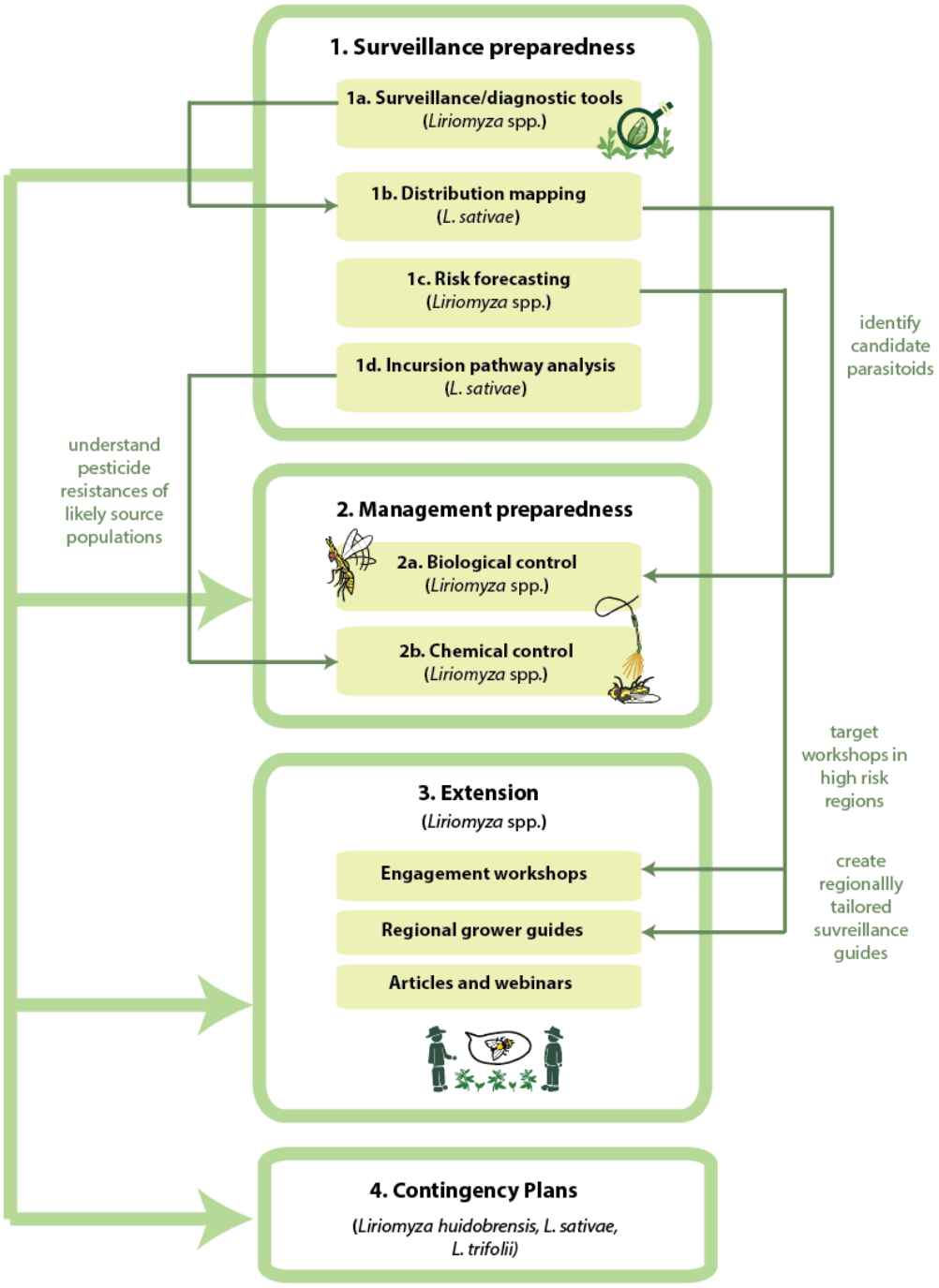


Figure 2. Major project components, highlighting examples of cross component synergisms. Image credit: Dr. Elia Pirtle, Cesar Australia

The management of these components required a holistic perspective to ensure potential synergisms were exploited. For example, modelling high risk incursion areas (component 1c) naturally fed into extension outcomes (component 3) by providing regional focus for these programs and thus increasing chances of pest detection and effective management. The diagnostic tools developed as part of component 1a were applied to surveillance activities (component 1b), allowing for rapid determination of geographic and host range expansions of VLM within the TSI and NPA. All research findings informed extension activities, allowing for the creation of regionally tailored preparedness guides (component 3), as well as informing the updated contingency plans (component 4).

The outcomes of this project were achieved through collaboration between researchers and extension staff with extensive expertise in: (i) leafminer biology and management, (ii) predictive modelling, (iii) pesticides and resistance, (iv) preparation of contingency plans and emergency permits to support biosecurity response, (v) quarantine and biosecurity fieldwork in north QLD and the TSI, (vi) and industry engagement. To ensure outcomes and outputs were relevant and provided value to industry and government, a project steering committee was established and included representatives from key project partners (including Assoc. Prof. Paul Umina, Dr. Peter Ridland, Dr. Elia Pirtle, Madeleine Quirk, Zarmeen Hassan, Dr. Jessica Lye and Lisa Brassington), plus John McDonald (National Biosecurity Manager of Greenlife Industry Australia) and Rebecca Sappupo (Manager, Policy (Incident Response & Preparedness), Plant Biosecurity and Product Integrity Program, Biosecurity Queensland). The PSC met twice yearly to oversee project direction and provide feedback on project deliverables.

Below we outline in more detail the methodologies employed across the four major components of this project.

Component 1) Surveillance preparedness

Component 1a) Surveillance and diagnostic tools

To facilitate the creation of reliable and cost-effective surveillance plans for exotic *Liriomyza* across diverse surveillance goals, industries, and jurisdictions within Australia, this project undertook several experimental studies to develop effective surveillance and diagnostic tools. This included: (i) quantifying the level of effort required for visual detection of leaf mine damage; (ii) the development of a new 'environmental DNA' or 'eDNA' based molecular test that allows diagnosis of empty leaf mines; and (iii) updating qPCR molecular assays for several economically significant *Liriomyza*, including VLM, ASLM, SLM*, L. bryoniae*, *L. chinensis,* as well as the common Australian species *L. brassicae*. Methods for each of these three subcomponents are summarized below, and full details will be available in associated scientific publications.

(1) measurement of surveillance effort for leaf mine damage

Due to the elusive nature of the *Liriomyza* flies and the characteristic leaf mining caused by the larvae, early detection efforts are best focused detecting leaf mine damage. A 'mock surveillance' approach was used to quantify the level of survey effort (as time spent searching per area) necessary for surveillance programs to detect leaf mining with a predetermined confidence level when pests are present at low densities (1% to 3% of plants affected). Mock surveillance is a useful method whereby an exotic pest is visually simulated and the detection probability of practitioners under various conditions is measured (Moore et al. 2011; Hauser et al. 2012, 2016). A major benefit of this approach is that locally meaningful detection rates can be investigated under a range of field conditions and observer behaviours, without the requirement of pest presence. We undertook, two 'on-farm detectability trials', involving farmers, farm advisors and other industry personnel, and used mock leaf mine damage to quantify the detectability of damage in commercial vegetable crops in Victoria. We then undertook two 'mock survey validation trials', which involved gardeners and government biosecurity officers in the TSI where VLM are present; these were run to compare the detectability of mock leaf mine damage against real damage caused by VLM. The results of these trials were analysed together to recommend a surveillance speed that would maximize detection likelihood by balancing survey effort with area coverage (Pirtle et al. *in preparation b,* see 'Refereed scientific publications' for an abstract). Finally, two 'survey guideline trials', involving land and sea rangers and government biosecurity officers in northern Australia, were performed. These examined the effectiveness of the surveillance guidelines derived from the first two experiments on detectability rates.

(2) development of an eDNA test for empty leaf mines

Experimental work was conducted within the TSI and NPA between 2018 and 2019 to develop a robust diagnostic

method for empty leaf mines based on environmental DNA (eDNA), which exploits traces of DNA left inside 'empty' leaf mines (Pirtle et al. 2020). This involved three major experiments. The first, conducted on Thursday Island, involved recording emergence dates of VLM larvae from leaf mines, and then collecting and preserving those leaf mines at a variety of known 'ages post fly emergence' up to one month. The second, conducted on Thursday Island, involved the collection of a large sample of randomly selected leaf mines, known to be created by VLM, of various ages and sizes to test the 'in-field' sensitivity of the test. The third experiment involved collection of empty leaf mines of unknown leafminer origin and applying the eDNA test to look for evidence of host range and geographic range expansions by VLM.

(3) updating qPCR tests for economically significant *Liriomyza* and the creation of an in-field diagnostic test

The existing qPCR molecular assay for VLM, ALM and ASLM (Sooda et al. 2017) was updated to ensure compatibility of the VLM primers to the haplotypes present within the TSI and NPA. Three additional qPCR assays were developed, to allow diagnosis of the exotic *L. bryoniae* (a polyphagous pest of many vegetable crops, such as tomatoes, and widespread in China) and *L. chinensis* (a pest of *Allium* crops, common in east Asia) as well as the common naturalised Australian species, *L. brassicae* (not considered a pest but visually indistinguishable from its exotic relatives and preferring many of the same crops, therefore posing considerable problems for surveillance).

Furthermore, assays for each of these six species were tested against one another as well as an array of native and naturalized species present in Australia, including *Liriomyza chenopodii, L. katoi, L. yasumatsui, Ophiomyia alysicarpi, Ophiomyia solanicola, Phytomyza plantaginis, P. syngenesiae and Ceradontha sp.*, to ensure specificity of each assay to its target species alone. Finally, the qPCR assays were tested using a field-ready multiplex system (Biomeme three 9 system) in Melbourne, Victoria, to distinguish *L. brassicae* from *P. syngenesiae* (van Rooyen et al. *in preparation,* see 'Refereed scientific publications' for an abstract).

These various experiments were combined with a literature review of factors known to influence surveillance for *Liriomyza* spp. (Pirtle et al. *in preparation a,* see 'Refereed scientific publications' for an abstract). This included biological and ecological factors, mathematical and statistical guidelines for identifying the most cost-effective options, and quantifying the level of detection confidence associated with those options. These learnings have been incorporated into the industry contingency plans (see below). This review will be published in a peer-review journal and should serve as the starting point for the development of surveillance plans for exotic *Liriomyza* spp. within Australia.

Component 1b) mapping

Four surveillance field trips targeting agromyzid leafminers, their host plants, and their parasitoids were conducted within QLD between 2018 and 2019. This included a 2.5 week road trip from Cairns to Seisia (in the NPA) and then into the TSI which aimed to: (i) survey for VLM and its host plants through regions of Cape York Peninsula (CYP) and into the TSI; (ii) survey for parasitoids attacking VLM in the TSI and NPA, and (iii) survey for possible reservoirs of parasitoid wasps in other native agromyzid species found throughout the CYP.

The results of these surveillance trips were combined with existing records for VLM, parasitoids, and known host plants to create an interactive map (available at https://cesaraustralia.shinyapps.io/VLMportal/) to explore distributions across Australia.

Component 1c) Risk models

To further develop our understanding of the risk profiles of the exotic *Liriomyza*, a novel, spatially-explicit simulation framework, called the Establishment, Spread, Impact and Management framework (ESIM) was applied to create establishment, spread and impact risk predictions for VLM, ASLM, and SLM in Australia. The framework includes modules for population establishment, growth, and spread, which are overlaid onto the spatial distribution of susceptible horticultural productivity to estimate economic impacts through time under various incursion scenarios (Maino et al. 2019, Maino et al. *in preparation a,* see 'Refereed scientific publications' for abstracts). This approach was also applied to predicting the distribution of parasitoids within Australia (Maino et al. *in preparation b,* see 'Refereed scientific publications' for an abstract).

In this exercise, we: (i) predicted the suitability of climate for each species in Australia, (ii) collated production values for at-risk crops in Australia, (iii) predicted the risk of each pest species spreading through production regions, and (iv) estimated the economic cost to industry based on the potential for each pest to establish and

spread to high-risk commodities.

Component 1d) Incursion pathway analysis

To identify the most likely pathway taken by VLM to reach the TSI, we conducted a RADseq (restriction site-associated DNA sequencing) incursion pathway analysis. We sourced samples of VLM from across the globe, including locations in the mainland USA and Hawaii, China, Vietnam, Kenya, Papua New Guinea, Timor Leste, and Indonesia. Local samples were sourced from Seisia and the TSI. Using double-digest Restriction Site-Associated DNA (ddRAD) sequencing, we developed a panel of 5095 single nucleotide polymorphisms (SNPs) based on 25 worldwide VLM populations. We used this genomically-based approach to study the genetic structure and the differentiation of populations and to the understand origin(s) and connections of the recent invasions into Australia (Xu et al. *in preparation,* see 'Refereed scientific publications' for an abstract).

Component 2) Management preparedness

Component 2a) Biocontrol

A review was compiled, relying on published literature, covering current knowledge of the parasitoids of VLM, ASLM and SLM in different areas of the world, and the parasitoids of agromyzid species already present in Australia. It identified research priorities that if undertaken, would enable growers to fully integrate these natural enemies into control strategies. Additionally, this information was paired with surveys conducted within the CYP and TSI targeting agromyzid leafminers, their host plants, and their parasitoids, aiming to identify parasitoids attacking VLM in Australia, and identify possible reservoirs of parasitoid wasps in other native agromyzid species found along the CYP.

This project also supported two postgraduate students in the PEARG group at the University of Melbourne to undertake studies on aspects of Australian parasitoids of agromyzids. Marianne Coquilleau completed her M. Phil. thesis focusing on the distribution and seasonality of parasitoid wasps found on Australian agromyzids (Coquilleau *in review*). Xuefen Xu has focused on molecular studies of agromyzids and associated parasitoids as part of her PhD. She has used several molecular markers to characterise the main Australian agromyzid and parasitoid species. She is also determining the occurrence, spatial distribution and identity of the endosymbiont bacteria *Wolbachia* in agromyzid species and their parasitoids in Victoria.

Component 2b) Chemicals

A review was compiled, relying on published literature and data available within the APVMA, which reports on chemical control options employed overseas for control of VLM, ASLM, and SLM, and consolidates the relevant information in preparation for appropriate chemical permits for ongoing management of these species in Australian horticultural crops. Included in the review is a consideration of the potential for insecticide resistance, the effect of chemicals on beneficial species, and eradication of pest incursions into production regions.

In order to provide control options to growers this project developed a number of APVMA permits. Pesticide candidates for permits were selected based on their efficacy against *Liriomyza* spp. and their suitability for use in Integrated Pest Management systems, which was based on the previously described review of scientific literature, outputs from this project and overseas labels. Additionally, a comparison of Australian and overseas use patterns (i.e. rates/crop) was undertaken to identify those pesticides that are used overseas for leafminer control at rates comparable to existing Australian use patterns. This comparison ensures that permits were only applied for those products that are effective against *Liriomyza* and do not pose residue, operator or crop safety issues, therefore providing suitable control options for Australian producers.

Component 3) Communication and engagement

Communication and engagement efforts aimed to raise awareness of *Liriomyza* spp., their impact overseas, and potential impact in Australia. Communication and engagement also incorporated a research extension aspect, whereby industry would be informed about R&D developments within the project relating to VLM control options,

in order to increase industry preparedness.

At the commencement of the project, we developed a communication and engagement (C&E) plan for the vegetable and nursery industries, which was subsequently updated when the project was expanded to include SLM and ASLM, to include C&E activities for the melon and potato industries.

In line with the C&E plan, communication and extension activities were facilitated in Australian horticulture regions. Such activities included industry workshops, grower visits, written updates for industry magazines, e-newsletters, factsheets, webinars, videos, CYP engagement, and a project extension package.

In the first two years of the project, extension activities were undertaken in all horticultural producing states and territories. During these engagement trips, the project extension team worked closely with regional industry bodies, grower associations, large growing operations, and consultants, to deliver workshops and mock surveillance activities. Invitations were extended to vegetable and nursery producers as well as the wider industry. Communication activities were produced in line with these extension activities based on industry needs and as required by the project.

Communication and engagement activities evolved with the project. High-risk regions were identified based on the results of modelling. The results found that high-risk locations include Lockyer Valley, Mareeba/Atherton, Bowen-Gumlu, Bundaberg, Sydney, Melbourne, Devonport, Darwin, and Perth. The modelling results allowed extension efforts to focus on these high-risk regions to deliver targeted engagement activities. In 2019-20, workshops were delivered in Mareeba/Atherton, Bowen-Gumlu, Bundaberg, Sydney, and Darwin. However, due to COVID-19, Perth, Lockyer Valley, Devonport, and Melbourne workshops were delivered as webinars.

Intrinsic to C&E methodology was monitoring and evaluation of project activities. A monitoring and evaluation (M&E) plan was developed at the outset of the project, and all C&E activities were undertaken in line with this plan. The M&E plan sought to answer a range of key evaluation questions in areas of effectiveness, relevance, process effectiveness and efficiency. Throughout the project, M&E data was supplied to Hort Innovation in each milestone report. A detailed analysis of M&E, including responses to key evaluation questions, has been provided in the 'Monitoring and evaluation' section of this report.

Component 4) Contingency plans

Three continency plans for VLM, ASLM and SLM were reviewed and extensively updated. Contingency plans follow standard PHA contingency plan templates and utilised the learnings from other components of the project such as the surveillance and modelling work. A literature review was undertaken to gather the most recent information on the biology, host plants, diagnostics and control/management options for each pest in order to support the development of the contingency plans.

Each contingency plan was reviewed by government and industry stakeholders as well as all PSC members. At project completion, the three contingency plans will be circulated to government agencies to support emergency response in the event of a *Liriomyza* spp. detection. A Draft copy of the SLM contingency plan was provided to the Consultative Committee on Emergency Plant Pests (CCEPP) which has formed in response to the recent detection of SLM in NSW and QLD (AUSVEG 2020).

Outputs

Surveillance preparedness

- A field-ready multiplex (qPCR) molecular diagnostic test, covering exotic pests VLM, ASLM, SLM*, L. bryoniae, L. chinensis*, and naturalized Australian species *L. brassicae,* has been developed, with specificity ensured against a range of other native and naturalized agromyzids within Australia (van Rooyen et al. *in preparation*, see 'Refereed scientific publications' for an abstract).
- A novel diagnostic method for diagnosing VLM via extraction of trace environmental DNA left behind within empty leaf mines. The leaf mine eDNA method developed within this project has been shown to be robust under field conditions, producing positive results in the majority of randomly collected field samples (Pirtle et al. 2020). This test can be extended to ASLM and SLM via the experimental work undertaken in the qPCR assay development.
- An analysis on the probability of wind-assisted dispersal of VLM using the HYSPLIT particle dispersion model and CSIRO's TAPPAS interface. This included an analysis of risk-based models and risk-based maps, which incorporated considerations for vulnerable commodities, climate-based establishment risk, seasonality, and human-assisted dispersal pathways. The report is available in Appendix 1.
- A novel, spatially-explicit simulation framework, called the Establishment, Spread, Impact, and Management framework (ESIM) was applied to create establishment, spread and impact risk predictions for VLM, ASLM, and SLM in Australia (Maino et al. 2019, Maino et al. *in preparation,* see 'Refereed scientific publications' for an abstract). The framework includes modules for population establishment, growth and spread, which are overlaid onto the spatial distribution of susceptible horticultural productivity to estimate economic impacts through time under various incursion scenarios. A report is available in Appendix 2 which demonstrates how model outputs can inform incursion responses.
- Two online interactive web tools were created, based on the ESIM framework and distribution mapping activities. This includes: (i) the VLM portal, which includes maps of VLM distribution, host plant distributions (weeds and cultivated crops), parasitoid distributions, VLM establishment risk predictions and VLM spread risk predictions; and (ii) the DARABUG2 lifecycle estimation tool, which allows lifecycle stage durations to be calculated across regions and seasons for VLM, ASLM and SLM.
    - The VLM portal can be accessed here: https://cesaraustralia.shinyapps.io/VLMportal/
    - DARABUG2 can be accessed here: https://cesaraustralia.shinyapps.io/darabug2/
- A comprehensive review, which consolidates experimental, statistical, and predictive work to create reference material for the development of technically justified and cost-effective surveillance programs within Australia for exotic *Liriomyza* (Pirtle et al. *in preparation a,* see 'Refereed scientific publications' for an abstract). This work will serve as a reference point for the development of surveillance plans for exotic *Liriomyza* within Australia across a range of surveillance goals including early detection, delimiting and area freedom surveillance.

Management preparedness

- A review of potential biological control agents effective against VLM was completed (see Appendix 3) and a modified version later published in the journal *Austral Entomology* (Ridland et al. 2020). This review was later expanded to cover ASLM and SLM (see Appendix 4).
- A review on mass-rearing techniques for biological control agents suitable for use in protected cropping (see Appendix 5).
- A report on control of VLM via their primary natural enemies, parasitoid wasps, as well as the parasitoid communities of additional native and naturalized agromyzid leafminer in the TSI and CYP. This was based on surveillance data collected across four field survey trips to these regions (see Appendix 6).
- A review of the chemical management options available for control of polyphagous *Liriomyza* spp. in Australian horticulture crops (see Appendix 7).

- Several minor use permits were secured (see Appendix 7 for a summary). In addition, we developed chemical permit packages for chemicals that do not currently have permits available. These are available to support future permit applications; they are accessible by contacting Plant Health Australia, AUSVEG, or Hort Innovation.

Communication and engagement

- Major C&E outputs resulting from this project include a project awareness flyer, *Liriomyza* species factsheet, an exotic leafminers speed drawing (original video developed in 2018, and updated in 2020), NPA and TSI community guides, industry preparedness and management guides, region-specific guides, webinars, articles, and podcasts.
- In addition, we delivered 19 workshops across all horticultural producing states and territories, delivered extension activities in the NPA and TSI in 2018 and 2019, and visited more than 100 vegetable/potato growers over the lifetime of the project. A full list of C&E outputs, with links where relevant, can be found in Appendix 8.
- Additionally, a CYP Biosecurity Scoping Study Report was developed to support engagement with NPA and TSI communities and foster shared biosecurity goals, which can be found in Appendix 10 as well as here: https://www.horticulture.com.au/growers/help-your-business-grow/research-reports-publications-fact-sheets-and-more/cape-york--torres-strait-biosecurity-scoping-report/

Contingency plans

- Contingency plans for VLM, ASLM and SLM. Copies of the contingency plans, which include comprehensive host plant lists, can be found in Appendix 10, 11 and 12. These plans have been provided to the Subcommittee on National Plant Health Surveillance for consideration.

Scientific publications

- We have written several scientific papers, which have either already been published in peer-reviewed scientific journals or will be published in the near future. See ‘Refereed scientific publications’ for further information.

Outcomes

This project has increased industry preparedness in the event that production areas experience an incursion of VLM, ASLM, and SLM. This outcome has been achieved via the holistic approach the project took towards preparedness, building knowledge in surveillance and management techniques and relying heavily on the involvement of stakeholders, in particular indigenous communities in the TSI and Far North QLD where VLM is present. Education provided through the extension program has increased knowledge among communities affected by VLM, as well as production groups at risk of VLM, ASLM, and SLM. The project has increased Australia's capacity to ensure faster response time to outbreaks; thereby increasing chances of successful eradication, through the development of contingency plans, effective surveillance and diagnostic tools, emergency use permits, biological and chemical management advice, and community engagement. Indeed, Australia experienced an incursion of SLM within the final month of the project's completion, and project outputs were able to be immediately leveraged to respond rapidly to the incursion and are facilitating a transition to management.

Specific outcomes are detailed below.

Surveillance preparedness

Desired outcome: Improved capacity for rapidly detecting and diagnosing incursions of exotic *Liriomyza* spp., improving chances of containment and eradication.

Achievement: This was achieved via the creation of technically justified and cost-effective surveillance and diagnostic techniques for *Liriomyza* spp. meeting international IPPC standards (International Plant Protection Convention, 1997) to support early detection, delimiting and area freedom surveillance goals. Surveillance guidelines were created by quantifying the likelihood of detection for leaf mine damage during visual surveys and were then validated via biosecurity officer and ranger training activities in the TSI and in Cairns, QLD. Validations confirmed that the surveillance guidelines improved detection probability by 3 times (Pirtle et al*. in preparation b*). Moreover, capacity for diagnosing incursions was improved via the creation of a novel diagnostic test for leaf mines, which allowed for the confirmation of host range expansions of VLM within TSI and the NPA as well as the confirmation of suspected occurrence records which previously could not be confirmed through the traditional reliance upon collection of live flies (Pirtle et al. 2020). Finally, diagnostic capacity was improved via the validation of rapid in-field qPCR assays, for both leaf mines and flies, targeting exotic *Liriomyza* spp. (van Rooyen et al. *in preparation,* see 'Refereed scientific publications' for an abstract). All surveillance and diagnostic techniques developed through this project have been incorporated into pest specific contingency plans, are being incorporated into National Surveillance Protocols, are/will be published as peer reviewed scientific articles, and have been extended to key government and industry representatives (including members of the Subcommittee on National Plant Health Surveillance, chief plant health officer departmental staff, state government biosecurity extension officers, vegetable industry development officers, and regional grower groups) to ensure availability and adoption.

Desired outcome: Improved effectiveness of surveillance programs targeting *Liriomyza* spp. in Australia, and improved incursion responses, including targeted containment and control measures.

Achievement: This was achieved through the application of a novel modelling framework that predicts possible growth rates and rates of spread, based on biological knowledge, biophysical approaches, and modern climatic datasets. Additionally, existing modelling programs were used to investigate the risk of natural spread into northern Australia via wind patterns, ultimately supporting the conclusion that human mediated spread pathways are of higher risk for exotic *Liriomyza* spp. Thus, project activities, including further predictive and awareness raising work, focused on high-risk human pathways. Indeed, the recent incursion in the greater Sydney region is located around one of Australia's most populated and high-risk regions. The ESIM predictions for VLM have supported surveillance and preparedness planning through their presentation in the user friendly VLM portal, while predictions for all three species formed core components within contingency plans and will be published as peer reviewed scientific articles. Moreover, risk predictions have been extended to key government and industry representatives (including members of the Subcommittee on National Plant Health Surveillance, chief plant health officer departmental staff, state government biosecurity extension officers, vegetable industry development

officers, and regional grower groups) to ensure availability and adoption. The project's focus on creating flexible predictive tools allowed for the rapid creation of predictions specific to the parameters of the recent incursion of SLM into the greater Sydney region, which were provided to the CCEPP within two days of notification of the incursion.

Desired outcome: Determine the most likely origin of the incursion population of VLM present in the TSI and NPA, thus improving understanding of pathway risks.

Achievement: This was achieved through the completion of an incursion pathway analysis for the Torres Strait populations of VLM, using overseas VLM samples and next-generation molecular sequencing tools. This analysis identified that VLM populations from Australia including TSI (Seisia, Thursday Island, Boigu Island, Masig islands) are genetically similar, and all appear to have connections to Bali, suggesting that regions around Indonesia may represent a probable source of the VLM detected in Australia.

Desired outcome: Confirm present geographic and host range of VLM within the TSI and NPA, assessing any evidence for pest presence along CYP, and map the distribution of potential host plants and natural enemies for VLM across Australia to determine establishment potential as well as possible options for biological control.

Achievement: This was achieved via four field surveys conducted within the TSI and CYP aimed at mapping the distribution of VLM near the incursion front. The 2018 survey trip along CYP yielded no evidence of VLM presence south of its known range in Seisia, QLD. Within the TSI and NPA, an additional occurrence record was confirmed for Zuna/Entrance Island through the use of the novel eDNA test for leaf mine damage. Across all field surveys, several new host preferences for the TSI and NPA populations were recorded, including chili, basil and passionfruit (all common garden plants) as well as a common weed, snakeweed, (*Stachytarpheta jamaicensis*). Unconfirmed evidence of VLM was observed in another common weed *Alternathera ficoidea* (joyweed), While neither of these weeds appeared to be highly preferred (with limited numbers of leaf mines detected in snakeweed on both Thursday Island and Horn Island and confirmed as VLM via molecular tests, and with a single suspicious mine discovered in joyweed on Thursday Island which could not be confirmed as VLM on the spot), their detections are still notable. Should VLM's preference for these common weeds grow, their abundance would contribute to increasing host availability and connectivity within Cape York Peninsula (between the NPA and production areas further south).The VLM portal tool was created to compare distribution of VLM within Australia to the distributions of common weed hosts and of likely natural enemies. Moreover, a review of literature along with additional surveillance in the TSI and CYP and a postgraduate student project yielded additional records for parasitoid wasps in Australia, which were integrated into the VLM portal. These maps have facilitated the communication of risk during extension activities, via a depiction of both the ubiquitous presence of available plant hosts for VLM across Australia, as well as a promising distribution of parasitoid wasps.

Management preparedness

Desired outcome: Establishment of foundational knowledge to support future Australian IPM programs for exotic *Liriomyza* spp. and development of information packages for industry that provide scientifically robust methods for monitoring, eradication (if considered feasible), and management.

Achievement: Historic knowledge of *Liriomyza* spp. control was thoroughly reviewed, including IPM programs involving monitoring techniques, economic thresholds, chemical options, biological control options, in particular through the use of parasitoid wasps, augmentative release programs and mass rearing protocols, and instances of reported insecticide resistance. This information was compiled into four industry reports (Appendices 3, 4, 5, and 7), and simplified into a concise grower management guide which was distributed at all project workshops (linked within Appendix 8). The key messaging around the importance of natural control of exotic *Liriomyza* spp. and the risk of secondary release as a result of improper chemical usage has been a strong focus of all project awareness raising activities, as well as during government and industry update webinars responding to the recent SLM incursion. Identification of parasitoids attacking the incursion population of SLM are already underway through government surveillance, supported by the project team. Identification of these parasitoids will be facilitated through the morphological and molecular identification work already completed in this project. Detailed recommendations for IPM techniques that can be implemented immediately, as well as those that require further data to be collected now that SLM is present within Australia, are provided later in this report.

Additionally, postgraduate students increased our understanding of Australian parasitoids of agromyzids and how they might be leveraged for biocontrol. Marianne Coquilleau determined that common parasitoids within Victoria include *Hemiptarsenus varicornis*, an unidentified *Asecodes* sp., *Chrysocharis pubicornis*, *Diglyphus isaea,* and *Neochrysocharis formosa,* and she demonstrated that there were reservoirs of parasitoid wasp species in weedy areas which could potentially exploit polyphagous *Liriomyza* leafminers in Australian horticultural regions. The all-female line of *N. formosa* is the first Australian record of this adventive species (Coquilleau *in review*). Xuefen Xu has used molecular markers to characterise the most abundant parasitoids of agromyzids in Victoria, which will assist in the identification of parasitoids of SLM in Australia. She has also sequenced the full genome of the *Wolbachia* species infecting *L. brassicae*, laying groundwork for important investigations into the potential for *Wolbachia* to improve mass rearing prospects for *Liriomyza* parasitoids.

Desired outcome: Proactively secure chemical permits to ensure rapid responses to incursions.

Achievement: Emergency use 'shelf' permits were developed, with a focus on VLM due to its presence in Australia. Importantly, these permits also included ASLM and SLM wherever was possible (and wherever not possible, information packages were prepared to ensure rapid permit approval after an incursion). This resulted in 5 permits already being available upon SLM's incursion into NSW and QLD, and the approval of an additional 4 permits within one month after the incursion, at the time of writing this report.

Communication and engagement

Desired outcome: Increased awareness of *Liriomyza* spp. through targeted extension activities in vegetable, nursery, melon and potato growing regions.

Achievement: During the project, project partners used ESIM outputs to identify regions and crops at high risk of pest establishment, spread and impact. Focusing on the regions identified in these models, we undertook workshops and delivered targeted materials in those locations to increase awareness of *Liriomyza* spp. At the project outset, networks were developed with key industry stakeholders such as Greenlife Industry Australia and Australian Melons Association to communicate project updates in order to raise awareness of *Liriomyza* spp. In regions identified as high-risk, networks were established with local grower groups such as Bundaberg Fruit and Vegetable Growers' Association. More information on how these achievements were monitored and evaluated can be found in the monitoring and evaluation section of this report.

Desired outcome: Increased uptake and understanding of project research learnings to ensure that industry and government are adequately prepared for exotic *Liriomyza* spp.

Achievement: Increased industry and government uptake and understanding of preparedness activities for *Liriomyza* spp. was achieved through provision of specific resources, mock surveillance activities, educational workshops based on RD&E developed throughout the project, and government meetings. Project partners developed an industry surveillance plan for VLM and an industry management plan for *Liriomyza* spp. based on research findings. The surveillance plan communicates scientifically robust ways to survey a farm for the presence of leafminers. In addition, surveillance training activities were undertaken to maximise early detection likelihood for *Liriomyza* spp. Growers and agronomists undertook mock surveillance exercises in Tyabb, VIC and Werribee, VIC, which acted as preparedness activities. These activities also assisted with the development of surveillance guidelines which further increased industry preparedness. A mock surveillance activity was also undertaken with growers and agronomists in Atherton, QLD, however this was for extension purposes only, and did not include a research element. In year three, we ensured that workshop and webinar presentations focused significantly on monitoring, surveillance, and management techniques in order to support industry. In addition to industry preparedness, we worked to increase uptake and understanding of preparedness research activities to government stakeholders including federal NAQS staff and state government (chief plant health officer departments and biosecurity extension officers). All jurisdictions have been updated on research learnings and were given access to the VLM preparedness plan, management plan, and regional response plans.

Desired outcome: Strengthened biosecurity engagement within communities in the TSI and CYP.

Achievement: In order to strengthen biosecurity-related engagement within communities in the TSI and CYP,

relationships were established with TSI and NPA stakeholders in the early stages of the project. These relationships assisted project partners to access the necessary audiences to communicate information on VLM and deliver successful extension trips. For example, project partners worked with Torres Strait Regional Authority (TSRA) from the project outset, and in the third year of the project, TSRA helped facilitate community-based VLM identification workshops on Thursday Island. Similarly, advice from project partner NAQS and TSRA significantly assisted project partners in coordinating ranger workshops and mock surveillance activities on Thursday Island and in Cairns. In order to further strengthen biosecurity-related engagement in CYP and TSI, the project team developed educational resources for these communities, based on research undertaken throughout the project. These resources included a parasitoid wasps guide and a VLM surveillance plan. The development of a CYP scoping study also strengthened biosecurity engagement. The CYP scoping study identified current biosecurity activities occurring in these regions and how the work of the project team was contributing. The study also highlighted key areas that could be focused on in the future to address gaps in current engagement activities.

Contingency plans

Desired outcome: Ensure industry preparedness for VLM, ASLM and SLM by updating existing industry contingency plans.

Achievement: Three pest specific contingency plans were created which represent the culmination of achievements within surveillance and management learnings. Each species specific contingency plan contains relevant biological and ecological pest information, analyses of high risk pathways, comprehensive host plant lists which indicate scientific confidence in the ability of the pest to complete its lifecycle in each host, establishment, spread and impact predictions via the ESIM framework, information regarding the most up to date and scientifically rigorous surveillance and diagnostic techniques, and summaries of biological and chemical control options. These contingency plans have been provided to the Subcommittee on National Plant Health Surveillance for consideration. The SLM contingency plan was able to be immediately leveraged upon SLM's incursion into Australia, informing discussions about market access and supporting efforts to delimit the affected area.

Monitoring and evaluation

In line with the project's M&E plan developed at the project outset, project partners have answered the following key evaluation questions, which set out to monitor the effectiveness, relevance, process effectiveness and efficiency of the project. *(Note: While several key evaluation questions, developed at the project outset, are specific to VLM, due to the project variation - which expanded focus of communication and extension activities to ASLM and SLM - some monitoring and evaluation data reported herein applies to all three species. These instances are indicated with an asterisk.)*

Key evaluation question: To what extent has the project increased industry knowledge, awareness and preparedness for VLM*?

Project communication and extension activities have evidently increased industry knowledge, awareness and preparedness for VLM. During the project, 19 workshops were delivered to more than 450 people across all horticulture producing states and territories of Australia. The monitoring data indicates that as a result of these workshops, producers are subsequently more knowledgeable about the pest's biology, distribution, identification, and potential impacts.

At some workshops, monitoring data was collected both before and after presentations. This includes workshops held in Bowen (2017), Tyabb (2018), Bundaberg (2018), Mareeba (2018), Werribee (2018), Shepperton (2018), and a project webinar (2020). Data shown in Table 1 highlights new knowledge gained as a result of workshops and webinars in various regions. In general, knowledge change was continually high following workshops. For example, before the Bowen (2017) workshop, eight participants stated that they had 'low' knowledge of VLM and five stated that they had 'moderate' knowledge. Following the workshop, zero stated that they had 'low' knowledge, 11 had 'moderate' knowledge, and three had 'high' knowledge. Overall, the majority of respondents also stated that project outputs would be useful for their business. Other workshop monitoring data has been included in Appendix 13 (M&E data).

Table 1. Knowledge change of VLM through workshops and project webinar

Location	**Level of knowledge of VLM before workshop** *Note that not all participants answered both questions, leading to differences in total number of responses.*	**Level of knowledge of VLM after workshop** *Note that not all participants answered both questions, leading to differences in total number of responses.*	**How useful do you expect project outputs to be for your occupation?**
Bowen 2017	High (0)	High (3)	Highly useful (7)
	Moderate (5)	Moderate (11)	Moderately useful (3)
	Low (8)	Low (0)	Slightly useful (4)
			No (0)
			N/A (0)
Tyabb 2018	High (0)	High (1)	Highly useful (0)
	Moderate (3)	Moderate (7)	Moderately useful (4)
	Low (6)	Low (1)	Slightly useful (0)
			No (0)

			N/A (0)
Bundaberg 2018	High (1)	High (4)	Highly useful (6)
	Moderate (2)	Moderate (7)	Moderately useful (4)
	Low (7)	Low (1)	Slightly useful (0)
			No (0)
			N/A (0)
Mareeba 2018	High (2)	High (6)	Highly useful (4)
	Moderate (1)	Moderate (2)	Moderately useful (4)
	Low (5)	Low (4)	Slightly useful (2)
			No (0)
			N/A (1)
Werribee 2018	High (1)	High (18)	*Question not asked*
	Moderate (0)	Moderate (7)	
	Low (28)	Low (2)	
Shepperton 2018	High (0)	High (4)	Highly useful (3)
	Moderate (2)	Moderate (3)	Moderately useful (2)
	Low (5)	Low (0)	Slightly useful (1)
			No (1)
			N/A
Project webinar 2020	High (3)	High (9)	*Question not asked*
	Moderate (18)	Moderate (18)	
	Low (13)	Low (9)	

In addition to workshop M&E data, some community impact data was collected by examining the suspected exotic leafminer reports made to the Victorian government by community members. These reports, given their relatedness to the project, were forwarded to the project team. Seven reports were submitted based on information coming from the project webinar, the Department of Agriculture, Water and the Environment's website, or through MyPest Guide reporter. In most cases, reports were *Phytomyza syngenesiae* and in one case, the pest was not an agromyzid (see Appendix 13 for more information).

We have developed an industry legacy plan, based on engagement with industry and communities over the course of the project. The legacy plan outlines projects and organisations that could be contacted in future to ensure legacy of MT16004 project outcomes (see Table 2 below).

Table 2. Legacy arrangements for Project MT16004

Project legacy arrangements		
Project/program	**Summary**	**Action taken**
VegNET project	The Vegetable Industry extension network has officers stationed in major production zones throughout Australia. They are tasked with extending vegetable RD&E project outputs to industry.	VegNET Industry Development Officers informed about the project. They have received the extension resource package.
AUSVEG Farm Biosecurity Program	The project will run until 2021. Project officers incorporate updates on exotic leafminers using information provided by the Hort Innovation project. The urban biosecurity subproject includes exotic leafminer awareness components.	Exotic leafminers incorporated into project extension and communication activities. Information on AUSVEG website. Extension resource package provided to project officers.
Greenlife Industry Australia Biosecurity Project	The national biosecurity project that maintains BioSecure HACCP as well as represents industry during exotic pest incursions.	Exotic leafminers incorporated into project extension and communication activities. Information on Greenlife Industry Australia website. Extension resource package provided to project officers.
Australian Melon Association Biosecurity Project	Newly launched project through the plant health levy.	Exotic leafminers incorporated into project extension and communication activities. Information on Australian Melons Association website. Extension resource package provided to project officers.
ExtensionAUS platforms	The ExtensionAUS platform and the Community of Practice (COP) sites it hosts have been developed to meet the needs of rural and regional Australia for the extension of knowledge and to enable collaboration on important issues. The platform is likely to become more horticulturally focused in coming years.	AUSVEG and Cesar Australia contribute extension materials and existing communications materials to Urban Plant Health Network CoP.
Plant Health Australia	Host resources on Plant Health Australia website.	Resources provided to Plant Health Australia to host on their website.
Queensland Department of Agriculture and Fisheries	Host resources on Business Queensland Vegetable Leafminer page.	Resources uploaded to Business Queensland Vegetable Leafminer page.
Local and regional councils	In QLD, local councils have a responsibility to develop a biosecurity plan for their region.	Meetings held to provide information for consideration and inclusion in updated biosecurity plans across QLD in future.
Torres Strait councils	In QLD, local councils have a	Meetings held to provide information for

	responsibility to develop a biosecurity plan for their region.	consideration and inclusion in updated biosecurity plans in the TSI in future.
Cape York Peninsula Scoping Study	Identifies further legacy arrangements for the CYP and TSI	Formally submitted to Hort Innovation and accessible on their website. See Appendix 9 for the full report.

Key evaluation question: To what extent has the project increased knowledge and awareness of VLM* in CYP and Thursday Island?

Engaging with CYP and Thursday Island residents, rangers, and schools actively increased knowledge of VLM in the region. The first engagement trip, occurring in 2018, focused on engagement with students, tourists, communities, rangers, and various associations. This trip resulted in the development of a scoping study, which is available on the Hort Innovation website (see Appendix 9). The second engagement trip, occurring in 2019, saw the project team returning to CYP and Thursday Island, engaging with students, communities, and rangers.

Rangers

Rangers are responsible for managing a range of plant and animal pests and diseases of biosecurity importance across the TSI and NPA. Before the project team engaged with the rangers, there was limited understanding of VLM including its host list, biology, identification, surveillance, and management. In 2018, we delivered briefings for Apudthama Land and Sea rangers (Seisia, Bamaga, Injinoo) and Torres Strait Island Regional Council staff, and Torres Strait Regional Authority Land and Sea rangers. In May 2019, the project team delivered training sessions to approximately 40 NAQS Biosecurity Officers on Thursday Island and approximately 20 Torres Strait Regional Authority rangers, from both TS and NPA, who were participating in a Biosecurity Fundamentals workshop in Cairns.

Schools

In 2018, the project team undertook mock surveillance activities with primary school students and delivered presentations on the pest's biology, parasitoid wasps and VLM damage to secondary school students. The secondary schools expressed an interest in future visits from project members. In 2019, the project team facilitated engagement activities with schools, two of which were the same schools as visited in 2018 (see Appendix 13 for images taken at these workshops). Resources were shared with teachers, who mentioned that they would use these resources in future teaching materials.

Community

In both 2018 and 2019, the project team engaged with communities in CYP and the TSI. These included residents, tourists, and business owners. A tourist survey was undertaken in 2018 to gauge the level of biosecurity awareness of short-term visitors to CYP, find out where they source biosecurity information before travelling, and determine the origins of these tourists. It was found that 90% of tourists surveyed were domestic visitors and the majority (95% of tourists surveyed) had driven up the Cape through Coen, QLD. In response to the question 'Have you ever heard of the vegetable leafminer?' 15% of those surveyed answered 'yes' and 85% answered 'no'. Of those who answered 'yes', sources of knowledge were Landline, TAFE, TV, Online, and previous employment at QDAF. In response to the question 'Has your new knowledge raised your awareness about the importance of biosecurity on CYP and in the Torres Strait Islands?' 97% of respondents answered 'yes' and 3% answered 'no' adding that awareness was already high. It was also noted that information packages were available to collect from the Cape York Inspection Peninsula Biosecurity Centre in Coen, Queensland. More details are provided in Appendix 13. In 2019, the project team focused on working with residents of Thursday Island, facilitating a 2-hour training session whereby residents were given the opportunity to identify VLM and parasitoid wasps under a microscope and learnt about VLM biology, identification, damage, and management (see Appendix 13). Fifteen Thursday Island residents attended the workshop held at Frog Gully Community Garden, as well as staff from TSRA, AUSVEG, and Cesar Australia. Residents of Thursday Island had limited prior knowledge of VLM, however, the workshop helped raise awareness of the pest and its presence in TSI. Feedback from the training session suggested that communities would be more mindful of biosecurity in future, particularly when travelling from Thursday Island to other parts of Australia.

Key evaluation question: To what extent has the project met the needs of vegetable and nursery levy payers,

industry stakeholders (e.g. consultant agronomists) and the indigenous community at Seisia?

In order to meet the needs of vegetable, nursery, melon and potato levy payers, project partners developed extension materials, surveillance recommendations, and management options to support preparedness for *Liriomyza* pests. For example, we developed several preparedness and management plans which were distributed to industry via workshops across high-risk regions. The national preparedness plan, specific to VLM, provided an overview of the pest's biology, damage, and impact, as well as scientifically supported advice about conducting on-farm surveillance, sample collection and how to report suspect detections via the Exotic Plant Pest Hotline. Regionally-specific preparedness plans were also developed, which feature seasonal risk prediction data for the region, at-risk commodities in that region, and predicted regional impacts. The leafminer management plan provided foundational knowledge that would be necessary to adopt IPM approaches to management of *Liriomyza* pests, should they arrive. The project team provided information to support Greenlife Industry Australia's Pest ID tool (www.pestid.com.au), an open access website that is used by plant producers to help them identify, detect, and manage plant pests).

Following the detection of SLM in Greater Sydney, the project's focus on creating flexible predictive tools allowed us to rapidly create commodity-specific risk summaries for industry representatives (including the melons and nursery industries). This highlighted crops and production regions at highest risk of incursions and subsequent pest establishment.

The project has also met the needs of the indigenous community on CYP by facilitating training sessions with TSI and NPA ranger groups during 2018 and 2019. Following training sessions, we shared resources, including laminated images of VLM and feeding damage, a PowerPoint presentation file, and surveillance guides and biocontrol guides for distribution. These guides highlighted the importance of keeping SLM and ASLM out of the TSI and CYP and how early detection surveillance in these regions would support the rest of Australia. The guides included instructions for surveillance and reporting.

Key evaluation question: To what extent were the target engagement levels of vegetable and nursery levy payers achieved?

Throughout this project, the project team exceeded targets for engagement, both in communication and extension. The development of a C&E plan and M&E plan at the outset of the project were continually referred to in order to achieve engagement targets.

We delivered 19 workshops and eight webinars, which not only met but exceeded project targets. Key delivery regions included Perth, Adelaide, Melbourne, Devonport, Sydney, Lockyer Valley, Bundaberg, Bowen/Ayr, Mareeba/Atherton, and Darwin. There were a number of regions where in-person engagement activities and workshops occurred twice or three times over the course of the project. Where in person engagement could not occur as a result of COVID-19, workshops were replaced with webinars (see Appendix 14 for more information on changes due to Covid-19).

In terms of project communications, we delivered industry communiques including factsheets, videos, R&D articles in Vegetables Australia, Potatoes Australia and other industry magazines, quarterly e-newsletters detailing project activities and key dates for industry, and a project webinar. Conventional media was used to target more than 6,000 vegetable growers and 3,000 potato growers; this included 11 full-length (600-1000 word) articles for inclusion in Vegetables Australia and Potatoes Australia, as well as WA Grower, Lockyer Valley Growers Association News, Bundaberg 'Fresh Pickings' magazine, and Melon News. See Appendix 8 for more details.

Online media was used to target a wide audience and measure industry engagement. In particular, e-Bulletins including AUSVEG Weekly Update articles, AUSVEG Front Line e-Bulletins, and social media were utilised. The AUSVEG Weekly Update is distributed to more than 4,300 industry stakeholders and the AUSVEG Front Line Biosecurity Bulletin is distributed to more than 2,500 industry stakeholders. In total, 24 Weekly Update articles were written during the project's lifetime, which is an average of 8 per year, doubling our initial project targets. Furthermore, 12 Front Line e-Bulletins were distributed. Table 3 below shows the total number of Front Line e-Bulletins distributed throughout the project, highlighting email opens and link clicks. Email opens remained constant throughout the course of the project, whereas total link clicks tended to increase over time. See Appendix 8 for more details.

Table 3. Front Line e-Bulletins distributed during the project, including total email opens and link clicks

e-Bulletin title	Total opens (#)	Link clicks (#)
Announcement of new Hort Innovation funded project - MT16004 - RD&E Program for control, eradication and preparedness for Vegetable Leafminer (2017-2020)	770	17
Upcoming workshop - creating value from vegetable waste and managing vegetable leafminer	752	0
Upcoming events - Tyabb VIC and Bundaberg QLD	768	1
Upcoming pest and disease workshops in QLD - Ayr and Bowen	665	0
Upcoming workshops in Bundaberg QLD and Darwin NT	657	0
Learn about exotic leafminers	679	19
Learn about exotic leafminers (repeated)	663	14
Learn about exotic leafminers with this new video	575	41
Suspect serpentine leafminer (*Liriomyza huidobrensis*) detection in Western Sydney, NSW	570	8
Upcoming Gippsland-focused webinar	624	28
Serpentine leafminer (*Liriomyza huidobrensis*) detected in Western Sydney, NSW and Fassifern Valley, QLD	588	6

Social media was also used throughout this project to measure engagement. The AUSVEG Twitter account (@AUSVEG) has more than 4,700 followers and the AUSVEG Biosecurity Twitter account (@biosecurityveg) has more than 580 followers, the AUSVEG LinkedIn account has more than 3,400 followers, the AUSVEG Facebook Page has more than 3,500 followers, and Cesar Australia's Twitter account (@cesaraustralia) has more than 1,600 followers. There have been a significant number of social media posts during this project, however notable social media posts included a Tweet on serpentine leafminer by @AUSVEG, which received 6,925 impressions, and a Tweet about the Darwin workshop by @biosecurityveg, which received 2,742 impressions.

YouTube has also played a large role during this project, as it was the platform in which project webinars and videos were held. The VLM speed drawing developed in 2018 received 622 views and 983 impressions. The updated exotic leafminer speed drawing, developed in October 2020, has received 375 views to date. The exotic leafminers project webinar, in which Madeleine Quirk, Dr. James Maino, Dr. Elia Pirtle, Dr. Peter Ridland, and Rohan Burgess presented, was uploaded to YouTube as four shorter videos. The first video, 'introduction to exotic leafminers', received 54 views and 645 impressions. 'Assessing the regional risk of exotic leafminers' received 27 views and 585 impressions, 'Surveying for exotic leafminers' received 45 views with 816 impressions, and Management of exotic leafminers - biological and chemical control' received 53 views with 1,400 impressions. It is also important to note that this webinar was live, and there were 61 attendees watching the live broadcast. The Western Australia industry webinar facilitated in June 2020 received 52 views and 957 impressions. See Table 4 for further details.

Table 4. Project videos that are held on AUSVEG's YouTube channel, including upload date, views and impressions

Video	Date uploaded	Views	Impressions
VLM speed drawing	June 2018	622	983
Exotic leafminers speed drawing	October 2020	375	227

Project webinar - Introduction to exotic leafminers	July 2020	54	645
Project webinar - Assessing the regional risk of exotic leafminers	July 2020	27	585
Project webinar - Surveying for exotic leafminers	July 2020	45	816
Project webinar - Management of exotic leafminers - biological and chemical control	July 2020	53	1,400
Western Australia - Fall armyworm and exotic leafminers update	June 2020	52	957

Key evaluation question: To what extent have project outputs been used by government agencies?

Project outputs have been used by government agencies to guide future preparedness activities and further safeguard the vegetable, nursery, melon and potato industries. Moreover, government agencies have shared project outputs via their online platforms and networks to increase the project's reach.

The project partners engaged effectively with state governments throughout the project. A QDAF representative sat on the Project Steering Committee and QDAF currently hosts MT16004 project resources on the Business Queensland website. In addition, project partners worked closely with the Victorian Department of Jobs, Precincts and Regions (DJPR), exchanging samples and expertise. The project team also met with approximately 30 state government stakeholders from all jurisdictions to provide a final update on the project, including a summary of project outputs, project findings and future recommendations. At this meeting, government stakeholders were shown where the project extension package is held and trained in how to use the package. In particular, stakeholders commented on the usefulness of the industry preparedness plan, industry management plan, and regional response plans for industry stakeholders.

The Department of Agriculture Water and the Environment's NAQS staff assisted with activities that were undertaken in CYP and TSI throughout the course of the project. Furthermore, detailed information regarding surveillance guidelines and diagnostic options developed within this project was provided upon request to support future surveillance activities conducted by NAQS in CYP and TSI. At the end of the project, key NAQS staff were provided with a comprehensive extension package.

During the final month of the project, SLM was detected in Sydney, NSW. In order to support discussions and deliberations at the CCEPP meeting following the detection, the project provided the CCEPP with number of resources including the draft contingency plan, draft plant host list, a spread prediction summary paper, a summary of permits developed through the project and chemical permit packages for unsubmitted permits. The resources that were provided to the CCEPP were timely (provided within 2 days of project partners being informed of the incursion) and proved to be useful in supporting both government and industry during the incursion response process, emphasizing the significant extent to which the outputs have been utilized by government.

Key evaluation question: To what extent were key stakeholders satisfied with communication materials and extension events as a way to increase their knowledge and awareness of VLM*?

As discussed in response to previous key evaluation questions, key stakeholders were satisfied with communication materials and extension events. This is shown by the number of articles and updates written for conventional media and the number open rates, click rates, views, and impressions gained through online media. This has also been demonstrated by the extensive engagement undertaken in key production regions. Some anonymous examples have been included below:

- "I have seen the quick 4-minute video on LinkedIn and have reported suspected leafmines to the government" - Anonymous, 2019 regarding the VLM speed drawing developed in 2018.
- "The surveillance plan presents well with good information about the pest" - Victorian Agronomist, 2019 regarding the draft surveillance plan circulated to industry in 2019.
- "I enjoyed the webinar as it was very informative with knowledgeable speakers" - Victorian Agronomist, 2020 following the project webinar.

- "Engagement activities suggested by AUSVEG will provide good legacy outcomes" - Project Steering Committee, 2020
- "It (the webinar) was really interesting and well presented. As a result of watching, I thought the leafminer presentation would be good to use as an article in the next issue of our magazine." - Journalist for grower association, 2020.
- "I recently attended the leafminers webinar and I was wondering if you might be able to assist (with leafminer identification)" - State government staff member, 2020.
- "I was wondering if we could hold a meeting to discuss some work, I am proposing for next financial year for rangers to undertake host plant control and monitoring for leafminers in TSI and Northern Peninsula Area" - Ranger leader, anonymous, 2020.

Key evaluation question: Has new knowledge been gained about ways to improve biosecurity engagement with CYP communities?

New knowledge has been gained about the best courses of action for continuing engagement with CYP communities. Key recommendations have been outlined in the CYP Scoping Study (see Appendix 9), which was supplied to Hort Innovation in 2019. It is recommended that this document is used to help guide future engagement activities with CYP and TSI communities in the biosecurity space. These recommendations are further detailed in the 'Recommendations' section of this report.

https://www.horticulture.com.au/growers/help-your-business-grow/research-reports-publications-fact-sheets-and-more/cape-york--torres-strait-biosecurity-scoping-report/

Key evaluation question: Has new knowledge been gained about the risk of VLM* spread to production areas?

Through experimental work focusing on the likely pathway taken by VLM to arrive in the TSI, and the risk of further spread from this pathway, as well as entry via additional pathways across Australia, we have gained significant knowledge regarding the risk of VLM spread into production regions. First, we identified that the primary risk pathway for VLM is expected to be the movement of humans driving spread, rather than natural spread driven by normal wind patterns from Papua New Guinea or Timor Leste (however, extreme weather events significantly drive up the risk of natural spread; Appendix 1). Then, to further investigate the risk associated with human movement, we applied a novel simulation framework to create establishment, spread and impact risk predictions. This approach was also applied to better understand SLM and ASLM spread risk. The results of these predictions have implications for better targeting the location and timing of surveillance programs across a range of surveillance goals; and were also instrumental in targeting our project's awareness raising activities to production regions at highest risk of VLM incursions and establishment. Moreover, the ESIM framework has allowed tailored predictions to be rapidly created in response to the recent discovery of SLM in Western Sydney (see Appendix 2), to support the CCEPP which formed in response to this detection.

Key evaluation question: What efforts did the project make to improve efficiency (in scientific method and industry engagement)?

Several improvements were made to traditional surveillance and diagnostic methods for VLM, improving efficiency in scientific method. For example, a robust method based on environmental DNA (eDNA) was developed exploiting traces of DNA left inside 'empty' leaf mines, which are straightforward to collect and persist longer in the environment than the fly. This extends the window of possible diagnosis to at least 28 days after a leaf mine becomes empty and allows for visually indistinguishable leafmining damage caused by VLM to be genetically differentiated from that of other flies (Pirtle et al. 2020). This test reduces the need to spend considerable amounts of time searching for live material (which in many cases will not be possible, as populations on very local scales can fluctuate dramatically). Additionally, the project improved efficiency for visual surveillance programs by experimentally measuring the survey effort required to have sufficient confidence in a surveyors' ability to detect low densities of leaf mine damage, taking into account the trade-off between survey effort (walking speed), and coverage (distance) (Pirtle et al. *in preparation a*). Finally, acknowledging the high requirement for survey effort for detecting leaf mines at low densities, which can be impractical in ongoing surveillance programs that cover large geographical areas, we conducted risk predictive work to identify means to better target surveillance efforts for

more cost-efficiencies. For example, increased surveillance activities during high risk seasons (Maino et al. 2019, see 'Refereed scientific publications' for an abstract), and targeting regions with highest risk pathways for spread (Maino et al. *in preparation a,* see 'Refereed scientific publications' for an abstract).

Improvements were also made to efficiencies in incursion response times. Following the detection of SLM in Western Sydney in October 2020, project partners were able to rapidly provide scientifically rigorous information to the CCEPP. After the detection was made public, the project team compiled supporting documents within two days to distribute to the CCEPP. These documents included a draft contingency plan (see final contingency plan in Appendix 10), a draft host list, and the ESIM model outputs for Western Sydney (see Appendix 2).

Project activities undertaken in years 1 and 2 improved efficiency in targeting communication and engagement activities. During this time, project partners applied risk models in order to identify high risk areas to focus engagement efforts in year 3 of the project with the purpose of improving efficiency and relevancy. The modelling results indicated that high-risk locations should include Lockyer Valley, Mareeba/Atherton, Bowen-Gumlu, Bundaberg, Sydney, Melbourne, Devonport, Darwin, and Perth.

Finally, this project improved the efficiency of engagement. We utilised platforms that were likely to reach a wide audience. Such platforms included YouTube, Twitter, LinkedIn, and Facebook. Project-specific videos on the AUSVEG YouTube channel received a significant number of views.

Key evaluation question: Have project findings been published in peer-reviewed scientific journals?

Multiple journal articles have been produced as a result of this project. To date, one journal article has been published in a peer-reviewed journal, titled *Potential for biological control of the vegetable leafminer, Liriomyza sativae (Diptera: Agromyzidae) in Australia with parasitoid wasps*. Two others have been submitted and are currently under consideration in peer-reviewed international journals, while several others are on track to be submitted shortly. Refer to Section 'Refereed scientific publications' for further information.

Recommendations

Below we provide a number of recommendations related to VLM, ASLM and SLM.

We have also developed a list of recommendations that are specific to the recent discovery of SLM in NSW and QLD. This includes several opportunities to apply learnings and tools developed throughout this project, as well as opportunities to validate surveillance, diagnostic, and risk predictive tools. We also outline research and extension gaps that will be important to address in order to improve management outcomes for this pest.

Note: there is some overlap between the SLM incursion-specific and the broader leafminer recommendations we provide below.

General recommendations relevant to VLM, ASLM and SLM

Surveillance

- **The novel in-field diagnostics for leaf mines and flies should be integrated into existing government and industry early detection programs and future delimiting or area freedom programs:** The eDNA test for leaf mines allows rapid triaging of suspicious damage as part of early detection surveillance. Moreover, it can support efficient collection of baseline absence data for exotic leafminers preceding incursion events, as well as more efficient establishment of 'pest free areas' after incursion events.
- **Surveillance guidelines should be integrated into existing government and industry early detection programs and future delimiting or area freedom programs:** The scientifically tested survey effort guidelines developed within this project (which take into account the trade-off between search time and area coverage) should be used to ensure surveillance programs achieve acceptable levels of confidence in detection probability. These guidelines have already been shown to improve the detection success of VLM leaf mines for biosecurity officers and rangers within the TSI.
- **The surveillance review publication should serve as a standard starting point for *Liriomyza* spp. surveillance programs in Australia**: The comprehensive review of surveillance considerations for *Liriomyza* spp. in Australia (Pirtle et al. *in preparation* a, see 'Refereed scientific publications' for an abstract) should be taken as a starting point for planning all future *Liriomyza* programs across any surveillance goals (early detection, delimiting, and area freedom programs) in order to ensure consistency, effectiveness, and cost-efficiency.

Further research priorities

- **Measurement and validation of survey effort guidelines for *Liriomyza* spp. across different regions and growing systems:** The survey effort guidelines, which have been validated for VLM within urban habitats in the TSI, will require validation in different growing systems (for example, field crops versus closed cropping) upon incursion of any exotic *Liriomyza* spp. Local testing following the mock surveillance framework would allow for the development of similar recommendations that account for regional differences, other exotic leafminer species and different incursion scenarios.

Risk forecasting

- **The establishment potential and spread rate predictions should be used by government and industry to prioritise the timing and locations of early detection surveillance programs for *Liriomyza* pests:** Predicted pest seasonality allows for surveillance to target periods of highest growth potential to ensure efficient use of surveillance resources for pest detection. Predicted pest seasonality informs appropriate duration of delimiting programs. Predicted spread rates support prioritisation of highest risk entry points for early detection programs. These predictions should also be taken into account if any Pest Free Areas are to be established after an incursion of exotic *Liriomyza* spp.

- **The spread and impact risk potential predictions should be used by industry to identify production regions at highest risk of *Liriomyza* pest incursions:** Predicted spread rates allow estimates of the preparedness timeframe before production regions may be affected from various incursion locations in the absence of containment measures. This would allow preparedness programs to be established in those regions so industry can rapidly adopt necessary phytosanitary control measures in the event of incursions.
- **The establishment predictions and the life cycle predictor tool DARABUG2 should be used to inform appropriate duration and revisit frequency of any delimiting surveillance programs for *Liriomyza* pests**: Delimiting surveillance should continue at a frequency of 1 full life cycle length (which will vary with region and season), plus 1 to 2 weeks. This frequency means new leaf mines would be present during a revisit, if a population is still present in the area. To account for regional and seasonal variation in life cycles, the DARABUG2 tool can be used to calculate life stage durations for any date across Australia and is accessible at https://cesaraustralia.shinyapps.io/darabug2/. Predicted pest seasonality can further inform the appropriate duration of a delimiting program.
- **Establishment potential and seasonality predictions should guide any future awareness raising campaigns for exotic *Liriomyza* pests:** Awareness raising efforts should be targeted to highest risk regions to improve chances of early detection via general surveillance. Increased awareness should lead to more appropriate management practices, fewer outbreaks, smaller overall populations, and thus slower spread.

Further research priorities

- **Narrow the spatial resolution of the spread module of the ESIM framework**: The resolution of current simulations (9 km x 9 km grid cell and monthly timesteps) was designed to inform broad scale spread predictions. Extension of this model to finer resolutions (e.g. 50 m x 50 m) and shorter time steps (e.g. days) will better support incursion responses (e.g. trapping densities, delimitation requirements, etc.) immediately following new detections.
- **Explore cost benefits of incursion management approaches**: Explore the cost-benefit of multiple management scenarios in biosecurity response for VLM, SLM and ASLM (e.g. local and jurisdiction level quarantine, surveillance, communication and extension). This will help to ensure that the chosen interventions provide the greatest return on investment and resources are not wasted.
- **Additional sampling in PNG and Indonesia for a finer resolution picture of likely incursion pathways and source populations of VLM**: Based on the results of the RADseq analysis undertaken here, it remains unclear if PNG is the definitive pathway for the current incursion population of VLM in TSI. Increased resolution between populations from southern and eastern PNG, as well as additional Indonesian samples, particularly from Java and other islands, would enhance understandings of pathways and allow more precise pathway risk mitigation strategies.

Pest management

Growers and advisors should familiarize themselves with appropriate monitoring tools for *Liriomyza* pests: A key consideration when monitoring is that damage is cumulative and thus population declines cannot be observed by monitoring leaf mine damage on plants alone. Overseas, sticky traps and pupal trays are a simple, cheap and essential part of population monitoring. Monitoring should be prioritised around young establishing crops which are particularly vulnerable to leafminer damage, and within closed cropping where natural enemies may be scarce. Information on these techniques are available within the review of biological control options as well as the illustrated guide to key IPM considerations for exotic leafminers (see Appendix 3 and 4 and link to management guide in Appendix 8).

- **Growers and advisors should familiarize themselves with recommended *Liriomyza* specific IPM principles:** Several project outputs are designed to assist horticultural industries transition to IPM programs in the event of an incursion. This includes thorough reviews of biological, chemical options and prospects for mass rearing (see Appendix 3, 4, 5 and 7), as well as a concise illustrated guide to key IPM considerations for exotic leafminers (see link to management guide in Appendix 8). Among other management practices, growers and advisors should avoid the use of broad-spectrum insecticides in order to avoid releasing populations from natural biological control.

- **Industry should review existing chemical management practices to ensure compatibility of IPM management of *Liriomyza* pests:** Current chemical management practices in crops that are economically vulnerable to *Liriomyza* spp. in different growing regions should be reviewed to assess the likely non-target effects of these practices on future leafminer (and parasitoid) populations.

Further research priorities

- **Improved taxonomic resolution of parasitoids of Australian agromyzids likely to attack *Liriomyza* pests, along with the development of molecular markers for rapid identification of parasitoids**: These tools will greatly assist researchers assessing the impact of endemic parasitoids. Initial targets should include: (i) establishing the specific status of the Australian populations of *Diglyphus isaea*; (ii) determining if *Neochrysocharis formosa* is present in Australia and determining how many *Neochrysocharis* spp. are present; (ii) re-examining specimens of *Trigonogastrella* sp. collected in SA and Victoria to determine numbers of species; and (iv) clarifying the taxonomy and biology of the complex of opiine braconid parasitoids already found attacking agromyzids in Australia, including determining the number and host range of *Opius* spp. (see Appendix 3 and 4 for more details). Voucher specimens of all collections should be lodged in recognised insect collections.
- **Determining what agromyzid species in horticultural regions will act as reservoirs of parasitoids for *Liriomyza* pests:** Parasitism of a range of agromyzid species in major horticultural regions should be assessed to determine parasitoid assemblages at various times of the year. Targeted agromyzids should include: (i) *Cerodontha* spp. (grass sheath miners), which are widespread but little studied in Australia; (ii) the highly polyphagous *Tropicomyia polyphyta* in QLD and NSW; (iii) *Phytoliriomyza praecellens* and *Phytoliriomyza mollis* infesting *Rhagodia candolleana* and *Rhagodia parabolica* in southern Australian coastal areas; (iv) *Calycomyza lantanae*, *Ophiomyia lantanae* and *O. camarae*, which have been released and established against lantana in coastal QLD and NSW; (v) *Phytoliriomyza jacarandae* attacking widespread ornamental jacaranda trees, a nontarget agromyzid that is likely to be a parasitoid reservoir in horticultural regions; and (vi) *Ophiomya alysicarpi*, which infests *Alysicarpus* spp. and *Desmodium* spp. in the CYP and TSI. (see Appendix 3 and 4 for more details)
- **Understanding the biology, ecology and distribution of endemic parasitoids in Australia for use in biological control:** Research is warranted on *Zagrammosoma latilineatum*, *Closterocerus mirabilis*, *Trigonogastrella parasitica*, and *Opius cinerariae* to assess their potential role in controlling *Liriomyza* pests in open-air production as well as their potential suitability for augmentative biological control. The immature development and life history traits of endemic species such as *Z. latilineatum* and *C. mirabilis* should be compared to the Australian populations of *D. isaea*, to assess their suitability for augmentative biological control. Any female-only strains of parasitoids found in the field parasitising agromyzids should be evaluated and characterised for endosymbionts, due to implications for mass rearing protocols (see Appendix 3 and 4 for more details).
- **Investigate the insecticide resistance status of *Liriomyza* pests**: Understanding the insecticide resistance status of Australian populations of VLM and SLM would be highly valuable. In the absence of established genetic diagnostics, the most likely approach to determine resistance is to conduct laboratory phenotypic bioassays. Given the present (restricted) location of VLM in Australia, and in the absence of a known 'insecticide-susceptible' population, this is challenging. There will be greater opportunities for phenotypic testing of Australian SLM populations.
- **Apply for a minor use permit for indoxacarb:** A minor use permit is warranted for the control of *Liriomyza* spp. in brassica, fruiting vegetables and potentially leafy vegetable crops (pending availability of supporting data). The recommended rates for VLM control falls within the current labelled use of indoxacarb in the brassica and fruiting vegetable crops, and the recommended rates for VLM control falls within twice the current labelled use of indoxacarb in the leafy vegetable crops.

Communication and engagement

- **Availability and awareness of project extension package:** To assist with project legacy, a project extension package has been developed. Furthermore, industry extension officers, industry bodies, and

state government agencies have been trained how to use this resource. It will be freely available via https://tinyurl.com/ExoticLeafminerPackage until 1 December 2021. It is recommended this resource is made accessible via the Hort Innovation website following this date.

- **Integration of project extension package into VegNet activities:** We worked closely with VegNET extension officers during this project. VegNET extension officers are thus aware of the project and have been provided a link to the project extension package. It is recommended that VegNET extension officers use this resource on an ongoing basis to extend important information about leafminers to industry.

Further communication and engagement priorities

- **Biosecurity communication and engagement in CYP and TSI:** It would be valuable to develop biosecurity awareness training modules and associated materials for specific use by tourism and travel companies that cater to CYP and TSI. Human movement into and out of the CYP presents a high risk for the dispersal of VLM from TSI into key production regions, particularly in QLD.
- **Future biosecurity preparedness projects:** MT16004 was a cross-industry program that bought together researchers, extension experts and biosecurity officers. As a result, we were able to effectively prioritise research needs, evolve the direction of the project and communicate our messages authoritatively to a wide range of audiences (i.e. vegetable, potato, melon, nursery growers/industry). It is our recommendation that future preparedness projects follow a similar cross-industry funded and multi-disciplinary delivery model.
- **Closer linkages with local government:** In the future, it will be increasingly important for biosecurity-related projects to work with local councils across Australia. In QLD, local councils have a responsibility to develop biosecurity plans for their regions. While these biosecurity plans have a wide focus on weeds and pest animals rather than pests and diseases, future projects should leverage this requirement to work with councils and strengthen biosecurity awareness and knowledge at a local level, particularly to increase the availability of information on plant biosecurity
- **Translation of project outputs into local languages**: In future projects, it would be valuable to consider translation of community extension materials into the local languages in order to increase community awareness and uptake.

Recommendations for SLM incursion response and management in Australia

- **The surveillance and diagnostic techniques developed within this project should be used to delimit the extent of the SLM incursion and identify the realised host range:** The eDNA test for leaf mines (Pirtle et al. 2020) will allow a comprehensive investigation of the range of weed and native plant species currently affected by SLM. Leaf mines should be collected from an abundance of species, suspected hosts or not, to collect absence data for leaf mines associated with native or naturalised leafminer species. The surveillance guidelines (see surveillance guide linked in Appendix 8, and more information within the 'Refereed scientific publications') will ensure high confidence that low level infestations of SLM are detected.
- **The establishment and spread predictions developed within this project should be used to target SLM surveillance and extension efforts**: Despite the CCEPP decision that SLM in not feasible to eradicate, effective delimiting of current infestations and early detection of new outbreaks is still important to ensure early interventions before populations grow to large numbers, driving further spread. Continued awareness raising, targeting regions predicted to be most affected by SLM, will be important to encourage early reporting of new outbreaks and appropriate management interventions.
- **The DARABUG2 life cycle predictor tool could support SLM surveillance and management:** DARABUG2's regionally and seasonally specific life cycle duration predictions will support surveillance, by determining surveillance program parameters such as revisit frequency, as well as management, by giving indications of the window of time for successful intervention when small populations of SLM are discovered.

- **Rotations between different chemical MoAs:** While it is currently unclear if Australian populations of SLM have resistance to insecticides, this species is known to evolve resistance overseas. This project has secured chemical permits that reflect a range of MoAs. Growers should immediately follow resistance management guidelines for SLM, including the rotation of chemical MoAs (this should consider all chemical use in the system, not just chemicals directed at SLM).

Future research and extension priorities

- **Develop an interactive portal to explore regional and seasonal risk:** This would build on the VLM portal and the predictive work already completed for SLM as part of this project (Pirtle et al. *in preparation b,* see 'Refereed scientific publications' for an abstract) to improve communication of risks to industry and predicted pest seasonality for targeted monitoring and management.
- **Validating leafminer survey effort guidelines:** Survey effort guidelines were developed for farm situations, and validated against VLM populations within the TSI under urban situations. These guidelines should be validated against SLM in realistic paddock settings, using low density infestations where present. This information will have benefits across all pest *Liriomyza* species.
- **Validating in-field diagnostics**: In-field molecular tests have been developed for larval/pupal/adult SLM, as well as empty leaf mines. Validations of these in-field tests should be conducted using SLM specimens and leaf mines collected from affected regions in NSW and QLD.
- **Weed reservoir surveys:** Surveillance should be conducted to identify weed and native plant species, which will provide important non-crop reservoirs for SLM at different times of the year. As there are many native or naturalised leafminer flies already present in Australia and common among weeds, the eDNA test for leaf mine damage (see point above) would be a valuable tool.
- **Parasitoid surveys:** Surveillance should be conducted to determine the extent of natural control of SLM within new incursions, and identify important parasitoid wasp species. This could be done morphologically and/or with DNA barcodes already established in our project. It will be important to conduct surveys across the entire known range of SLM and across several points in the season, as parasitoid dynamics are expected to vary greatly with location and season (Ridland et al. 2020; Coquilleau *in review*). Moreover, surveys across multiple years will be important to capture longer-term variations in parasitoid dynamics.
- **Collecting fine scale spread data:** There have been very few overseas studies on the spread dynamics of early incursions of pest *Liriomyza*. This incursion presents an opportunity to collect and utilise valuable spread data to validate coarser resolution (9km x 9km) spread potential predictions made via the ESIM framework. Additionally, it would allow the model to be adapted to create finer scale resolution predictions (e.g. 50 m x 50 m) to further advise incursion responses (e.g. trapping densities, delimitation requirements, etc.).
- **Seasonal forecasts:** The biological information, eco-climatic data already compiled by the project team, and subsequently developed model of establishment risk, can be used to conduct seasonal pest risk forecasting for industries based on present climatic conditions in the current growing season. This would help to highlight seasonal weather conditions that are favourable to population growth and pest outbreaks.
- **Chemical usage and permits:** SLM permits are in place covering multiple MoAs on most affected crop groups, however additional permits may be required to ensure appropriate control options are available to growers of affected crops. The following minor use permits are considered highest priority for SLM: (i) Cyromazine in the following vegetable crops: Broccoli, Fruiting vegetables – cucurbits, Fruiting vegetables – other than cucurbits (excluding mushrooms and corn), Head lettuce, Legume vegetables, Root and tuber vegetables, Stalk and stem vegetables; (ii) Abamectin in the following vegetable crops: Fruiting vegetables, including cucurbits (excl. sweet corn and mushrooms), Leafy vegetables (excl. lettuce), Legume vegetables, Root and tuber vegetables, Bulb onions, Cabbage (head), Celery, Rhubarb, Bulb vegetables (incl. leeks and spring onions). Linked to this, there is a need to understand what chemicals growers are presently using and how this can be nuanced to ensure appropriate management programs are being employed. A demonstration site could prove to be quite powerful in this regard.

- **Chemical baseline data and resistance, and development of resistance management strategies:** Understanding the insecticide resistance status of Australian populations of SLM will be key to ongoing management. In addition to determining any pre-existing resistances of the SLM incursion population, there is a need to generate baseline chemical sensitivity data for key insecticides which will be needed to monitor for future resistance evolution in the field. In the absence of established genetic diagnostics, the most likely approach to determine resistance is to conduct laboratory phenotypic bioassays. Resistance status should also be used to inform the creation of resistance management strategies that rely upon MoA rotations and IPM practices.
- **Update *Liriomyza* management plan for SLM:** Once key information about SLM management under Australian conditions is obtained, the *Liriomyza* management plan developed through this project (see link in Appendix 8) should be revised specifically for SLM. This may include updating information on registered chemical use permits, insecticide resistances, biological control options, monitoring techniques (including the use of sticky trap and pupal tray techniques and techniques for monitoring parasitism rates) and the application of Australian specific economic thresholds.
- **RADseq pathway analysis:** A library of global SLM samples should be compiled to determine the likely entry pathway (and identify the likely number of incursion events) for the SLM populations present in NSW and QLD. This analysis would follow a similar approach to one established for the Seisia/TSI populations of VLM, allowing connectedness between populations to be established along with sources of incursions (Xu et al. *in preparation*). The project team already hold specimens from Kenya and Indonesia and have connections which can be leveraged to source additional populations such as Malaysia and China. SLM population samples from NSW and QLD should be preserved in absolute alcohol and stored appropriately for future genetic work. Samples collected as part of DAWE border interceptions (while not providing as precise of origin locational data as specimens collected by overseas collaborators) should also be leveraged to expand the geographic coverage of the sample library.
- **Development of economic thresholds and validation of monitoring methods (such as sticky traps and pupal trays)**: Thresholds for pupa counts within trays, and sticky traps within field crops and closed cropping have not been developed within Australia for exotic *Liriomyza* pests. These techniques should be validated for SLM and thresholds developed. Existing work on parasitoid biology and population modelling could be extended to develop thresholds for chemical pest management (i.e. when parasitoids present are insufficient for pest control and pesticide applications become warranted).
- **Ongoing awareness of IPM options:** As demonstrated through this project, ongoing awareness raising activities will be critical to ensure uptake of management practices against SLM in Australia. This campaign could include face to face workshops, delivery of webinars, and the development and distribution of industry communiques, all of which were an integral part of the communication and engagement strategy used in this project.

Refereed scientific publications

One paper has been published in a peer-reviewed journal. Several others have been submitted (and are currently under review) or are in preparation. Details are provided below.

Published

Potential for biological control of the vegetable leafminer, *Liriomyza sativae* (Diptera: Agromyzidae), in Australia with parasitoid wasps

Ridland, P. M., Umina, P. A., Pirtle, E. I., & Hoffmann, A. A. (2020). Potential for biological control of the vegetable leafminer, *Liriomyza sativae* (Diptera: Agromyzidae), in Australia with parasitoid wasps. *Austral Entomology*, 59(1), 16-36.

Abstract: The vegetable leafminer, *Liriomyza sativae* Blanchard, poses a risk to vegetable and nursery production in mainland Australia since established in Cape York in 2015. Effective control overseas depends on maximising the impact of natural enemies. Problems with polyphagous *Liriomyza* pest species typically result from the destruction of their parasitoids by excessive use of non-selective insecticides. Field studies are reviewed to identify parasitoid species involved in the biological control of *L. sativae* in open-air and glasshouse production internationally and to assess the current knowledge of parasitoids of agromyzids in Australia. Overseas, invading *Liriomyza* populations have frequently been exploited by endemic parasitoids (often found on non-pest agromyzid species), and non-crop hosts have played a role as reservoirs of these parasitoids. The few published Australian field studies on the occurrence of agromyzid flies and their parasitoids show a large community of wasps attacking agromyzids, with species mainly from the Eulophidae, Pteromalidae and Braconidae. The most abundant recorded species are two cosmopolitan eulophid species, *Hemiptarsenus varicornis* (Girault) and *Diglyphus isaea* (Walker), and four Australian species: two eulophid species, *Zagrammosoma latilineatum* Ubaidillah and *Closterocerus mirabilis* Edwards & La Salle, one pteromalid species, *Trigonogastrella* sp., and one braconid species, *Opius cinerariae* Fischer, for which there is little biological information. One deficiency in the known assemblage in Australia is the absence of parasitoids from the Eucoilinae (Hymenoptera: Figitidae), a subfamily with several abundant species attacking agromyzids overseas. The composition and impact of the endemic parasitoid assemblage in Australia on populations of *L. sativae* needs to be assessed adequately in the field before the importation of additional exotic parasitoid species is contemplated. Overseas, two species, *D. isaea* and *Dacnusa sibirica* Telenga, are reared commercially for augmentative biological control, although the relatively high cost of production has restricted their release to protected cropping situations. Knowledge gaps remain locally about the taxonomy, distribution, host range and life cycle of parasitoids, and their potential impact on *L. sativae*.

Under review

A molecular method for biomonitoring of an exotic plant-pest: Leafmining for environmental DNA

Pirtle, E., Van Rooyen, A., Maino, J., Weeks, A., & Umina, P. (2020). A molecular method for biomonitoring of an exotic plant-pest: Leafmining for environmental DNA. *Authorea Preprints*.

Under review with *the Journal of Molecular Ecology*

Abstract: Understanding how invasive species respond to novel environments is limited by a lack of sensitivity and throughput in conventional biomonitoring methods. Arthropods in particular are often difficult to monitor due to their small size, rapid lifecycles, and/or visual similarities with co-occurring species. This is true for the agromyzid leafminer fly, *Liriomyza sativae*, a global pest of vegetable and nursery industries that has recently established in Australia. A robust method based on environmental DNA (eDNA) was developed exploiting traces of DNA left inside 'empty' leaf mines, which are straightforward to collect and persist longer in the environment than the fly. This extends the window of possible diagnosis to at least 28 days after a leaf mine becomes empty. The test allowed for visually indistinguishable leafmining damage caused by *L. sativae* to be genetically differentiated from that of other flies. Field application resulted in the identification of new local plant hosts for *L. sativae*, including widely distributed weeds and common garden crops, with important implications for the pest's ability to spread. Moreover, the test confirmed the presence of a previously unknown population of *L. sativae* on an island in the TSI. The developed eDNA method is likely to become an important tool for *L. sativae* and other leafmining species

of biosecurity significance, which, historically, have been difficult to detect, diagnose and monitor. More generally, eDNA is emerging as a highly sensitive and labour-efficient surveillance tool for difficult to survey species to improve outcomes for agricultural industries, global health, and the environment.

Seasonality and community composition of parasitoid wasps of four agromyzid leafminer species (Diptera: Agromyzidae) in Victoria

Marianne P. Coquilleau (in review). *Seasonality and community composition of parasitoid wasps of four agromyzid leafminer species (Diptera: Agromyzidae) in Victoria* (Master's thesis, Pest and Environmental Adaptation Research Group, The University of Melbourne, Melbourne, Australia).

Abstract: The vegetable leafminer, *Liriomyza sativae* Blanchard (Diptera: Agromyzidae), was first detected in the Torres Strait islands in 2008, crossed over to the tip of Cape York in 2015 and now poses a threat to Australian crops. This species is an economically important secondary pest in many parts of the world; populations readily evolve resistance to insecticides and benefit from the use of chemical controls against other pests, which kill their natural enemies. *Liriomyza sativae* is expected to spread into Australia's agricultural production areas and it is therefore valuable to investigate the local biological agents that could contribute to future integrated pest management (IPM). I assessed the abundance and diversity of the hymenopteran parasitoid wasps of four common agromyzid flies in southern Victoria (*Liriomyza brassicae* (Riley), *Liriomyza chenopodii* (Watt), *Phytomyza plantaginis* Goureau, and *Phytomyza syngenesiae* (Hardy)) to evaluate their potential as candidates for control of *L. sativae*. I monitored six sites in Melbourne over a period of 18 months by collecting mined leaves from a range of plant species and rearing adult flies and their parasitoids. Additional sites around Victoria were also sampled to offer a snapshot of the agromyzid presence across the state. Eleven wasp species were identified in total, two of which were only classified to morphospecies level, including known parasitoids of *L. sativae* overseas and species reared from *L. sativae* populations from far north Queensland. Ninety percent of the parasitoids were eulophids (Hymenoptera: Eulophidae), with the rest consisting of opiines (Braconidae) and Pteromalinae (Pteromalidae). The adventive *Chrysocharis pubicornis* (Zetterstedt) was the most abundant parasitoid, reared almost entirely from *Phytomyza* hosts, followed by an *Asecodes* sp. and the introduced *Diglyphus isaea* (Walker). We recorded the first male *Ph. plantaginis* for Australia. Males were only found at Melbourne locations and these populations tended to have a female-biased sex ratio. Variation in sex ratio may be host plant dependent, as the female:male ratio was 4:1 from the host *Plantago lanceolata* L. while it was 1:1 in more limited samples from *Plantago major* L. The four common local agromyzids reached peak abundance at different times of the year and together supported a stable community of both adventive and native parasitoids. Of the species reared, several are known to attack *L. sativae* including *D. isaea, Hemiptarsenus varicornis* (Girault) and *Neochrysocharis formosa* (Westwood). The wasps reared include candidates for augmentative or conservation biological control that should be further considered in the event that *L. sativae* becomes a widespread pest of Australian crops.

In preparation

Forecasting the potential distribution of the invasive vegetable leafminer using 'top-down' and 'bottom-up' models.

Maino, J. L., Pirtle, E. I., Ridland, P., & Umina, P. A. (2019). Forecasting the potential distribution of the invasive vegetable leafminer using" top-down" and" bottom-up" models. bioRxiv, 866996.

Abstract: The vegetable leafminer, *Liriomyza sativae*, is an internationally significant pest of vegetable and flower crops, that was detected for the first time on the Australian mainland in 2015. Due to the early stage of its invasion in Australia, it is unclear how climatic conditions are likely to support and potentially restrict the distribution of *L. sativae* as it expands into a novel range and threatens agricultural production regions. Here we predicted the future establishment potential of *L. sativae* in Australia, using both a novel 'bottom-up' process-based model and a popular 'top-down' correlative species distribution model (SDM), leveraging the unique strengths of each approach. Newly compiled global distribution data spanning 42 countries was used to validate the process-based model of establishment potential based on intrinsic population growth rates, as well as parameterise the correlative SDM. Both modelling approaches successfully captured the international distribution of *L. sativae* based on environmental variables and predicted the high suitability of non-occupied ranges, including northern regions of Australia. The largely unfilled climatic niche available to *L. sativae* in Australia demonstrates the early stage of its Australian invasion, and highlights locations where important vegetable and nursery production regions in Australia are highly vulnerable to *L. sativae* establishment.

Modelling population growth and dispersal to estimate accumulating impacts of exotic leafminers

Maino, J. L, Pirtle, E. I., Ridland, P., Schouten, R, & Umina, P. (*in preparation a*)

Abstract: Biosecurity capacity for early detection, containment and eradication is limited by the poor predictability of pathways by which pests are likely to spread, establish, and impact industry. Vegetable leafminer (Liriomyza *sativae*, VLM) is an international pest of vegetable crops that has recently established in Australia for the first time. VLM poses a risk to Australia's significant horticultural production and subsequent access to international markets, the potential impact of which has yet to be estimated. Here we develop an ecological approach to estimating gridded impacts based on climate driven population growth within grid cells and human assisted dispersal between grid cells. Gridded annual production values of affected horticultural crops are then overlaid onto the spatial distribution of the invading pest population through time. Findings of the simulation suggest that the potential impacts of VLM in Australia are substantial, particularly for vegetable and nursery production across Australia's east coast. As is frequently observed with other invading animals, the accumulation of economic impacts accelerates with time, highlighting the importance of early intervention (e.g. industry preparedness and awareness). Through repeating the simulation with different settings, certain entry points were identified that resulted in faster accruing impacts, which may form a means to prioritise any surveillance and mitigation activities.

Integrating predator-prey dynamics with mechanistic species distribution models to forecast biocontrol potential for an invading pest

Maino, J. L., Pirtle, E. I., Ridland, P., Lye, J. C. & Umina, P. A. (*in preparation b*).

Abstract: When arthropod pests expand into new locations, they will encounter novel habitats, food sources, and predators and parasitoids. These mitigating factors, which may limit the ability to establish, and are therefore of interest to agricultural industries and the associated supply chain likely to be impacted by invading species. While abiotic factors such as climate are routinely considered in forecasts of the establishment risk of exotic pests, far fewer studies have explored the impact of natural and augmented biological pest suppression in novel ranges. Here, using a novel approach based on population growth rates and predator-prey dynamics, we investigate the ability of two ecologically distinct parasitoid wasps to mitigate impacts of a newly arrived pest in Australia, the vegetable leafminer, *Liriomyza sativae* (Blanchard). Life-history data was compiled on *L. sativae* and two idiobiont eulophid parasitoid wasps, *Diglyphus isaea* (Walker) and *Hemiptarsenus varicornis* (Girault), to estimate population growth responses of both pest and parasitoids to climate across regions and seasons. These population growth responses were then linked using a Holling type II Lotka-Volterra predator-prey model. Results show that the climatic suitability for the two natively occurring parasitoids overlaps considerably with *L. sativae*, highlighting the potential natural biocontrol for this pest as it moves into new regions. Both region and season impacted the efficacy of biological control, including the time to pest-suppression and which parasitoid species conferred optimal control. This study provides a novel ecological approach to dynamically estimate mitigating biotic and abiotic factors during the invasion of an exotic species into a novel range.

Towards a technical basis for surveillance of *Liriomyza sativae, L. trifolii,* and *L. huidobrensis* (Diptera: Agromyzidae) in Australia

Pirtle, E. I., Ridland, P., Burgess, R., Taylor, S., Maino, J. L. & Umina, P. A. (*in preparation a*).

Abstract: The availability of standardized and scientifically robust surveillance guidelines is paramount to reducing economic impacts associated with biosecurity incursions. Surveillance is required across the plant biosecurity continuum from early detections during new incursions to demonstrating areas of pest freedom to support trade, to ongoing pest management once a pest is widely established. Each application will have different goals and thus different surveillance requirements. There is a range of considerations in designing such surveillance programs, including the risk profile of the pest, (establishment, spread and impact risk across region and season), aspects of biology and ecology that will influence the appropriate surveillance methods to employ, and the detectability of the target and the resulting level of survey effort required to achieve sufficient confidence of detection. Polyphagous *Liriomyza* leafminers represent a major risk to horticulture globally. These species are not yet established within Australian agricultural regions, however one species, *L. sativae*, has recently established in an isolated area in the far north of Queensland. This review consolidates experimental, statistical, and predictive work to create reference material for the development of technically justified and cost-effective surveillance programs

within Australia for exotic *Liriomyza*. This work will serve as a reference point for the development of surveillance plans for exotic *Liriomyza* within Australia.

Using mock surveillance to quantify exotic pest detectability prior to establishment.

Pirtle, E. I., Umina, P. A., Hauser, C. E. & Maino, J. L. (*in preparation b*).

Abstract: Quantifying detectability of exotic pests in local contexts is crucial for designing efficient and regionally relevant surveillance guidelines. Ironically, for areas most in need of preparedness advice, the study of detectability is hampered by the pest's absence. Here, we present a mock surveillance method whereby pest symptoms are simulated on plants, and the detection probability of visual inspection by practitioners is measured across a range of high priority regions at risk of incursions. Detectability of simulated leaf mining damage was found to be no different to real damage in surveillance trials conducted on the vegetable leafminer, *Liriomyza sativae*, a recently established pest in Australia. Additional trials confirmed that reducing the natural search speed of survey participants by half increased the odds of detecting a leaf mine by an estimated 3.0 times. However, as movement speed mediates a trade-off between the total area covered (the number of pest encounters) and sensitivity (detection probability for each encounter), we found that a survey effort of 10 s/m enhanced field-level detection of *L. sativae* at a variety of abundances, and available times for surveillance. This study demonstrates that mock surveillance trials, through their use of visual effects to mimic plant pest symptoms, can produce locally tested recommendations to enhance preparedness for exotic pests.

Extending a multiplex quantitative PCR assay for the detection of key biosecurity leaf mining pest species

van Rooyen, A., Pirtle, E. I., Ridland, P., Umina, P. A., & Weeks, A. R. (*in preparation*).

Abstract: The Agromyzidae are a group of small flies whose larvae feed internally on plants, often as leaf and stem miners, with a high level of host specificity. However, five species of Agromyzidae: *Liriomyza sativae, L. trifolii, L. huidobrensis, L. bryoniae* and *L. chinensis*, are highly polyphagous, known as serious agricultural pests and therefore considered biosecurity threats to Australia's plant-based agricultural industries. Surveillance and diagnosis of these pests can be difficult due to their small size, rapid lifecycle and visual similarities between species and the damage they inflict on plants. Molecular diagnostic quantitative PCR (qPCR) assays have been developed to differentiate between *L. sativae, L. trifolii* and *L. huidobrensis*. Similarly, environmental DNA (eDNA) methods have recently been developed for the vegetable leafminer*, L. sativae*, increasing the window of detectability through DNA left in leaf mines, providing a more sensitive and cost-efficient monitoring and surveillance technique for detecting *L. sativae*. Here we extend the molecular diagnostic assays to three other Agromyzid species, *Liriomyza bryoniae, Liriomyza chinensis* and *Liriomyza brassicae*. We demonstrate the specificity of the assays for all six Agromyzid species on a range of non-target leaf miner species collected from nine different countries and demonstrate the specificity of each assay. Finally, we test the utility of the assay combined with a simple extraction procedure as a rapid field diagnostic using the portable Franklin™ quantitative PCR machine to identify larval, pupal and leaf mine samples. We discuss the application of these assays and the rapid infield diagnostic test for improving surveillance activities and management responses for these biosecurity threats.

Genome-wide SNPs of vegetable leafminer, *Liriomyza sativae*: insights into the recent Australian invasion

Xu, X., Qiong, Y., Ridland, P., Umina, P.A., & Hoffmann, A. (*in preparation*).

Abstract: *Liriomyza sativae*, the vegetable leafminer, is an economic pest originally from the Americas which has now colonized all continents except Antarctica during the last ~30–40 years. It has been recorded from nine plant families, although its preferred hosts tend to be in the Cucurbitaceae, Fabaceae and Solanaceae families. In 2015, *L. sativae* landed on the Australian mainland and established in Seisia, posing a threat to Australian agriculture and horticulture. In this study, using double-digest Restriction Site-Associated DNA (ddRAD) sequencing, we developed a panel of 5095 single nucleotide polymorphisms (SNPs) based on 25 worldwide *Liriomyza sativae* populations. We used this genomically-based approach to study the genetic structure and the differentiation of *Liriomyza sativae* populations and to the understand origin(s) and connections of the recent invasions into Australia. Our principal component analyses (PCA) indicate the existence of two major genetically differentiated population clusters; one consists of populations from Hawaii and the other includes the rest of the invasive populations. An additional PCA

analysis without Hawaii populations demonstrated that populations from Australia including Torres Strait islands (Thursday Island, Boigu Island, Masig islands) and northern Cape York Peninsula (Seisia) are genetically close and appear to have connections to Bali, suggesting that regions around Indonesia may represent a probable source of *Liriomyza sativae* detected in Australia, whereas populations from Asia and Africa were more distantly related. The Seisia and Thursday Island populations are genetically indistinguishable. Isolation-by-distance (IBD) was established based on a significant Mantel test. Our results suggest a gradual expansion of *L. sativae* across the region and into the mainland although our analyses were limited by sample availability and more samples from the region as well as the Americas should be tested.

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Xu, X., Qiong, Y., Ridland, P., Umina, P., & Hoffmann, A. (*in preparation*). Genome-wide SNPs of vegetable leafminer, Liriomyza sativae: insights into the recent Australian invasion.

Intellectual property, commercialisation and confidentiality

No IP, commercialisation or confidentiality issues to report

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Appendices

Appendix 1. Wind-borne dispersal potential of the vegetable leafminer (*Liriomyza sativae*)

Appendix 2. Summary of ESIM (Establishment, Spread, Impact and Management) model outputs for *L. huidobrensis* in Western Sydney

Appendix 3. Prospects for biological control of the vegetable leafminer*, Liriomyza sativae*, in Australia

Appendix 4. Parasitoids of the polyphagous agromyzid leafmining flies, *Liriomyza huidobrensis, L. trifolii* and *L. sativae* (Diptera: Agromyzidae)

Appendix 5. Prospects for mass-rearing parasitoids to enable augmentative biological control of polyphagous agromyzid leafminers (*Liriomyza* spp.) in Australia

Appendix 6. Agromyzid leafminers, their hosts, and their parasitoid wasps within the Torres Strait Islands and Cape York Peninsula, with a focus on vegetable leafminer (*Liriomyza sativae*)

Appendix 7. Review of chemical management options for polyphagous *Liriomyza* spp. in Australian horticultural crops

Appendix 8. C&E Outputs developed throughout the project

Appendix 9. MT16004: Cape York & Torres Strait Biosecurity: a scoping study undertaken on behalf of the Australian vegetable and nursery & garden industries

Appendix 10. Contingency Plan: Serpentine leafminer (*Liriomyza huidobrensis*)

Appendix 11. Contingency Plan: Vegetable leafminer (*Liriomyza sativae*)

Appendix 12. Contingency Plan: American serpentine leafminer (*Liriomyza trifolii*)

Appendix 13. Monitoring and evaluation data

Appendix 14. Communications and engagement update following Covid-19

Appendix 1: Wind-borne dispersal potential of the vegetable leafminer (*Liriomyza sativae*)

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Contents

Key Points 2
Background 3
Methods 3
Results 4
Discussion 4
References 5

Key Points

- Simulation modelling was performed to assess the potential of wind to contribute to further spread of vegetable leafminer in Australia
- Wind-assisted spread from established populations in Far North Queensland was predicted to mostly deposit dispersing populations off-shore.
- While it is plausible that wind could facilitate Indonesian populations of leafminer in dispersing to the Northern Territory, the likelihood is low.
- Given the lack of evidence that wind will be a major contributor to the spread of vegetable leafminer under present conditions, human-assisted dispersal pathways should be prioritised.

Background

There is a growing appreciation of the biosecurity risks posed by the rapidly increasing domestic and international flow of goods and people as economies increase in scale and connectivity (Wichmann et al., 2009). Within Australia, a list of 40 'high risk' biosecurity species has recently been identified by government (Department of Agriculture and Water Resources, 2016), which includes several species of Agromyzidae flies, including the vegetable leafminer (*Liriomyza sativae*), the American serpentine leafminer (*Liriomyza trifolii*), the Serpentine leafminer (*Liriomyza huidobrensis*) and the tomato leafminer (*Liriomyza bryoniae).* These species were previously identified as high risk to Australian horticulture (Jovicich, 2009). In 2008, *L. sativae* was detected for the first time in the north Australian islands of Torres Strait (Blacket, Rice, Semeraro, & Malipatil, 2015), and then on the Australian mainland at Seisia in 2015 (IPPC, 2017). The pest has not yet been detected in any other regions of Australia despite ongoing surveillance efforts. As a highly polyphagous insect capable of infesting a broad range of vegetable and flower crops, *L. sativae* represents a significant threat to the production value of vegetables (valued at $3.9b in 2016/17) and nurseries (valued at $1.6b) (ABS, 2018). The majority of *L. sativae* damage occurs during larval feeding between the upper and lower leaf surface, which curtails photosynthetic ability and reduces marketability of some crops (Parrella, 1987). *L. sativae* is also prone to evolving insecticide resistance, making control and eradication difficult (Mason, Johnson, & Tabashnik, 1987). Overseas, where *L. sativae* is established in production areas, insecticide-based control of *L. sativae* has disrupted beneficial predators and parasitoids leading to secondary outbreaks of *L. sativae* (Parrella, 1987). Keeping Australian vegetable production areas free of trade-sensitive plant pests is critical, particularly with industry's increased focus on international export and current reliance on interstate trade.

The role of wind-assisted, long-distance dispersal remains an issue of concern to the future spread and establishment of vegetable leafminer in Australia. Here, using a dispersal model driven by atmospheric data, we explore the potential role of wind-assisted dispersal in transporting vegetable leafminer. Specifically, we answer two questions: what is the capacity of vegetable leafminer to disperse from its current Australian range in Far North Queensland; what risk do neighbouring island countries pose for incursions of vegetable leafminer in the Northern Territory.

Methods

The Hybrid Single Particle Lagrangian Integrated Trajectory Model (HYSPLIT) is a computer model that is used to compute air parcel trajectories and deposition or dispersion of atmospheric pollutants developed by NOAA and Australia's Bureau of Meteorology (Draxler & Hess, 1998).

The climatic data driving the model was supplied by NCEP/NCAR Reanalysis project which is a state-of-the-art analysis/forecast system to perform data assimilation using past data from 1948 to the present (Kalnay, Kanamitsu, Kistler, & others, 1996).

An interactive interface for the HYPSLIT particle dispersion model called TAPPAS was developed by CSIRO (Durr et al. 2015) and utilised in the following simulations. To explore windborne dispersal potential from current range of *L. sativae* in Far North Queensland a forward simulation from Seisia was conducted. In contrast, a backwards simulation was conducted to explore possible source locations for a hypothetical windborne incursion into the Northern Territory where the destination location was taken as Darwin. A forward direction of simulation is used to model where the pest might be dispersed to from a known or suspected source, while backwards simulation is used to model where the pest may have come from.

Default model parameters for a small dipteran insect were used in the simulation. Specifically, the properties of the dispersing particle (i.e. a vegetable leafminer adult) was assumed to have 1 mm diameter, a density of 1 g/cm^3. Each simulation ran for 23 hours, where 10,000 individuals were released each hour at a height 1 m above ground in the first three hours and were able to survive in the air column for a maximum of 20 hours.

Results

The simulations considered here suggest that vegetable leafminer dispersing via wind from their current Far North Queensland range in Seisia are unlikely to contribute significantly to further southward dispersal events (Figure 1A). The majority of long-distance deposition occurred in the north-west of Seisia reflecting prevailing wind patterns.

The backwards simulation estimating source population of a hypothetical windborne incursion in Darwin suggest vegetable leafminer could have been blown within a 20 hour period from as far west as Rote Island, Indonesia (Figure 1B). However, attention should be paid to the logarithmic scale of the estimated concentration across aggregated simulations, which spans several orders of magnitude.

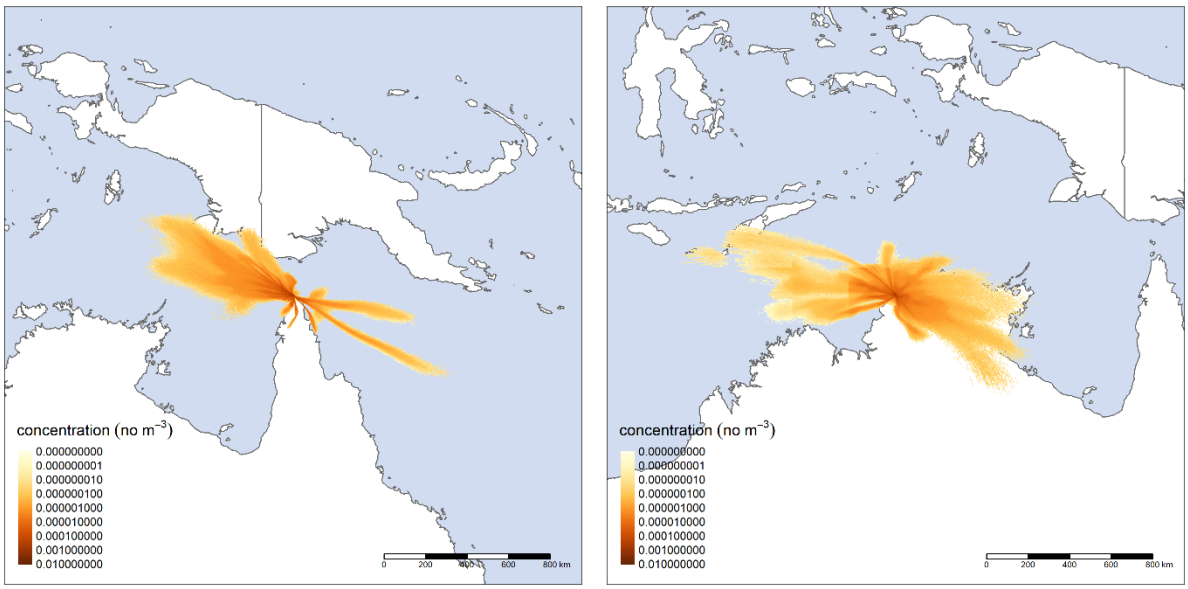


Figure 1. HYSPLIT windborne dispersal simulation results summed across all weekly simulations across the year. Forward simulations were performed assuming a source population of vegetable leafminers at Seisia to estimate the deposition patterns throughout the year. A backwards simulation was performed for Darwin to estimate potential source populations of incursion events in the Northern Territory.

Discussion

Due to the lack of evidence that windborne dispersal is a significant contributor to leafminer dispersal in Australia, human-mediated pathways should be prioritised, particularly given the accumulation of research pointing to this as a major contributor to the movement of invasive species (Capinha, Essl, Seebens, Moser, & Pereira, 2015; Wichmann et al., 2009). Such pathways include transport of contaminated produce, or “hitch-hiking” adults associated with the large volume of human movement, particularly during the tourist season in Far North Queensland.

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Appendix 2: Summary of ESIM (Establishment, Spread, Impact and Management) model outputs for *L. huidobrensis* in Western Sydney

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Introduction

To further develop our understanding of the risk profiles of the exotic *Liriomyza*, a novel, spatially-explicit simulation framework, called the Establishment, Spread, and Impact Management framework (ESIM) was developed and applied to create establishment, spread and impact risk predictions for *Liriomyza sativae, L. trifolii, and L. huidobrensis* in Australia (Maino et al *unpublished*). The framework includes modules for population establishment, growth, and spread, which are overlaid onto the spatial distribution of susceptible horticultural productivity to estimate economic impacts through time under various incursion scenarios.

In this exercise, we followed four steps: 1) we predicted the suitability of climate for *L. huidobrensis* in Australia, 2) we collated production values for at-risk crops in Australia, 3) we then predicted the risk of VLM spreading through production regions, 4) we estimated the economic cost to industry based on the potential for *L. huidobrensis* to establish and spread through at risk commodities

In the ESIM model we can vary the following parameters:

- climatic conditions
- pest sensitivity to climate
- incursion entry point
- time of arrival (e.g. autumn or spring)
- size of initial population
- local spread rates
- human-assisted spread rates
- flexibility to include other processes if required

The current limitations include

- a coarse resolution (9 km grid size and monthly time steps)
- a lack of spread rate data for *L. huidobrensis* (so we utilize D. suzukii spread data for model parametrization, due to it being a similarly small fly)
- an assumption that field temperature responses follow the temperature response of population growth rate as measured by Mujica et al (2016).

The results of these predictions have implications for better targeting the location and timing of surveillance programs across a range of surveillance goals, and these are highlighted in italics under each output description.

Currently available ESIM outputs:

L. huidobrensis establishment potential predictions

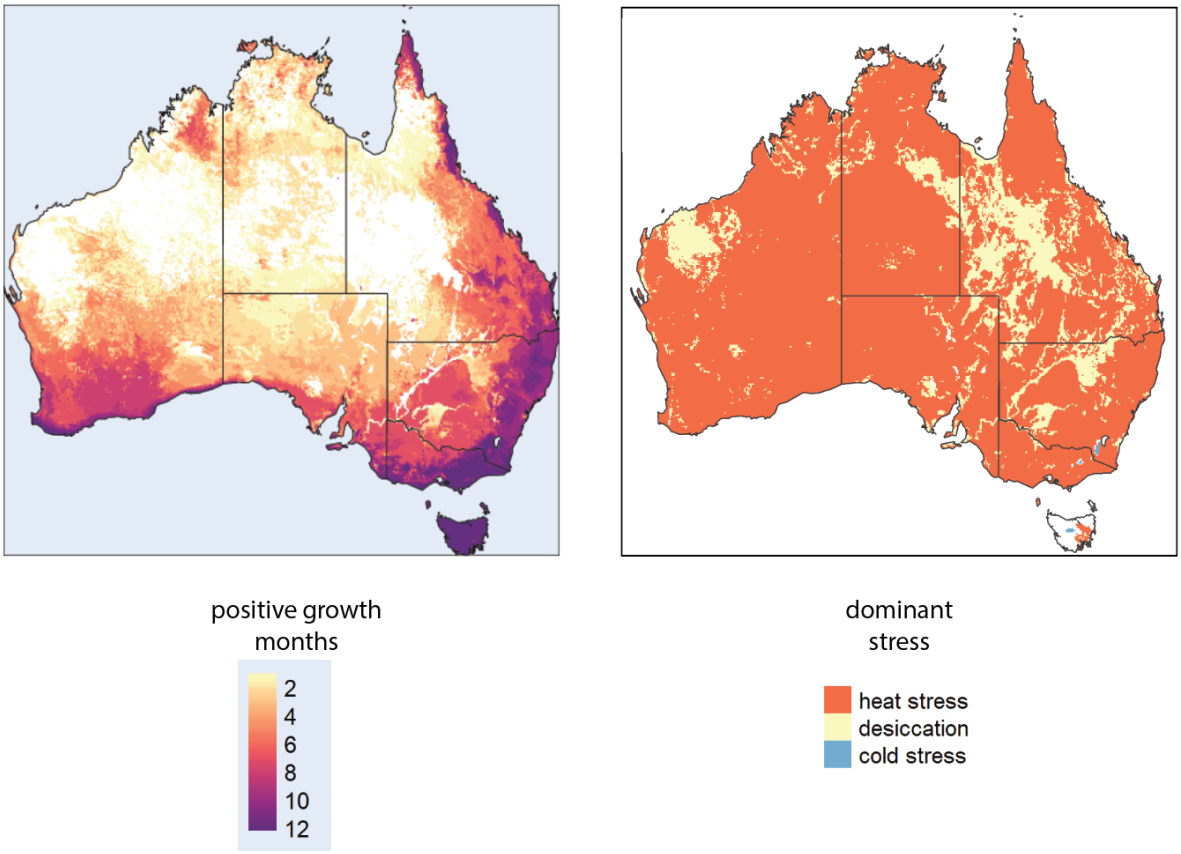


Figure 1. Aggregated yearly establishment potential of *L. huidobrensis* expressed as the percent of months across the year with possible activity based on temperature and moisture constraints, and predicted dominant stressors based on the highest mortality rate from desiccation, cold, and heat stress across the year, using the ESIM framework.

Table 1. Expected seasonality of *L. huidobrensis* across the year at Sydney and Western Sydney. Green means highest establishment risk (where the population growth potential was greater than 51% of the maximum predicted growth rate) yellow means lower establishment risk (where the population growth potential was between 1 and 50% of the maximum), and red means lowest establishment risk (where predicted growth rates are negative, meaning populations are predicted to decline).

Growing region (central point)	**Seasonality**											
	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Sydney, NSW												
Western Sydney, NSW												

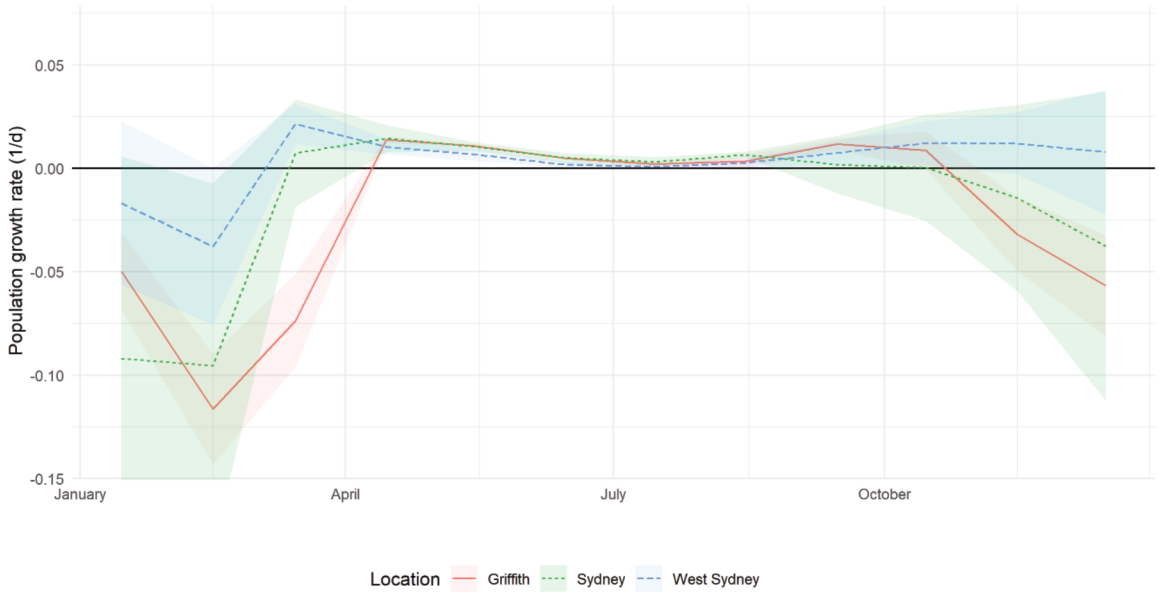


Figure 2. Predicted population growth rates for *L. huidobrensis* at three growing regions within NSW.

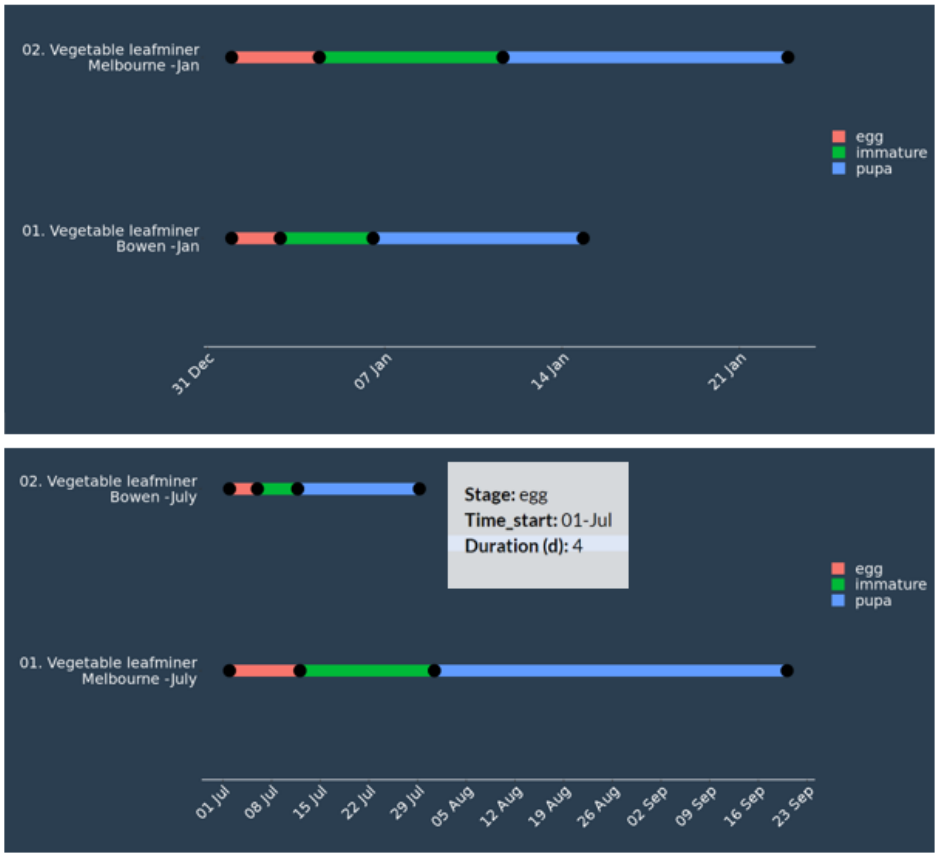


Figure 3. A screenshot of DARABUG2, a life-stage development web app. Using the life-stage developmental response of temperature as measured by Mujica et al (2016), we incorporated a life-stage development model for *L. huidobrensis* into a simple web interface that allows users to predict the duration of various life stages under different regional climates. App can be accessed at https://cesaraustralia.shinyapps.io/darabug2/.

Applications of establishment prediction outputs:

- *Predicted pest seasonality allows for surveillance to be timed to periods of highest growth potential to yield highest probability of pest detection*
- *Predicted pest seasonality informs appropriate duration of delimiting program*
- *Predicted pest seasonality allows for surveillance to be timed to periods of highest growth potential to yield highest probability of pest detection*
- *Establishment potential influences the level of survey effort which is negotiated as the requirement to demonstrate PFA status*
- *Establishment potential guides awareness raising campaigns in highest risk regions to improve chances of early detection via general surveillance*
- *Predicted pest generation time (see DARABUG2) informs appropriate revisit frequency of delimiting program*

L. huidobrensis spread potential

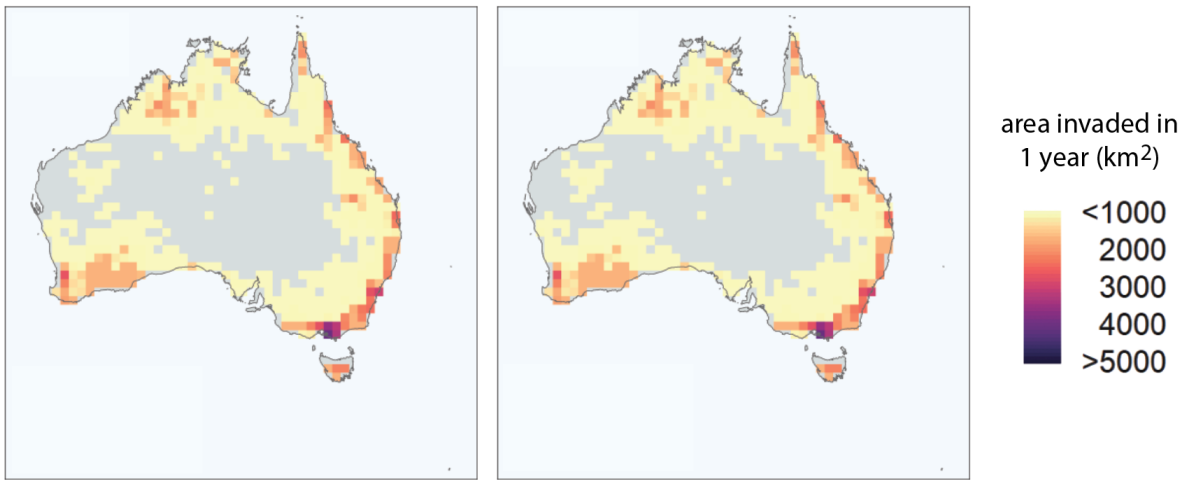


Figure 4. Rate of spread from locations across Australia. For each grid cell an incursion is simulation with the mean predicted area invaded by *L. huidobrensis* in one year shown by the colour gradient. The mean predicted area is estimated from 5 replicated simulations. Incursions were commenced in both July (winter incursion) and January (summer incursion), with minor differences detected. Predictions are made via the ESIM framework.

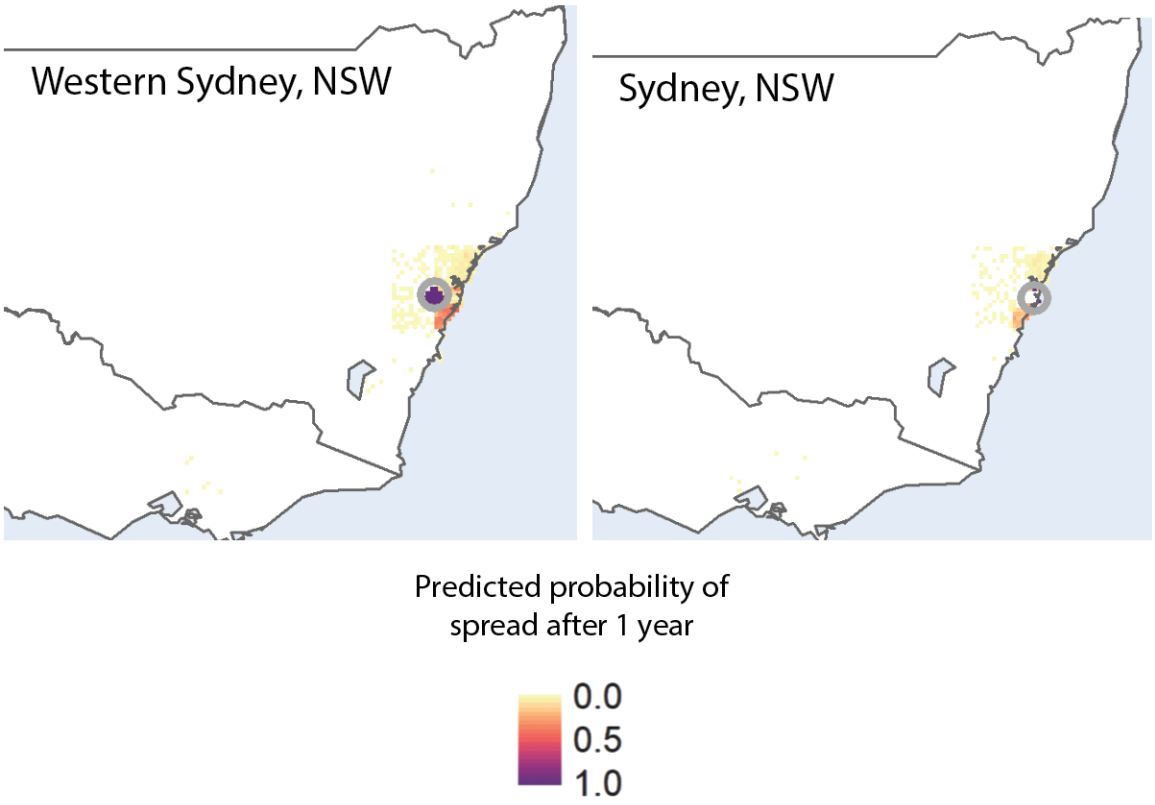


Figure 5. Probability of spread from Western Sydney and Sydney for *L. huidobrensis*, as a result of human movement and regional climate suitability. Probabilities denote the proportion of 100 replicate simulations in which a grid cell became established one year following the incursion scenario.



Figure 6. A dispersal prediction animation.

Below is a link to an example of *L. huidobrensis* population spread starting from Western Sydney across five years, as a result of local diserpal, human-assisted movement, and regional climate suitability using the ESIM spread module:
https://www.dropbox.com/s/hdrv62791za73tl/4_Liriomyza%20huidobrensis%20ESIM%20model%20outputs_dispersal%20from%20Western%20Sydney_051120.gif?dl=1

Applications of spread prediction outputs:

- *Predicted spread rates allow prioritization of highest risk entry point for early detection programs*
- *Predicted spread rates indicate level of effort that will be needed to maintain area freedom*
- *Predicted spread rates allow estimation of the preparedness timeframe before growing regions may be affected from various incursion locations in the absence of containment measures*

Appendix 3: Prospects for biological control of the vegetable leafminer, *Liriomyza sativae*, in Australia

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Contents

Executive summary **4**

1. Background **6**

1.1 Leafmining by insects 6
1.2 Polyphagous agromyzids 6
1.3 Host plant preferences of agromyzid parasitoids 7
1.4 Impact of insecticides on agromyzid parasitoids 8
1.5 Managing *Liriomyza* spp. in commercial crops 8
1.6 Augumentative biocontrol in protected cropping 9
1.6.1 Mass-rearing *9*
1.6.2 Use of thelytokous strains *9*
1.6.3 Use of banker plants *10*
1.6.4 Quality control of mass-reared natural enemies *10*
1.7 Augumentative and conservation biocontrol in open-field cropping 10
1.7.1 Re-distributing field-reared parasitoids *10*
1.7.2 Inter-cropping *11*
1.7.3 Parasitoid reservoirs and floral resources *11*
1.8 Other biological control agents 12

2. Hymenopteran parasitoids of *Liriomyza sativae* **13**

2.1 Parasitoids reared from *Liriomyza sativae* 13
2.2 Life history strategies of agromyzid parasitoids 14
2.3 Parasitoids of Agromyzidae in Australia 16
2.3.1 Nomenclature *16*
2.3.2 Australian Field Studies *16*
2.4 Details of Key Parasitoid Species 23
2.4.1 Eulophidae: Eulophinae *23*
2.4.2 Eulophidae: Entedoninae *25*
2.4.3 Braconidae: Opiinae *27*
2.4.4 Braconidae Alysiinae: Dacnusini *28*
2.4.5 Pteromalidae *28*
2.4.6 Figitidae: Eucoilinae *29*
2.5 Seasonality of Australian agromyzids and their parasitoids 29

3. Identifying potential reservoirs for parasitoids in key horticultural regions **30**

3.1 Australian Agromyzidae 30
3.2 Agromyzids common on weeds in southern Australia 31
3.3 *Tropicomyia polyphyta* 31
3.4 Agromyzids attacking lantana in Australia 32
3.5 Agromyzids attacking *Chenopodium* spp. 33
3.6 Agromyzids attacking grasses 33

4. Is there a need to import of exotic parasitoids into Australia? **33**

4.1 Classical Biological control programs overseas 33
4.1.1 Hawaii *33*
4.1.2 Guam *34*
4.1.3 Tonga *34*
4.1.4 Senegal *34*
4.1.5 Kenya *34*
4.1.6 New Zealand *35*
4.2 The Australian situation 35

5. Knowledge Gaps **35**

6. References **37**

7. Appendices **61**

Appendix 1. Parasitoid species reared from *Liriomyza sativae.* 61

Appendix 2. Parasitoid species reared from Agromyzidae in Queensland (Kleinschmidt 1970) 67
Appendix 3. Hymenopteran Parasitoids of Agromyzidae collected in Australia by State/Region 70
Appendix 4. Lower development threshold (°C) and thermal constant (day-degrees) for *Hemiptarsenus varicornis*, *Diglyphus isaea*, *Neochrysocharis formosus*, *N. okazakii, Opius cinerariae* and *O. caricovarae* on a range of agromyzid hosts 75
Appendix 5. Known species of Agromyzidae in Australia with host plant status (Spencer 1963, 1977; Elliott 2011; Lonsdale 2014) 79
Appendix 6. Queensland distribution records of Agromyzidae, extracted largely from Spencer (1963, 1977) and Kleinschmidt (1970) – updated 2017–12–01 82
Appendix 7. Host range for *Tropicomyia polyphyta* in Australia Kleinschmidt 1960, 1965, 1970; Spencer 1963, 1977a; Maunsell *et al.* 2017) 87

Executive summary

Relevant research from Australia and the rest of the world has been reviewed to identify the key parasitoid species involved in the biological control of vegetable leafminer, *Liriomyza sativae*, (Diptera: Agromyzidae) in open-air production and in glasshouses. Other biological control components currently being used or researched overseas (polyphagous predators, entomophagous nematodes, endophytic entomophagous fungi and the use of sterile insect release) are also considered to assess their potential role as components in Integrated Pest Management (IPM) programs for *L. sativae* in Australian horticultural crops.

Overseas, problems with the polyphagous *Liriomyza* pest species are universally associated with destruction of their parasitoids by excessive use of non-selective insecticides. It has been demonstrated repeatedly that to utilise parasitoids to control agromyzid leafminers, such as *L. sativae*, as part of an IPM program, it is necessary to avoid the use of broad-spectrum insecticides, to develop economic thresholds to reduce numbers of sprays applied, to understand the impact of host plant on parasitoid preferences, and to understand the role of non-crop hosts as reservoirs of parasitoids (ideally from non-pest agromyzid species).

With the ongoing dispersal of polyphagous *Liriomyza* species (*L. sativae*, *L. trifolii* and *L. huidobrensis*) through the world, studies in many countries show that these invading *Liriomyza* populations are rapidly exploited by endemic agromyzid parasitoids. The few published Australian field studies on the occurrence of agromyzid flies and their parasitoids show there is a substantial community of parasitoid wasps attacking agromyzids in Australia, with species mainly from Eulophidae, Pteromalidae and Braconidae. The most abundant species from these studies are two well-known cosmopolitan species (*Hemiptarsenus varicornis* and *Diglyphus isaea*) and four Australian species (*Zagrammosoma latilineatum, Trigonogastrella parasitica*, *Closterocerus mirabilis* and *Opius cinerariae*) for which little is known about their biology and ecology.

With increased surveying in Australia, it is highly likely that the numbers of species of both agromyzids and their parasitoids will increase substantially. Not all parasitoid species in these published studies have been formally identified, highlighting the importance of ongoing molecular studies to aid in identification. We believe strongly that the endemic parasitoid assemblage in Australia will be sufficient to control *L. sativae* and that there is no strong case to import additional exotic parasitoid species at this stage.

Since 70% of Australian species of Agromyzidae have no host plant records, there would be significant difficulties conducting host-specificity tests for any potential imports of exotic agromyzid parasitoids for a classical biological control program in Australia. In addition, agromyzid parasitoids tend to be not host-specific and indeed some species are known to attack lepidopteran leafminers as well as Agromyzidae. Again, this lack of host-specificity will make importation of potential agromyzid parasitoids very difficult because of potential impact on non-target species of agromyzids.

Fortuitously, *Diglyphus isaea,* the prime target parasitoid species for mass-rearing, is already present in Australia. It was first found in Sydney, New South Wales in 2001 and has since been found in Victoria, Tasmania and ACT, frequently on *Phytomyza syngenesiae* infesting sow thistle, *Sonchus oleraceus*. The species has also established in New Zealand.

In Europe and North America, only two species, *Diglyphus isaea* and *Dacnusa sibirica*, are currently reared commercially for use in augmentative biological control. The relatively high cost of production has restricted usage of these natural enemies to glasshouse producers. The more important mass-reared parasitoid, *Diglyphus isaea*, is an ectoparasitoid – the female wasp lays its egg in the mined leaf close to the agromyzid larvae. After hatching, the parasitoid larva then feeds on the host larva and finally pupates in the leaf. The female wasp also kills many host larvae through host-feeding (to obtain sufficient reserves to allow oviposition). Host feeding is an important component of mortality on hosts. This leads to a dilemma - host feeding is important for the development of parasitoid eggs but adversely impacts on the economics of mass-rearing as a large proportion of available hosts are killed outright rather than being a host for parasitoid larvae. Mass-production issues also include a tendency for a biased male sex ratio.

The results of this review, while comprehensive, highlight knowledge gaps as related to parasitoid control of agromyzid pests, particularly in Australia. These knowledge gaps are captured and discussed in detail within the final section of this review, which details further research and development that is required in order to more fully understand parasitoid control of *L. sativae* and other agromyzids.

In summary, key research needs identified in this review, and important from an Australian horticultural industry perspective, include:

- Delineating the current distribution of *Diglyphus isaea;*
- Establishing whether there are climatic biotypes for *Hemiptarsenus varicornis* and *Zagrammosoma latilineatum* – two eulophid species found from southern Australia to Cape York (and Indonesia);
- Determining what other agromyzid species in Australian horticultural regions will act as reservoirs of parasitoids for *L. sativae*;
- Studying the biology, ecology and distribution of endemic parasitoids in Australia (*Zagrammosoma latilineatum, Trigonogastrella parasitica*, *Closterocerus mirabilis* and *Opius cinerariae*) to assess their potential role in controlling *L. sativae* in open-air production as well as their potential suitability for use in augmentative biological control of *L. sativae*;
- Clarifying the taxonomy and biology of the complex of opiine braconid parasitoids already found attacking agromyzids in Australia; and
- Assessing the demographic performance and dispersal behavior of candidate parasitoid species following any mass-rearing initiative in Australia.

In light of increasing usage of IPM by horticulturalists, the significant investment Hort Innovation has made thus far to encourage uptake of IPM, and the recent detection of two more exotic *Liriomyza* species in Timor-Leste, information gaps captured in the final section of this review will be important in guiding further Hort Innovation investments into integrating biocontrol with parasitoid wasps into production systems.

This review also serves to progress development of *L. sativae* management and contingency planning within MT16004, which involves a significant extension component that seeks to educate growers about the pest and support adoption of effective management practices should *L. sativae* spread.

1. Background

The polyphagous agromyzid fly, *Liriomyza sativae* (Blanchard) (Diptera: Agromyzidae) was first detected in Australia at Seisia, on the tip of Cape York in 2015 (IPPC 2017), posing a threat to many horticultural crops grown in Australia. Based on the experiences in other parts of the world, it is clear that hymenopteran parasitoids will the most important group of beneficials controlling *L. sativae* in Australia. This review focuses on the current knowledge of the parasitoids of *L. sativae* in different areas of the world, and the parasitoids of agromyzid species already present in Australia. It identifies research questions to be answered to ensure that growers might best integrate these natural enemies into their control strategies for *L. sativae* as it invades horticultural production areas of Australia.

1.1 Leafmining by insects

The leafmining habit, where insect larvae feed and develop between the upper and lower epidermis of leaves (Liu *et al.* 2015a) has evolved many times in insects. Connor & Taverner (1997) estimated that worldwide, at least 10,000 species from 51 families of insects from 4 orders: Lepidoptera (34 families); Coleoptera (6 families); Diptera (9 families); Hymenoptera (3 families). The preponderance of lepidopteran leafminers has also been observed in Australia in the few detailed studies undertaken of leafmining to date. For example, Maunsell *et al.* (2017), in a study of leafminers in sub-tropical Australian rain forest, found 55 morphospecies of leafminers: Lepidoptera (41 species), Coleoptera (5 species), Diptera (4 species - 3 of which were Agromyzidae) and Hymenoptera (1 species). These findings do complicate field surveying for agromyzids because the majority of mines observed in non-agricultural settings will not be caused by agromyzids.

Benefits and disadvantages to the leafmining insects compared with external leaf feeders were reviewed by Connor & Taverner (1997). Benefits included (i) buffering of temperature within the leaf, (ii) protection from UV radiation, (iii) an increased feeding efficiency (either by avoiding plant defences or consuming more nutritious plant tissue) and (iv) reduced attack by predators. Disadvantages of leafmining were identified as (i) the loss of mobility of larvae, leading to higher levels of mortality from parasitism, (ii) mortality associated with premature abscission of leaves; and (iii) reduced fecundity due to smaller size compared with related external leaf-feeding insects.

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. The group is taxonomically difficult and species differentiation frequently relies on examination of the male genitalia. Molecular tests are proving very valuable for identification (Scheffer & Lewis 2005; Scheffer *et al.* 2006) since immature stages and adult females can be reliably identified (Blacket *et al.* 2015). Phylogenetic analysis has also been greatly assisted by new molecular tools (Scheffer 2005; Scheffer *et al.* 2007; Winkler *et al.* 2009).

1.2 Polyphagous agromyzids

Nearly all agromyzid species are very host-specific but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world (Spencer 1973, 1990). While only *Liriomyza sativae* has been recorded in Australia (IPPC 2017), the other key polyphagous species: *L. bryoniae*, *L. huidobrensis*, *L. trifolii* and *Phytomyza horticola*[1] all pose a significant quarantine threat to Australia as they are all now present in South East Asia.

[1] In an extensive molecular study of the *Phytomyza* and *Chromatomyia* genera using 3 genes (mitochondrial COI gene and the nuclear genes CAD (rudimentary) and PGD), Winkler *et al.* (2009) found that the *Chromatomyia* genus is (i) nested within the main lineage of *Phytomyza*, (ii) paraphyletic with respect to some *Phytomyza* species, and (iii) polyphyletic. Accordingly, *Chromatomyia* was synonymized with *Phytomyza* and 46 species described in *Chromatomyia* were transferred to *Phytomyza*. This nomenclatural change is followed in this review and so *P. horticola* and *P. syngenesiae* are now used in the text instead of *C. horticola* and *C. syngenesiae*.

Typically, these polyphagous leafminers are considered to have invaded countries via movement of infested plants (generally ornamentals such as chrysanthemum) (Spencer 1989). While fully-formed mines should be readily visible to quarantine officials, signs of early infestations are much less obvious and are easily overlooked (Spencer 1989).

Since the early 1990s, there has been a rapid movement of *L. sativae* and *L. huidobrensis* eastward through tropical and sub-tropical areas of Asia. *Liriomyza sativae* is the dominant agromyzid pest of vegetables in lowland areas (Spencer 1989; Andersen *et al.* 2002, 2008; Rauf *et al.* 2000) while *L. huidobrensis* is the dominant agromyzid pest of vegetables at higher elevations (>1000 m asl) in tropical Asia (Spencer 1989; Shepard *et al.* 1998; Sivapragasam & Syed 1999; Rauf *et al.* 2000; Tantowijoyo & Hoffmann 2010; Weintraub *et al.* 2017). Following the detection of *L. sativae* in Timor-Leste in 2003 (Walker 2005), it was subsequently detected in the Torres Strait in 2008 and Papua New Guinea in 2011 (Blacket *et al.* 2015) before being recorded at Seisia, Queensland in 2015 (IPPC 2017).

The biology and ecology of polyphagous *Liriomyza* spp. (including *L. sativae*) has been extensively reviewed by Spencer (1973, 1989), Parrella (1982, 1987), Parrella & Keil (1984), Minkenberg & van Lenteren (1986), Waterhouse & Norris (1987), Johnson (1993), Murphy & LaSalle (1999), Salvo & Valladares (2007), Kang *et al.* (2009), Liu *et al.* (2009, 2011), Musundire *et al.* (2011) and Reitz *et al.* (2013). Biological control of leafminers in glasshouse crops has been reviewed by Onillon (1999), Chow & Heinz (2004) and van der Linden (2004). In recent years, there has been much research conducted in Asian countries on aspects of the biology, ecology and control of *L. sativae* (*e.g.* Andersen *et al.* 2002, 2008; Johansen *et al.* 2003; Chen & Kang 2005 a, b; Hofsvang *et al.* 2005; Tran *et al.* 2005; Haghani *et al.* 2006, 2007a; Gao *et al.* 2012, 2014, 2017).

Worldwide, the polyphagous leafminers (including *L. sativae*) are secondary pests with outbreaks occurring when: (i) their suite of parasitoids become reduced in cropping systems by insecticides targeting other pests in the crop or the leafminers themselves, or (ii) naturally occurring parasitoids are not present in greenhouses. The value of parasitoids in regulating populations of *Liriomyza* spp. was recognized as early as 1884 (Webster & Parks 1913) who recorded 28 parasitoid species in their study.

> *"At times these minute enemies have become so numerous as to render even a careful study of the pest itself a matter of some difficulty. But for their presence these leaf-miners would beyond a doubt work much more serious ravages in the alfalfa fields of the West than they do at present. Indeed, one is inclined to wonder what the actual financial effects would be were some condition to arise suddenly whereby the numbers and efficiency of these natural checks were radically diminished."* Webster & Parks (1913)"

When polyphagous *Liriomyza* spp. invade another country, they will typically be attacked by a suite of generalist parasitoids already present in the country attacking a range of other non-pestiferous leafmining species (agromyzids or lepidopteran) (Murphy & LaSalle 1999; Reitz *et al.* 2013). Many of these agromyzid parasitoids have niche specialization (*i.e.* leafmines) rather than a taxonomic specialization (Ubaidillah *et al.* 2000; Salvo *et al.* 2011). Before importation of alien parasitoids into the country is undertaken, the local parasitoid fauna needs to be assessed and protected as a valuable biodiversity resource (Murphy & LaSalle 1999).

1.3 Host plant preferences of agromyzid parasitoids

Parasitoid species attacking the polyphagous *Liriomyza* spp. vary in their host plant preference (Musundire *et al.* 2012), so dominant parasitoid species vary between crops (Johnson & Hara 1987). Inevitably, with a polyphagous pest such as *L. sativae* which has a very broad host range, primarily but not exclusively, in the Fabaceae, Cucurbitaceae and Solanaceae, the composition of the parasitoid assemblage will vary with host. This can mean that a parasitoid species not common on a target horticultural crop may still play a very important role in regulating *L. sativae* populations on a regional scale. Similarly, Schuster *et al.* (1991) monitored parasitism in weeds adjacent to tomato crops in Florida and noted that weeds were a greater

source of parasitoids (77%) than the leafminers reared from the tomato crops (*L. sativae* and *L. trifolii*). The most common species found in the weeds was *Opius dissitus,* but the authors noted that the most common parasitoids found in the leafminer-infested tomato foliage were *Neochrysocharis diastatae* (reported as *N. punctiventris*) and *Diglyphus intermedius* (Schuster & Price 1985), which were less abundant in the weeds. Burgio *et al.* (2007) surveyed agromyzids and their parasitoids from weeds growing close to vegetable crops. Of the three most abundant species (*D. isaea*, *Neochrysocharis formosa* and *Pedobius metallicus*), only *P. metallicus* was considered unlikely to attack *L. huidobrensis* in the adjoining vegetable crops.

1.4 Impact of insecticides on agromyzid parasitoids

To control leafminer larvae developing inside the leaf, effective insectiides must be systemic or translaminar (*e.g.* abamectin) (Weintraub & Horowitz 1998), often aided by surfactants to increase movement of insecticide inside the leaf tissue (Babcock *et al.* 1999; Mujica *et al.* 2001; Weintraub & Mujica 2006; Bueno *et al.* 2007). Long residual insecticides which remain on the leaf surface will impact ovipositing adult leafminers and adult parasitoids searching for leafminer larvae but will not affect leafminer larvae (Kaspi & Parrella 2005). Laboratory bioassays conducted in Indonesia and Australia on *H. varicornis*, *Gronotoma micromorpha* (Indonesia only) and *D. isaea* (Australia only) showed that cyromazine, an insect growth regulator, was less disruptive than abamectin (Prijono *et al.* 2004; Bjorksten & Robinson 2005).

Secondary pest flare ups following killing of the parasitoid community by insecticides have been documented frequently with *Liriomyza* spp. (Hills & Taylor 1951; Oatman & Kennedy 1976; Johnson *et al.* 1980b, c; Hidrayani *et al.* 2005; Chirinos *et al.* 2017). Increased pest pressure was exacerbated by development of resistance in *Liriomyza* spp. to a range of insecticide classes (Parrella & Keil 1984; Leibee & Capinera 1995): organochlorines (Hills & Taylor 1951; Wene 1953; Genung 1957; Getzin 1960); organophosphates (Wolfenbarger 1958; Broadbent & Pree 1989); synthetic pyrethroids (Mason *et al.* 1987; Johansen *et al.* 2003); and spinosyns (Ferguson 2004). The impact of insecticides on parasitoids of *L. sativae* was demonstrated in Ecuador in a replicated trial (Chirinos *et al.* 2017) where weekly applications of lambda-cyhalothrin mixed with thiamethoxam resulted in a peak parasitism of 14% declining to 2% at the end of the trial (25 mines with unparasitized larvae per leaf at the end of the trial) whereas in the unsprayed plots peak parasitism was 85% (5 mines with unparasitized larvae per leaf at the end of the trial).

1.5 Managing *Liriomyza* spp. in commercial crops

Successful IPM programs to manage the spectrum of insect pests (including *L. sativae* and *L. trifolii*) have been implemented in field-grown horticultural crops such as tomato and celery in California (Johnson *et al.* 1980a, b, c, 1984, 1995; Trumble 1985; Reitz *et al.* 1999); watermelon in Hawaii (Johnson 1987; 2005; Johnson *et al.* 1989) and melon and lettuce crops in Arizona (Palumbo & Kerns 1998; Palumbo & Castle 2009). The foundations of these programs are to: (a) reduce initial leafminer pressure by (i) using uninfested transplants, (ii) destroying weeds and deep ploughing of senescent crops, and (iii) avoiding planting new crops adjacent to old crops (Capinera 2017); and (b) conserve parasitoids by (i) avoiding inappropriate chemical use (carbamates, organophosphates and synthetic pyrethroids) for control of *Liriomyza* spp. and the suite of other pests in the crop (Johnson *et al.* 1980b; Trumble & Toscano 1983), and (ii) using economic thresholds to delay and reduce sprays to allow the colonizing parasitoid populations to build up. For economic thresholds to be implemented successfully, effective sampling techniques are needed and, for *Liriomyza* spp., these include observing fresh stippling on leaves and counting of live larvae within leaves (since a mined leaf may no longer have a living leafminer larva present) (Heinz & Chaney 1995). Alternatively, assessments can be made by counting the number of *Liriomyza* puparia caught in sample trays on the ground surface (Johnson *et al.* 1980d), in situations where endoparasitoids emerging from the puparia are uncommon. Yellow sticky traps measure relative abundance of adult flies which is male-biased (Zehnder & Trumble 1984). While catches are very difficult to relate directly to crop damage, they can indicate times of migration into a crop (Palumbo & Kerns 1998).

1.6 Augumentative biocontrol in protected cropping

1.6.1 Mass-rearing

In protected cropping in Europe, Japan and North America, augmentative biological control is used extensively, but more widely in vegetable crops than ornamental crops (Chow & Heinz 2004), to ensure that parasitoids are present in the greenhouse as soon as leafminer damage is observed (Onillon 1999; Chow & Heinz 2004; Jacobson 2004; van der Linden 2004). The principal mass-reared parasitoid is the eulophid ectoparasitoid *Diglyphus isaea*, with some use also being made of *Dacnusa sibirica* (Hymenoptera: Braconidae: Alysiinae) early in the season (where *Liriomyza* spp. are overwintering in glasshouses) (van Lenteren & Tommasini 1999). Both these reared species initially invaded glasshouse crops in Europe naturally and were subsequently mass-reared – there was no systematic selection process to select the most appropriate species (or strains). Systems for mass-rearing of several eulophid species have been devised [*Diglyphus begini* (Parrella *et al.* 1989); *D. isaea* (Maharjan *et al.* 2017); *Hemiptarsenus varicornis* (Ozawa *et al.* 2004; Ho & Ueno 2008); *Neochrysocharis formosa* (Hondo *et al.* 2006a; Saleh *et al.* 2010); *N. okazakii* (Tran *et al.* 2007)], but as yet no other species apart from *D. isaea* and *D. sibirica* have been commercialized. To avoid superparasitism of 3rd instar *Liriomyza* hosts by *H. varicornis* when mass-rearing, Ho & Ueno (2008) recommended that 30–40 leafminers of the preferred 3rd larval instars should be offered daily per female parasitoid together with some 2nd instars for host feeding.

Unfortunately, *D. isaea* are very expensive to rear with retail unit cost ranging from USD 0.32 to USD 0.55 per wasp (assuming 50% males, the unit cost ranges from USD 0.64 to USD 1.09 per ♀wasp). Typically, 3–5 releases are made in glasshouses per crop. There can be foliage damage caused before the parasitoids control the leafminer population. In glasshouse chrysanthemum crops in the Netherlands, Zuijderwijk *et al.* (2008) observed that preventative introductions (five weekly releases) of *D. isaea* into the crop still required an initial treatment with cyromazine to reduce initial numbers of leafminer larvae. They also observed difficulties in establishing *D. isaea* in winter when growth lights were used because the adult parasitoids were being attracted into the growth lights and killed. Jacobson (2008) noted that costs could be reduced if parasitoids produced early in the season could be re-distributed to other parts of the glasshouse later in the season, effectively creating a within-crop rearing system. This approach should be explored as a way of reducing costs to growers, both in protected cropping and open production. With release rates of between 0.25–1 wasps per m^2 (USD 750 to USD 3,000 per hectare per release using USD 0.30 per wasp), it is not economic to use commercially-reared *D. isaea* augmentatively in the open field situation.

A further problem in mass-rearing *D. isaea* is the production of excess males. For example, Heimpel & Lundgren (2000) recorded 77% males on samples obtained from commercial insectaries. Assuming a unit cost of USD 0.32 per wasp, this equates to a cost per ♀of > USD 1.30. While this can be partially overcome with manipulation of host sizes offered to the wasps (Chow & Heinz 2005, 2006), production costs remain high since the shelf life for the adult wasps is very short. In addition, *D. isaea* kills many hosts for food so yield of parasitoids is reduced. Cold storage of mass-reared *D. isaea* & *H. varicornis* has been investigated in an attempt to increase storage life and so cater for unpredictable demand. Steinberg *et al.* (1991) and Burgio & Nicoli (1994) both found that honey fed *D. isaea* adults could be stored for up to a month at low temperatures without reducing survival, but fecundity was impacted. Chien *et al.* (2005a) stored pupae of *H. varicornis* at 10°C for several weeks and showed that while the survival was largely unaffected, the host killing capacity and fecundity of the wasps emerging from the cold-stored pupae were significantly reduced.

1.6.2 Use of thelytokous strains

One improvement to the rearing system would be using a female-only line. In Japan (Adachi-Hagimori & Miura 2008; Hagimori *et al.* 2006, 2008) and China (Yang *et al.* 2017), a widespread thelytokous line of *Neochrysocharis formosa* was shown to be caused by infection with a *Rickettsia* sp. Tagami *et al.* (2006) noted that the thelytokous strain of *N. formosa* was registered as a biopesticide in Japan in 2005. However, to date it appears not to have been commercialized; Japanese glasshouse producers still rely on importing *D. isaea* and *D. sibirica* from Europe.

Very recently, a new thelytokous species, *Diglyphus wani* Liu, Zhu & Yefremova, was described and found to be widespread in Qinghai, Hebei, Beijing and Shanxi Provinces, China (Ye *et al.* 2018). The mechanism for thelytoky has not yet been determined; the authors noted that a *Rickettsia* sp. was detected in relatively high frequency in the population, but that females in the laboratory continued to only produce female offspring for 5 generations after treatment with tetracycline (20 mg/ml). An initial study of the biological potential of *D. wani* suggests that it should be a highly valuable parasitoid for agromyzids, both in the field and in protected cropping (Ye *et al.* 2018), at least in areas where *D. isaea* is currently found. The discovery of *D. wani* is also a demonstration of the value of careful examination of the identity and biology of native parasitoids of agromyzids – this species has the potential to improve biological control in protected cropping worldwide.

1.6.3 Use of banker plants

Another approach to reduce costs of parasitoids would be to develop a "banker plant system", as commonly used with generalist aphid parasitoids reared on an alternative host (typically *Aphidius colemani* are reared on *Rhopalosiphum padi* growing on potted cereal plants which are placed in the glasshouse to allow *A. colemani* to target *Myzus persicae* or *Aphis gossypii* in the target crop) (Frank 2010; Huang *et al.* 2011; Jandricic *et al.* 2014). The requirements outlined by van der Linden (1992) for a successful banker plant system were: (i) both host plant and leafminer should be easy to breed in high numbers; (ii) the alternative leafminer should not be harmful to other crops; and (iii) the alternative leafminer should be a good host for the parasitoids. Additionally, the banker plant should not be a host for other pests, such as thrips, and thus act as a reservoir for plant pathogens. In the Netherlands, an open-rearing system using *Phytomyza caulinaris* reared on a buttercup species (*Ranunculus asiaticus*) to produce *Dacnusa sibirica* or *Diglyphus isaea* was tested successfully by van der Linden (1992). Since *D. isaea* acted as a facultative hyperparasitoid to *D. sibirica*, the system was not suited to rearing both species simultaneously. There has been no commercial uptake to date in Europe, but more recently Momoshita & Ohno (2016) studied *Pseudonapomyza spicata* as a candidate banker plant for parasitoids of *Liriomyza* spp.

1.6.4 Quality control of mass-reared natural enemies

It is difficult to maintain high quality mass-reared natural enemies. Selection within populations for traits favouring high levels of production in the mass-rearing facility, such as reduced ability to disperse and high parasitism rates at high host density, can be inherently opposed to those traits required for optimal effectiveness in the field such as high dispersal activity and high parasitism levels at low host density (van Lenteren & Tommassini 1999; van Lenteren 2003). The rapid rate of laboratory adaptation in laboratory cultures of insects was highlighted by Hoffmann & Ross (2018) who stressed the need to rear field-release populations under conditions that minimize the reduction of field fitness or ensure that colonies for field releases are renewed regularly from field material. If candidate species such as *H. varicornis* or *D. isaea* are mass-reared in Australia, it will be important to measure the variability in demographic performance and dispersal behavior between regional populations in Australia. There are IOBC quality control guidelines for the production of natural enemies, including *D. sibirica* and *D. isaea* (van Lenteren *et al.* 2003), which should be adapted for Australian requirements and species selected for mass-rearing.

1.7 Augumentative and conservation biocontrol in open-field cropping

1.7.1 Re-distributing field-reared parasitoids

In the former Soviet Union, inundative releases of *Phytomyza orobranchia* were used extensively for the biological control of the root parasitic weeds, *Orobranche cernua* and *O. cumana* (Klein & Kroschel 2002). This was accomplished by collecting *P. orobranchia* adults from overwintering puparia on *Orobranche* shoots held in a structure known as a "phytomyzarium'. The emerging flies were separated from parasitoids by an appropriate sized mesh screen when they emerged after winter. The flies were then released inundatively as part of the ongoing biocontrol strategy (in this case the parasitoids are killed) (Klein & Kroschel 2002). Kehrli *et al.* (2005) adapted this mass-emergence system to separate parasitoids from overwintering pupae of the horse chestnut leafminer *Cameraria ohridella* (Lepidoptera: Gracillariidae) for conservation or augmentation of parasitoids of the invasive leafminer. Kehrli & Phillips (2006) also used the

system to separate the Argentine stem weevil, *Listronotus bonariensis* from its parasitoid, *Microctonus hyperodae*.

A similar filtering system has been devised in Hawaii and Réunion for the separation of fruit fly parasitoids from fruit flies developing in fallen fruit. These structures, known as augmentoria, rely on mesh filters to collect the smaller parasitoids (Jang *et al.* 2007; Deguine *et al.* 2011a, b). These techniques could be adapted by Australian growers to move parasitoids from crop to crop relatively cheaply or to transport parasitoids from region to region (without the agromyzid flies) as a way of promoting biological control in newly-invaded areas.

1.7.2 Inter-cropping

In Peru, maize (*Zea mays*), a host of *L. graminovora*, and *Galinsoga parviflora* (Asteraceae), a common weed in potato crops and host of *L. sabaziae*, were planted within experimental potato crops to assess whether parasitism of *L. huidobrensis* in potato could be promoted (Mujica 2016). While parasitism levels were high in both conservation strips, parasitism in potatoes in the early stages of the potato crop were not enhanced. The recommendation was for conservation strips to be planted 50 days before the potato crop to enable parasitoids to provide more effective control earlier in life of the potato crop. Maize proved to be a superior parasitoid reservoir to *G. parviflora* (Mujica 2016). Intercropping peppers (*Capsicum annuum*) with sugar cane in China reduced the density of *L. huidobrensis* infesting the peppers and levels of parasitism were 70% in the intercropped (1 row pepper: 2 rows sugar cane) plots but only 6% on the pepper monoculture (Chen *et al.* 2011). Fenoglio *et al.* (2017) constructed spatially-explicit simulation models to examine the efficacy of *Vicia faba* as a trap crop for *L. huidobrensis* infesting potato. They found that 10% cover of the trap crop, together with a single insecticide spray in the middle of the season, was predicted to be the optimum solution. They did not look at the role of parasitoids in the trap crop as an alternative to the insecticide application.

In a very preliminary study, Herbert *et al.* (1984) showed that there was potential for reducing damage to glasshouse-grown chrysanthemums by using beans (*Phaseolus vulgaris* cv. Gold Rush) as a trap crop. There was no measurement of parasitism levels in either the chrysanthemums or the trap crop. Shahabuddin *et al.* (2015) observed that trap crops of cucumber, tomato and beans infested with *L. sativae* and *L. huidobrensis* grown beside crops of shallots in Central Sulawesi reduced crop damage and promoted parasitism of *L. chinensis* and *L. huidobrensis* in shallots by *H. varicornis* and *Chrysocharis pentheus*. However, it is necessary to assess the tri-trophic interactions on a case-by-case basis. In Hawaii, Johnson & Mau (1986) attempted to promote parasitism of *L. langei* (identified as *L. huidobrensis* in the paper)[2] in green onions by planting beans around the border. The beans were naturally infested with *L. sativae* and *L. trifolii*, but four of the parasitoid species found in the beans did not move onto *L. langei* in the onions.

1.7.3 Parasitoid reservoirs and floral resources

Conservation biological control seeks to contribute to the management of pests by supporting populations of natural enemies present in the agroecosystem and promoting their effectiveness (Begg *et al.* 2017). Two strategies, among many, at the local level (Landis 2017) are to enhance natural enemies and pest suppression by managing local habitats and to provide early-season resources for natural enemies. Several studies have looked at the agromyzid situation specifically. Massetti *et al.* (2010) found that flowering strips, with *Phacelia tanacetifolia* as the dominant component of the planting mixture, differentially promoted parasitism of ectoparasitoids over endoparasitoids of *L. huidobrensis* attacking lettuce crops in Italy, but there was no reduction in density of agromyzids in the trial. The authors considered that further research was needed to optimize plant mixtures and to understand how the dominant endoparasitoid species exploited flowering resources. Oida & Kawana (2017) tested the potential of *P. tanacetifolia* as a

[2] Scheffer (2000) demonstrated that the Californian and Hawaiian populations of *L. huidobrensis* formed a well-supported clade compared to *L. huidobrensis* populations found elsewhere. After additional testing with nuclear genes, Scheffer & Lewis (2001) resurrected *L. langei* for the California/Hawaii clade. In a recent detailed survey, only *L. langei* was found in Californian vegetable crops (Scheffer *et al.* 2014).

banker plant to improve parasitism of *L. chinensis* in field-grown onions (*Phytomyza horticola* colonized the banker plant naturally but did not attack the onions) and as an insectary plant (for feeding parasitoids emerging from *C. horticola*) to assist in the control of *L. chinensis* by parasitoids. In Argentina, Fenoglio & Salvo (2009) observed 19 parasitoid species attacking *L. commelinae* on *Commelina erecta* and that 13 of the species were shared with *L. huidobrensis* in the region. Since *C. erecta* is not considered to be a problem weed in horticultural crops, Fenoglio & Salvo (2009) suggested that the *C. erecta–L. commelinae* open rearing system had potential as a reservoir of generalist parasitoids and so assisting the biological control of *L. huidobrensis*. There is a need in Australian horticultural crops to understand the survival and spread of the generalist parasitoids already known to occur in the landscape, in order to enhance their effectiveness.

1.8 Other biological control agents

In high value protected cropping, other biological control options for *Liriomyza* spp. have been investigated but are not widely used at present. These include:

a. Predators: The most important predator of agromyzids are *Coenosia* spp., which are a widespread genus of muscid flies. Adults catch prey on wing (whiteflies, agromyzids, drosophilids etc. but also potentially parasitoids) and the soil-dwelling larvae are also predatory, feeding on other fly larvae in the soil such as fungus gnats (sciarids) (Eisenbach *et al.* 2005; Ugine *et al.* 2010; Mateus 2012). Mass-rearing procedures for *Coenosia* spp. using larval fungus gnats have been developed in Thailand (Kühne 1998), Portugal (Martins *et al.* 2015) and China (Zou *et al.* 2017). With the advent of effective IPM programs for *Liriomyza* spp. around the world, *Coenosia* spp. have been much more widely recognized in protected cropping systems (Couri & Salas 2010; Pohl *et al.* 2011; Bautista-Martínez *et al.* 2017) and their presence is a useful indicator of the health of the *Liriomyza* management system. In Australia, there are six recorded species of *Coenosia*. Seabra *et al.* (2015) has shown that two species described in Sydney by Malloch (1925a) (*C. imitatrix* and *C. subvittata)* were in fact males and females of the globally distributed *C. attenuata*. Spencer (1977a) noted that a specimen of *C. acuticornis* was pinned with a specimen of *Liriomyza brassicae* collected by Malloch (1925b) who had wrongly identified the agromyzid to be *Agromyza pusilla*. Malloch (1925b) noted that *C. acuticornis* was a common species in Australia and had been recorded from New South Wales, Queensland, Tasmania and Victoria (from where it was originally described). There is also a report of a *Coenosia* sp. predating on adults of the leafhopper, *Balclutha incisa*, in the field in Western Australia (Narhardiyati & Bailey 2005). Unfortunately, there were no voucher specimens kept and the fly was not identified to species.

b. Sterile insect technique (SIT): Gamma irradiation is used to sterilise adult flies released into the glasshouse to disrupt mating of the wild-type flies (Kaspi & Parrella 2003; Walker 2012; Sultan 2015; Sultan *et al.* 2017). SIT has been aimed at greenhouse production where *D. isaea* is unable to control *L. trifolii* populations without unacceptable levels of damage being caused to crop plants. Kaspi & Parrella (2006) concluded that a combination of parasitoid release and SIT was better than either method used alone. Tagami *et al.* (2006) *s*uggested *Wolbachia*-induced cytoplasmic incompatibility in *L. trifolii* could be used as an alternative to gamma irradiation for SIT. In Brazil, Parish *et al.* (2017) found no evidence of *Wolbachia* populations of *L. sativae*, *L. huidobrensis*, *L. brassicae* and *Calycomyza malvae* using 3 genes (16S, wsp and COI).

c. Entomopathogenic nematodes (Harris *et al.* 1990; Williams & MacDonald 1995; Head *et al.* 2000): Nematodes are applied to leaves as a spray application and gain entry into the leaves via the feeding punctures caused by the female *Liriomyza* spp. Very high humidity is required for survival and efficacy of the nematodes which restricts their usefulness.

d. Entomophagous fungi: Fungi is applied as a foliar spray (Bordat *et al.* 1988; Devkota *et al.* 2016), in an autodissemination device (Migiro *et al.* 2010, 2011; Gathage *et al.* 2016) or as an endophytic treatment where germinating seeds are colonised by fungal endophytes (including the entomopathogenic fungi, *Beauveria bassiana*) before planting (Akutse *et al.* 2013; Gathage *et al.*

2016). Preliminary field trials with strains of two endophytic fungi, *B. bassiana* and *Hypocrea lixii,* together with *Metarhizium anisopliae* applied in an autodissemination treatment gave promising results in terms of increased yields of common bean, *Phaseolus vulgaris* (Gathage *et al.* 2016). These methods require further development before being viable for commercial use.

2. Hymenopteran parasitoids of *Liriomyza sativae*

2.1 Parasitoids reared from *Liriomyza sativae*

Seventy one parasitoid species have been recorded from *L. sativae* worldwide (Table 1; Appendix 1). The family Eulophidae is the dominant parasitoid group (73% of species and equally spread between the subfamilies Entendoninae and Eulophidae), with the Braconidae, Pteromalidae and Figitidae families also being important. Important genera include *Diglyphus*, *Neochrysocharis*, *Hemiptarsenus*, *Opius*, *Phaedrotoma*, *Halticoptera*, *Banacuniculus* and *Gronotoma*. Specimens only identified to genus or reared from mixed collections of *Liriomyza* spp. (which included *L. sativae*) are not included in this list. As with any compilation of species names from the literature, there will be uncertainty about the validity of some species and sampling intensity varies dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem. Shaw (1994) addressed these problems when considering the meaning of "host range". His useful definition was *"the host range of a particular parasitoid species includes only the species of potential hosts that the parasitoid is usually able to attack successfully, following a pattern of search behaviour enabling it to encounter them regularly".* As a consequence, Shaw (1994) suggested that some genuine rearing records should be discounted if they represent freak events and which are no importance to the ecology of the parasitoid or host. His definition also implies that some hosts will be more important than others, both in terms of host suitability and also phenology (host availability). He also emphasized the need for quantitative expression of rearing data and introduced the concept that the "realised host range" was dynamic in space and time.

Table 1. Hymenopteran parasitoids reared from *Liriomyza sativae* worldwide summarized by Family

Family	Sub-family	Genera	Species	% Composition
Eulophidae		15	55	72%
	Entedoninae	8	28	
	Eulophinae	7	27	
Braconidae	Opiinae	4	12	16%
Pteromalidae		2	5	7%
	Miscogasterinae	1	4	
	Pteromalinae	1	1	
Figitidae	Eucoilinae	4	4	5%
	Total	23	76	100%

Noyes (1994) noted that published host-parasitoid lists of well-known species can be extremely inaccurate for a range of reasons including:

- Mixed series of hosts

- Mixed series of parasitoids and hyperparasitoids
- Misidentification of host and/or parasitoid due to inadequate taxonomy
- Misidentification of host/parasitoid due to poor taxonomy
- Author disregarding opinion of a taxonomist
- Misspelt parasitoid or host names
- Misidentification resulting from inadequate material
- Misidentification resulting from inadequate information

He cited the example of the list of parasitoids of the diamondback moth, *Plutella xylostella*, where 33 species were reliably recorded; 59 species required confirmation and 16 species were erroneous.

He suggested a number of ways to improve the accuracy of published host-parasitoid lists as follows:

- Do not repeat published host-parasitoid lists in subsequent publications unless they have been checked by specialists in both parasitoid and host taxonomy.
- Consult appropriate taxonomists before starting a research project, or even before starting to prepare a grant application.
- Isolate hosts and preserve their remains with the parasitoids that emerge.
- Include at least locality and host information with any material to be identified.
- Deposit voucher material of parasitoids and their host remains in a well-maintained, major collection.
- State in any publication who identified the host and/or parasitoids and state where the material has been deposited.

These suggestions should be followed closely for future Australian studies of agromyzid parasitoids, with the additional provision that molecular barcoding should be undertaken for parasitoids reared from agromyzids.

2.2 Life history strategies of agromyzid parasitoids

Agromyzid parasitoids exhibit a range of life history traits, which enables the parasitoid assemblage to persist (Pennachio & Strand 2006). The idiobionts paralyze their hosts immediately while the koinobionts allow their parasitized host to continue to develop until pupation, when the parasitoid larva starts to develop. The idiobionts are synovigenic (none or very few mature eggs present at eclosion) and the females need to feed on host nutrients for development of the eggs (Zhang *et al.* 2011). The host-feeding parasitoid encountering a host has to decide whether to oviposit immediately (investing in current reproduction) or to host feed and oviposit later (investing in future reproduction) (Zhang *et al.* 2011; Liu *et al.* 2014, 2015b). The koinobionts are pro-ovigenic (full complement of eggs at eclosion) and do not require host-feeding to maximise their fecundity. Table 2 lists some of the life history traits associated with idiobiont and koinobiont parasitoids (Mayhew & Blackburn 1999; Traynor & Mayhew 2005) but note that traits are trends and will vary. For example, the eulophid *N. formosa* is an idiobiont endoparasitoid rather than the more characteristic ectoparasitoid. Traynor & Mayhew (2005) suggests that koinobionts can be considered as having "slow" larvae but "faster" adults, while idiobionts have "fast" larvae but "slow" adults. Similarly, koinobiont parasitoids have been considered as "capital breeders (CB)" (where energy is stored as a larva and used during adult for reproduction) while idiobiont parasitoids have been considered as "income breeders (IB)" (where energy for reproduction is used from current energy harvest). However, Jervis *et al.* (2008) pointed out that the dichotomy of capital/income breeder was too simplistic and noted that it was necessary to look at reserves of individual nutrients (see Casas *et al.* 2005) – a parasitoid wasp can be a capital-breeder in terms of lipids but along the CB-IB spectrum for other nutrients such as protein and glycogen.

Table 2. The suite of life history traits typically associated with idiobiont and koinobiont parasitoids (adapted from Traynor & Mayhew 2005; Quicke 2015)

Idiobiont	Koinobiont
Permanent host paralysis	Temporary / no host paralysis
Generalist	Specialist
Ectoparasitoid	Endoparasitoid
Rapid larval development	Slow / delayed larval development
Long adult lifespan	Short adult lifespan
Lower lifetime fecundity	Higher lifetime fecundity
Host concealed	Host exposed
Host stage attacked larger than wasp	Host stage attacked often smaller than wasp
Host-feeding common	Host-feeding uncommon
Synovigeny	Pro-ovigeny
Large eggs with much yolk	Small eggs with little yolk
Oosorption	No oosorption
Male eggs preferentially laid in small hosts	No such relationship
Sexual dimorphism often pronounced	Sexual dimorphism absent / less pronounced
Mostly diurnal	Diurnal/nocturnal

The eulophid parasitoids are typical idiobiont parasitoids and cause host mortality in three ways: host parasitism, host feeding and host stinging (without oviposition or feeding) (Liu *et al.* 2015b; Cheng *et al.* 2017). Because idiobionts paralyze their hosts before they oviposit, the food resource of the agromyzid larva is fixed at time of oviposition, so eggs are generally laid in or beside the 3rd instar larva (the oldest stage). Ectoparasitoids such as *H. varicornis* or *D. isaea* feed on the paralyzed larva and then pupate beside the dead host larva before emerging from the mine. Idiobiont endoparasitoids, such as *N. formosa* or *N. okazakii*, also paralyze the host larva but the egg is laid inside the larva and the parasitoid larva feeds and pupates inside the host larva and also emerge from the mine. The idiobiont parasitoid females emerge with very few eggs so they need to feed on fluids of hosts to develop their eggs before oviposition. Host feeding is strongly related to wasp fecundity and longevity (Chien *et al.* 2004; Zhang *et al.* 2014; Cheng *et al.* 2017). Female wasps locate and puncture smaller larvae in the leaf then feed on the punctured larva which then dies.

The larval-pupal endoparasitoids in the Opiinae, Eucoilinae and some Pteromalidae are koinobionts. Since these wasps do not paralyze the host larva at oviposition, the agromyzid larva continues to mine the leaf until pupation. The parasitoid does not begin to develop until the host larva pupates. Any host feeding these wasps do, also tends to be non-fatal to the agromyzid larva. The wasp will oviposit into any of the instars of host larva since its larva will not begin development until its host begins to pupate. In most cases, the parasitoid is able to immunosuppress the host and so prevent encapsulation of its egg by the host haemocytes (Strand & Pech 1995). However there have been a number of reports where indigenous parasitoids are not able to successfully attack a newly-arrived agromyzid. In Europe, *Opius pallipes* was an important parasitoid of *L. bryoniae* in tomato glasshouses, but the eggs of this parasitoid were encapsulated in the larvae of *L. trifolii* and so was no longer viable for mass-release in glasshouses once *L. trifolii* became important (Sunderland *et al.* 1992) and *Dacnusa sibirica* was released in its place. In Japan, the dominance of *L. sativae* over *L. trifolii* has been attributed, at least partially, to the ability of *D. sibirica* to develop in *L. trifolii* but not *L. sativae* (mechanism unknown) (Abe *et al.* 2005; Abe 2017). In contrast, in mainland China (where *L. trifolii* was first recorded 11 years after *L. sativae*), Xing *et al.* (2017) observed the interaction between *L. sativae*, *L. trifolii* and the pupal parasitoid *Phaedrotoma* (= *Opius*) *biroi*. They observed that the proportional niche similarity and niche overlap between *L. sativae* and *P. biroi* was significantly greater than between *L. trifolii* and *P. biroi*. This meant that *L. trifolii* outperformed *L. sativae* since it avoided parasitization by *P. biroi* to a much greater degree than *L. sativae*. It is not known whether endemic *Opius* and *Phaedrotoma* spp. in Australia will be encapsulated by *L. sativae*. In Japan, Kemmochi *et*

al. (2016) observed that the koinobiont pteromalid, *Halticoptera circulus* was encapsulated by *L. trifolii* but not by *L. chinensis* or by *C. horticola*. It was uncommon for *L. sativae* to be parasitized by *H. circulus* in Japan, so they suspected that encapsulation may also be occurring with this species. They also noted that *H. circulus* was a common parasitoid of *L. trifolii* in California, and of *L. sativae* and *L. trifolii* in Hawaii (Johnson 1987), suggesting that encapsulation was not an issue in those areas.

Salvo & Valladares (2004) concluded that mine appearance significantly affected parasitic assemblage structure, with shape driving generalist species richness and contrast influencing that of specialists. Mine shape also affected parasitism rates, which were highest for generalists in linear mines, and for specialists in blotch mines. *Liriomyza sativae* was classed as a linear-blotch mine with high contrast so generalist parasitoids would be expected to be dominant. But note that Shaw (1994) questioned the dichotomy of generalist parasitoids (hosts with shared ecological characteristics) versus specialist parasitoids (hosts phylogenetically related) and considered that the host range of a particular parasitoid was a balance between both factors.

Typically, in any field study, there will be 2–4 parasitoid species which are dominant, with a number of other species in smaller numbers. For example, in a detailed field study in Java, Tantowijoyo & Hoffmann (2010) found 12 species of hymenopteran parasitoids attacking *Liriomyza* spp. but the dominant parasitoids of *L. sativae* at low altitudes were *Neochrysocharis beasleyi*, *N. formosa*, *Asecodes delucchii* and *H. varicornis*, whereas at high altitudes only two parasitoid species (*H. varicornis* and *Opius chromatomyiae*) were found attacking *L. huidobrensis* with *O. chromatomyiae* being the most common. The parasitoid assemblage can change with time. Wahyuni *et al.* (2017) found that in the Lesser Sunda Islands of Indonesia, *N. formosa* has become more abundant than *H. varicornis* in many vegetable crops infested with *Liriomyza* spp. They also noted that *O. chromatomyiae* is now also found in lowland areas, when previously it had been restricted to highland areas.

2.3 Parasitoids of Agromyzidae in Australia

2.3.1 Nomenclature

For the purpose of this review, the nomenclature of the Chalcidoidea follows the Natural History Museum Universal Chalcidoidea Database http://www.nhm.ac.uk/our-science/data/chalcidoids/ (Noyes 2018). The only exception is for *Neochrysocharis formosa* where we accept the gender of *Neochrysocharis*, based on the Greek ending *-charis*, is feminine. The nomenclature of the Opiinae follows Taxapad Ichneumonoidea http://www.ichneumonoidea.name/local.php?taxonidLC=90216107 (Yu *et al.* 2016). The systematics of the Opiinae remains controversial, with van Achterberg & Salvo (1997) restricting the definition of *Opius* to species with a basal lobe on the mandible while Wharton *et al.* (2013) retains a more conservative approach to the definition of the genus. A recent molecular study (Li *et al.* 2013) using n28S rDNA, D2 region, mt16S rDNA and COI supports the van Achterberg & Salvo (1997) classification.

2.3.2 Australian Field Studies

In Australia, there has been very little systematic sampling of agromyzids and their parasitoids. Without rearing records of agromyzids (to confirm host plants) and parasitoids (to confirm host insects), the current knowledge of the endemic parasitoid fauna in Australia remains sparse, both in terms of which species are present and how these species are distributed, temporally and spatially. To date, the handful of published and unpublished Australian field studies are from very restricted areas.

Parasitoid records from all the below Australian studies have been collated (Appendix 3) and Table 3 gives the Superfamily, Family and Sub-family of the genera of the parasitoids. Those genera most commonly recorded in Australia are in bold and listed in priority order for potential importance for impact on *L. sativae*. These species are *Hemiptarsenus varicornis*, *Diglyphus isaea*, *Zagrammosoma latilineatum*, *Neochrysocharis* spp., *Closterocerus mirabilis*, *Chrysocharis pubicornis*, *Trigonogastrella parasitica*, *Opius atricornis*, *O. cinerariae* and *Phaedrotoma oleraci.* Two genera, *Halticoptera* (Pteromalidae) and *Gronotoma* (Figitidae; Eucoilinae), are also highlighted because several records exist of these genera in Australia and

parasitoids from these genera are important in some countries for control of *Liriomyza* spp. Specific comments about these parasitoids are detailed in Table 4.

2.3.2.1 South Australia

1. Lardner (1991) conducted a 2-year (15 sampling dates) survey of *Liriomyza brassicae* on 4 host plant species in Adelaide.

Key Findings:

(a) *Hemiptarsenus varicornis* and *Zagrammosoma* sp. (but likely to be *Z. latilineatum* which was described from Indonesian and Australian material by Ubaidillah *et al.* 2000) were the two most abundant parasitoids (numerically and temporally) reared in the field from *Liriomyza brassicae*.

(b) Laboratory cultures of *H. varicornis*, *Opius cinerariae* and *Chrysocharis pubicornis* were established relatively easily but culturing of the other 4 parasitoid species was unsuccessful because of (i) a lack of numbers, (ii) difficulties in mating, or (iii) lack of interest by the parasitoids in attacking *L. brassicae* larvae.

(c) *Hemiptarsenus varicornis* was also reared from *P. syngenesiae* on sowthistle, *Calycomyza humeralis* on aster and *L. chenopodii* on *Chenopodium album* (white goosefoot). *Closterocerus* sp. (but likely to be *C. mirabilis* which was described from Victorian material in 2004 (Edwards & La Salle 2004)) was also reared from *P. syngenesiae*.

(d) Despite having access to world leading authorities in Eulophidae and Opiinae, 25% of parasitoids reared in the main survey (134 of 533 specimens) were unable to be identified. This highlights the urgent need for molecular studies to supplement the morphological studies already in place.

2. Wood *et al.* (2010) undertook a 9-month (12 sampling dates) survey of native saltbush (*Chenopodium = [Rhagodia] candolleana* and *C. parabolica*) plantings at two vegetable growing properties in the Virginia horticultural area in the Northern Adelaide plains. Both hosts were infested with a blotch leafminer putatively identified as *Phytoliriomyza praecellens*. This species was described by Spencer (1977a) from a female specimen but with no host plant records. Given that 70% of known Australian agromyzids do not yet have host plant records (Spencer 1977a), it is a salient example of the value in examining the parasitoids of agromyzids from as many host plants as possible.

Key Findings:

(a) Leafminers and parasitoids were 8 times more abundant on *C. candolleana* than *C. parabolica*.

(b) Ten parasitoid species were associated with *C. candolleana*; 7 of these species were reared from *C. parabolica*

(c) The most abundant species on *C. candolleana* in spring were the pteromalid, *Trigonogastrella* sp., and the two eulophids, *Z. latilineatum* and *H. varicornis*. However, in autumn, *Z. latilineatum* was the dominant species. The authors implied that the pteromalid was not *T. parasitica,* so it was likely to be *T. rabiosa*, which had previously been recorded from South Australia (Bouček 1988).

(d) High levels of larval mortality were observed on *C. candolleana*, reflecting the dominance of the idiobiont parasitoids on this host.

(e) Two *Opius* spp. were the most abundant taxa on *C. parabolica* but very few were found on *C. candolleana*. They were not identified to species. Was this a reflection of the reduced numbers of idiobiont parasitoids on *C. parabolica* which reduced hyper-parasitism and host-feeding losses of the koinobiont *Opius* spp.?

(f) There is potential for use of banker plants infested with non-crop damaging agromyzids in the field and in glasshouses to act as source of parasitoids for *Liriomyza* spp.

2.3.2.2 Victoria

3. Osmelak (1983) made observations for 3 months (August-October) in 1980 of leafmining in weeds and some vegetable crops (lettuce, artichoke) at Werribee South. Three leafmining species were identified: two drosophild species (*Drosophila busckii* and *Scaptomyza australis*) and one agromyzid species, *Phytomyza syngenesiae.*

Key Finding:
(a) Two parasitoid species recorded from leaves mined by *P. syngenesiae* were *H. varicornis* and *Trigonogastrella* sp. (likely to be *T. parasitica*).

4. Bjorksten *et al.* (2005) conducted a 4-month survey of weeds and some vegetable crops in southern Victoria, together with weekly monitoring 2 crops of Chinese cabbage (2 x 6 weeks) and 2 crops of beetroot (3 weeks in spring and 6 weeks in autumn). The agromyzid species examined were *Liriomyza brassicae*; *L. chenopodii* and *P. syngenesiae*.

Key Findings:
(a) *Liriomyza brassicae* (39 parasitoid specimens): the most abundant parasitoids were *H. varicornis, Diglyphus isaea* and *Closterocerus mirabilis*.
(b) *Liriomyza chenopodii* (338 parasitoid specimens): the most abundant parasitoids were *H. varicornis* and *D. isaea,* with *Opius cinerariae* and *Z. latilineatum* the next most abundant.
(c) *Phytomyza syngenesiae* (146 parasitoid specimens): the most abundant parasitoids were the native eulophid, *Closterocerus mirabilis*, the native pteromalid, *Trigonogastrella parasitica,* the adventive alysiine, *Dacnusa aerolaris,* and an unidentified entedoninae genus.
(d) Use of sentinel plants for sampling *Liriomyza* spp. yielded many more *Opius* spp. than sampling mined leaves, since *Opius* spp. develop off the plant in the puparia of *Liriomyza* spp. This needs to be considered in future sampling studies. Other examples of the use of sentinel plants to assess parasitism of *Liriomyza* spp. were by McClanahan (1977) and Amano *et al.* (2008).
(e) Very high levels of parasitism and larval mortality, presumably due to host feeding by idiobiont parasitoids, of *L. chenopodii* were found in test crops grown in autumn and spring.
(f) One specimen of *Neochrysocharis okazakii* was recorded. This species is of some importance for control of *Liriomyza* spp. in Indonesia, Japan and Vietnam. It has potential as a parasitoid of *L. sativae*.

2.3.2.3 Australian Capital Territory and New South Wales

5. Lambkin *et al.* (2008) undertook a 2-month survey of mined weeds in December and January in Canberra, ACT and at Forbes Creek, NSW with parasitoids reared from *P. syngenesiae* and *P. plantaginis*.

Key Findings:
(a) Six *Opius* morphospecies were reared on *P. syngenesiae*. This result highlights the need for improved diagnostics for this taxonomically difficult group. Since pupae of *P. syngenesiae* remain in the leaf, the likelihood of rearing opiines from leaf material is increased greatly compared with those *Liriomyza* spp. which pupate away from the leaf.
(b) *Hemiptarsenus varicornis* and *T. parasitica* were the most abundant parasitoids reared in their survey.
(c) *Diglyphus isaea* was reared from *P. plantaginis* but not from *P. syngenesiae*.
(d) Improved rearing techniques for parasitoids involved removing pupa from leaves into vials (both *P. syngenesiae* and *P. plantaginis* pupate in leaves).
(e) An unidentified *Neochrysocharis* sp. was recovered from *P. plantaginis*. Improved diagnostics are needed as *N. formosa* and *N. okazakii* are very important parasitoids of *Liriomyza* spp. overseas (Appendix 1).
(f) Malaise and emergence trapping showed that the grass-mining agromyzid, *Cerodontha milleri* was by far the most abundant of the agromyzids trapped. *Cerodontha* spp. are widespread in Australia (11 species described) and may prove to be very valuable reservoirs of parasitoids for *L. sativae*.
(g) Only female flies of *Phytomyza plantaginis* were reared. Parthenogenetic strains of this species are recorded in USA, Canada and New Zealand (Spencer 1990). The mechanism for this strain should be investigated and parasitoids reared from *P. plantaginis* should also be tested for sex ratio abnormalities and presence of endosymbionts.

2.3.3.4 South East Queensland and Northern NSW

6. A detailed survey was made of leafminers (primarily lepidopteran) in subtropical rain forest in the Lamington and Border Ranges National Parks (Maunsell 2014; Maunsell *et al.* 2015, 2017). Only three agromyzid species and some associated parasitoids were collected.

Key Findings:

(a) The highly polyphagous *Tropicomyia polyphyta* was the most common agromyzid species collected and was recorded from 17 species in 11 orders. Two parasitoid species were recorded – an opiine, Braconidae species B, and a pteromalid, *Herbertia* sp.
(b) *Phytoliriomyza queenslandica* was recorded from *Pittosporum multiflorum* and *P. oreillyanum* for the first time. One unidentified braconid parasitoid species was recorded.
(c) *Phytomyza vitalbae*, which is an adventive European species attacking *Clematis* spp., is recorded from *Clematis pickeringii* for the first time. No parasitoids were recorded.

2.3.3.5 Northern Territory

7. Parasitoids of the pigeonpea pod fly, *Melanagromyza obtusa* (Malloch) were collected from pods of *Cajanus latisepalus* in March and May 2004 (Makinson *et al.* 2005).

Key Findings:

(a) A *Callitula* sp. is the first record of a pteromalid from *M. obtusa*. Bouček (1988) noted that there were at least 8 species recorded in Australia and that in tropical areas, the main hosts were leafmining or stem-mining species on herbaceous plants.
(b) *Ormyrus* sp. (Hymenoptera: Chalcidoidea: Ormyridae). Species within this genus are primarily parasitoids of gall-forming insects. *Ormyrus orientalis* has been recorded from *M. obtusa* in India but the Australian species did not key out to this species.

2.3.2.6 Torres Strait

8. Tentative identification by John La Salle of 8 species of parasitoids of *L. sativae* collected by Luke Halling (NAQS) in 2008 from several islands in the Torres Strait.
Eulophidae: *Hemiptarsenus varicornis*, *Zagrammosoma latilineatum*, *Closterocerus* sp., *Quadrastichus sp.* (likely to be *Q. liriomyzae*), *Neochrysocharis* sp. – [possibly *N. okazakii*], *Cirrospilus brevicorpus* [=*ambiguus*] – originally described from *Liriomyza trifolii* and *Calycomyza lantanae* (Hansson & LaSalle 1996).
Pteromalidae: *Callitula* sp.; *Herbertia* sp.
Braconidae*: Opius* sp.

Key Findings:

(a) Even from these limited collections, it is clear that there is a suite of parasitoids already present in the Torres Strait that are moving onto the invading *L. sativae* and which may be important in regulating the pest populations. Further collections from *L. sativae* are needed in the Torres Strait and Far North Queensland to consolidate these initial findings.

2.3.2.7 Queensland

9. Queensland records from 1915 to 1958 catalogued by Kleinschmidt (1970) were largely from the Brisbane and Cairns area. Details of parasitoid species reared from each agromyzid species are given in Appendix 2.

Key Finding:

(a) Most species of parasitoids were reared from *Ophiomyia phaseoli*, *P. syngenesiae*, *L. brassicae*, *L. chenopodii* and *Calcomyza humeralis*.
(b) The most commonly reared parasitoids were *H. varicornis*, *O. atricornis* and *O. cinerariae.*

2.3.2.8 Other Australian data

10. The other major source of Australian information comes from the major monograph on Australasian Chalcidoidea by Bouček (1988) where records of a number of chalcidoid parasitoid species (reared and putative) of agromyzids are catalogued from Australian and international collections, together with the nomenclatural details for Australian species.

Key Findings:
Eulophidae:
(a) Australian distribution data and nomenclatural clarity for *Hemiptarsenus varicornis*
(b) Australian distribution data for the adventive *Chrysocharis pubicornisi*
(c) *Meruacesa* (=*Meruana*) *liriomyzae* recorded in Brisbane, without agromyzid host data, as well as a record from an unidentified cecid in New South Wales. It has been recorded attacking *L. sativae* and *L. brassicae* in Mauritius, *L. sativae* in Reunion and from a range of agromyzids in Africa.
Pteromalidae
(a) Australian distribution data and nomenclatural changes for *Trigonogastrella parasitica* and *T. rabiosa*
(b) Australian distribution data for pteromalid genera likely to include agromyzid parasitoids: *Ammeia*, *Callitula* spp., *Herbertia* spp., *Merismomorpha* spp., *Oxyharma* spp., *Sphegigaster* spp. and *Syntomopus* spp.,
(c) Noted presence of an as yet unnamed *Halticoptera* sp. in Queensland, which could be significant given the *importance* of *Halticoptera* spp. as parasitoids of *Liriomyza* spp. in North and South America.

Table 3. Superfamily, Family, Sub-family and Genus of the parasitoids reared from Agromyzidae in Australia

The twelve parasitoid genera currently considered to be most important in Australia for biological control of *L. sativae* are in bold and at the top of the list of each Sub-family (in priority order). The number of genera in each Sub-family is in parentheses.

		Chalcidoidea				Ichneumonoidea	Cynipoidea	Proctotrupoidea
Eulophidae	**Pteromalidae**	**Eurytomidae**	**Eupelmidae**	**Tetracampidae**	**Ormyridae (1)**	**Braconidae**	**Figitidae**	**Diapriidae (1)**
Eulophinae (5)	**Pteromalinae (4)**	**Eurytominae (2)**	**Eupelminae (2)**	**Tetracampinae (2)**		**Opiinae (2)**	**Eucoilinae (1)**	
Hemiptarsenus	***Trigonogastrella***	*Eurytoma*	*Eupelmus*	*Epiclerus*	*Ormyrus*	***Opius***	***Gronotoma****	Unknown species
Diglyphus	***Callitula***	*Plutarchia*	nr *Anastatus*			***Phaedrotoma***		
Zagrammosoma	*Oxyharma*							
Cirrospilus	*Sphegigaster*					**Alysiinae: Dacnusini (1)**		
Stenomesius	*Syntomopus*					*Dacnusa*		
Entedoninae (7)	**Herbertiinae (1)**							
Neochrysocharis	*Herbertia*							
Closterocerus								
Chrysocharis	**Miscogasterinae (2)**							
Asecodes	***Halticoptera****							
Apleurotropis	*Ammeia*							
Achrysocharoides								
Chrysonotomyia								
Tetrastichinae (3)								
Aprostocetus								
Quadrastichus								
Tetrastichus								

**important overseas but situation in Australia unclear

Table 4. Life history strategies of the putatively most important parasitoid species in Australia

Family names are bolded.

Family/Species	Life history characteristics and potential importance in Australia
Eulophidae	
Hemiptarsenus varicornis	• synovigenic idiobiont ectoparasitoid • widespread in Australia (Bouček 1988) • biology well known from overseas studies (Cheng *et al.* 2017) • *Wolbachia*-infection widespread in Japan (Tagami *et al.* 2006)
Diglyphus isaea	• synovigenic idiobiont ectoparasitoid • adventive species found in SE Australia (Bjorksten *et al.* 2005) • biology well known from overseas studies (Liu *et al.* 2014; Zhang *et al.* 2014) • mass-reared for biological control in protected cropping in Europe and North America
Zagrammosoma latilineatum	• synovigenic idiobiont ectoparasitoid • described from Indonesia and Queensland • no detailed biological studies have been undertaken • abundant in late-summer in SE Australia (Wood 2010)
Neochrysocharis formosa	• synovigenic idiobiont endoparasitoid • very important parasitoid in Asia and North America (Hernández *et al.* 2010; Liu *et al.* 2014, 2015b; Zhang *et al.* 2014) • distribution in Australia needs to be clarified • potentially valuable female-only strain in Japan due to *Rickettsia* sp. infection (Hagimori *et al.* 2006)
Neochrysocharis okazakii	• synovigenic idiobiont endoparasitoid (Tran *et al.* 2007, 2012) • common agromyzid parasitoid in Japan, Vietnam and Indonesia • specimens recorded from Victoria (Bjorksten *et al.* 2005) and Torres Strait • distribution in Australia needs to be clarified
Closterocerus mirabilis	• believed to be synovigenic idiobiont endoparasitoid but little is known of its biology • Described from Victoria where it is common (Bjorksten *et al.* 2005) • not yet recorded from Queensland • suspected as being present in Indonesia (Edwards & La Salle 2004)
Chrysocharis pubicornis	• synovigenic larval-pupal endoparasitoid • has been found in low numbers in South Australia (Lardner 1991) • recorded from New South Wales, South Australia, Tasmania and Victoria (Bouček 1988) • summer diapause recorded in Japan (Baeza Larios & Ohno 2007; Baeza Larios *et al.* 2007)

Family/Species	Life history characteristics and potential importance in Australia
Pteromalidae	
Trigonogastrella parasitica	• endoparasitoid • described from Victoria (Girault 1915b) • common in eastern Australia (Bouček 1988) • biology unknown
Callitula viridicoxa	• believed to be a synovigenic idiobiont but biology remains unstudied • Bouček (1988) notes that is the most abundant *Callitula* sp. in Queensland
Halticoptera sp.	• koinobiont larval-pupal endoparasitoid • genus important in South America for control of *Liriomyza* spp. • Bouček (1988) records several specimens from Australia but they have not been identified to species. A different unnamed *Halticoptera* sp. has been recorded in PNG (Bouček 1988),
Braconidae	
Opius atricornis	• koinobiont larval-pupal endoparasitoid • recorded from several agromyzid species in South Australia (Lardner 1991) and Queensland (Kleinschmidt 1970
Opius cinerariae	• koinobiont larval-pupal endoparasitoid (Kleinschmidt 1970; Lardner 1991; Belokobylskij *et al.* 2004)
Phaedotroma oleaceri	• koinobiont larval-pupal endoparasitoid • recorded from Queensland (Kleinschmidt 1970)
Figitidae	
Gronotoma domestica	• koinobiont larval-pupal endoparasitoid • recorded from Queensland (Bouček 1988), but only two specimens in collections • likely to be a parasitoid of agromyzids (Paretas-Martínez *et al.* 2013) • *Gronotoma micromorpha* is an important parasitoid of *L. trifolii* in Japan (Abe & Tahara 2003)

2.4 Details of Key Parasitoid Species

2.4.1 Eulophidae: Eulophinae

Hemiptarsenus varicornis

Hemiptarsenus varicornis is a polyphagous, multivoltine, solitary idiobiont ectoparasitoid which predominantly oviposits in 3rd instar agromyzid larvae it has paralyzed (Lardner 1991; Chien & Ku 2001a, b). It has been the most commonly recorded parasitoid of agromyzids in Australia: it has been reared from 6 agromyzid species in Queensland (Appendix 2) and has been recorded in every Australian state (Appendix 3). It has also been recorded attacking agromyzids in Asia (southern China, India, Indonesia, Iran, Japan, Malaysia, Pakistan, Saudi Arabia, Sri Lanka, Taiwan, Turkey), Oceania (Cook Islands, Fiji, Guam, Hawaii, New Caledonia, New Zealand, Papua New Guinea, Samoa, Tonga, Vanuatu), Africa (Ethiopia, Ghana, Kenya, Réunion, Senegal, Seychelles, Sudan, Tanzania) and southern Europe (Spain, Madeira, Canary Islands) (CABI 2018a; Noyes 2018). Given that *H. varicornis* has been recorded from a very wide range of latitudes: from 35°N [Shizuoka, Japan (Amano *et al.* (2008) to 42°S [Lake St Clair, Tasmania, Australia (Driessen *et al.* 2013; Driessen 2016;)], an investigation of thermal adaptation (Hoffmann 2010; Overgaard et al. 2011; Hands 2013) of Australian populations of *H. varicornis* along a latitudinal gradient should provide insights into its potential importance as a biological control agent for *L. sativae* in Australia.

In a comprehensive life history study using *L. trifolii* as a host, Cheng *et al.* (2017) observed that host killing by *H. varicornis* occurred in 3 ways: parasitism (26% of mortality), host feeding (58% of mortality) and host stinging, without host feeding (16%). Without access to agromyzid hosts, the adult wasps only survived for 2.7 days at 27°C when only fed water. However, with access to *L. trifolii*, each female *H. varicornis* had a mean longevity of 14.8 days with a mean number of host feeding events of 20.4 per day. The initial mean egg load of a female *H. varicornis* was 0.43 and the lifetime fecundity was 133.9 eggs so the ovigeny index (initial egg load/total fecundity) was 0.003, indicating that *H. varicornis* is strongly synovigenic (Jervis et al. 2001). Bordat *et al.* (1995b) also noted deaths due to host stinging by *H. varicornis*, where adults did not return to the paralyzed larva to either feed or oviposit. The importance of host killing by *H. varicornis* was also observed by Ho & Ueno (2002) and Chien & Ku (2001b).

Ho & Ueno (2011) demonstrated that *H. varicornis* males and females were unable to store carbohydrate sources very well and required daily access to carbohydrate solutions to ensure survival. In an attempt to improve rearing efficiency, Ho & Ueno (2007) mixed milk protein into 30% honey solution but the non-host protein did not alter the fecundity of *H. varicornis* females significantly. This finding supports the conclusion of Giron *et al.* (2004), studying host feeding by the eupelmid, *Eupelmus vuilletti*, who showed that lipids sourced from hosts was the limiting factor for egg production and that ingested host proteins did not in fact have much impact on fecundity (only 10% of ingested proteins went to eggs). The influence of temperature on the development of *H. varicornis* has been examined in multiple studies (Appendix 4) while Chien *et al.* (2004) also examined the influence of temperature on its host-killing capacity and noted increased host feeding above 25°C. Larvae of *L. trifolii* previously parasitized by *D. sibirica* were not avoided by either *H. varicornis* or *D. isaea* and there were no adverse effects on the performance of either of the ectoparasitoids (Mitsunaga & Yano 2004).

In Japan, Tagami *et al.* (2006) tested 15 species of parasitoids of *L. trifolii* and *L. bryoniae* for presence of endosymbionts but *H. varicornis* was the only species in which a *Wolbachia* strain was detected. The *wsp* sequences in *H. varicornis* collected in 1995 and 2004 were identical and the strain was very similar to the strain they detected in *L. bryoniae*. Laboratory trials with the *Wolbachia*-infected *H. varicornis* showed no evidence of cytoplasmic incompatibility or induction of parthenogenesis. It is not known whether Australian populations of *H. varicornis* are infected with *Wolbachia*.

Diglyphus isaea

Diglyphus isaea is distributed naturally very widely in Europe, Asia and parts of Africa (CABI 2018b; Noyes 2018) and is now also found in south-eastern Australia (Bjorksten *et al.* 2005; Lambkin et al. 2008), New Zealand (McGregor 1989; Martin 2017) and Brazil (Carvalho *et al.* 2011). A European strain has been mass-produced for many years for augmentative biological control in glasshouses in Europe, North America and Japan (Chow & Heinz 2004; van der Linden 2004; Hondo *et al.* 2006b). However, while Zhu *et al.* (2000) detected little morphological variation in *D. isaea* in China, Sha *et al.* (2006, 2007) detected at least 4 cryptic species of *D. isaea* in China using mitochondrial and nuclear markers. Hansson & Navone (2017) noted that the Chinese specimens had a shiny violet scutellum which is not seen in any European specimens, but they supported the need for further studies of *D. isaea* to establish whether there are in fact multiple species. Fisher & La Salle (2005) noted that coloration in the Chalcidoidea was quite variable.

Hondo *et al.* (2006b) compared the biology of commercially reared *D. isaea* (European origin) and a local strain. At 20°C and 25°C, the native strain was superior to the imported strain in terms of numbers of parasitoids killed by parasitization rather than host feeding. This is reflected in net reproductive rates of the two strains. Conversely, the imported strain killed many more hosts by host feeding than the native strain. These findings reinforce the need for further studies on assessing efficacy of strains of *D. isaea* for biological control of *Liriomyza* spp. On the basis of a series of temperature-development studies (Appendix 4) and its known distribution (Noyes 2018), it is likely that *D. isaea* will be better suited to southern Australia than northern Australia. Conversely, *Neochrysocharis* spp. are likely to be better suited

to northern parts of Australia than *D. isaea*. It will be important to predict the distribution of *D. isaea* and other key parasitoid species in Australia to complement field surveying.

In China, Zhang *et al*. (2011, 2014) made detailed studies on the importance of host feeding on the biology of *D. isaea* and demonstrated that host feeding by *D. isaea* females resulted in longer survival and higher fecundity. Each host meal yielded an extra 3.5 eggs per day and extended longevity by 0.78 days. Similar to *H. varicornis*, *D. isaea* was highly synovigenic, with an ovigeny index of 0.002. In contrast, *N. formosa* was only moderately synovigenic, with an ovigeny index of 0.12.

In New Zealand, Martin (2017) has recorded *D. isaea* on 8 agromyzid species (4 adventive; 4 native) (Table 5). Martin noted that the parasitoid was active throughout the year in Auckland but has a diapause stage in colder parts of New Zealand. While shipments of *D. isaea* from Pakistan were released in New Zealand around 1969 (McGregor 1989), Valentine & Walker (1991) noted that there were records of the species in New Zealand before these deliberate releases.

With the exception of *Phytoliriomyza jacarandae*, which has yet to be recorded in Australia, *D. isaea* has been recorded in Australia from the three other adventive species present in both countries. There are 11 *Cerodontha* spp. in Australia but there are no rearing records of either the agromyzids or their parasitoids.

Table 5. New Zealand hosts of the parasitic eulophid wasp, *Diglyphus isaea*, (Walker, 1838) (Hymenoptera: Eulophidae), from the Plant-SyNZ database (27 April 2017) (Martin 2017)

Scientific Name	NZ Common Name	NZ Biostatus
Cerodontha sp. (NZ)	Grass leafminer	native
Phytomyza syngenesiae	Ragwort leafminer	adventive
Liriomyza chenopodii	Australian beet miner	adventive
Liriomyza clianthi	Kaka beak leafminer	endemic
Liriomyza watti	New Zealand cress leafminer	endemic
Phytoliriomyza jacarandae	Jacaranda leafminer	adventive
Phytomyza plantaginis	Plantain leafminer	adventive
Phytomyza sp. '*Ranunculus amphitrichus*' of Martin 2011	Water buttercup leafminer	endemic

Zagrammosoma latilineatum

The genus *Zagrammosoma* was characterized by LaSalle (1989) who noted that members of the genus were parasitic on leafmining Lepidoptera and Diptera. Ubaidillah *et al.* (2000) described *Z. latilineatum* reared from *L. huidobrensis* in Indonesia and from a female specimen caught at Emerald, Queensland (host unknown). Fisher *et al.* (2005) noted that it is widespread throughout South East Asia but generally does not appear to be in high enough numbers to supply high levels of biological control. However, it is one of the more abundant parasitoids in SE Australia, particularly in early autumn, and further studies on its biology and ecology would be justified given its known distribution in Australia is extensive (Yorke Island to Melbourne).

2.4.2 Eulophidae: Entedoninae

Neochrysocharis spp.

Three species of *Neochrysocharis* have been recorded attacking agromyzid leafminers in South East Asia. *Neochrysocharis formosa* and *N. okazakii* are well studied, but little is known about the biology and ecology of *N. beasleyi* which was described from specimens reared from *L. huidobrensis* in Indonesia and Vietnam (Fisher & La Salle 2005).

Neochrysocharis formosa is an important parasitoid of leafminers, attacking agromyzids and several families of lepidopteran leafminers (CABI 2018c). Noyes (2018) lists 29 agromyzid species as hosts, together with 52 species of leafmining moths. It is a very important parasitoid of *Liriomyza* spp. in Asia (Chien & Ku 2001c; Chien *et al.* 2005b; Liu *et al.* 2014, 2015b; Zhang *et al.* 2014) and North America (Hernández *et al.* 2010, 2011b). In contrast, *N. okazakii* has been reared from 5 species of agromyzids, including *L. chinensis*, *L. huidobrensis, L. sativae* and *L. trifolii* in Japan, China, Indonesia and Vietnam. Specimens of both species have been identified in low numbers in Australia (Bjorksten *et al.* 2005) but often *Neochrysocharis* specimens collected in Australia have only been identified to genus so the distribution and abundance in Australia needs to be clarified. In New Zealand, *N. formosa* was recorded in 1989 as an unintentional introduction (Ward & Edney-Browne 2015) but its status in New Zealand remains unclear.

Zhang *et al.* (2014) made a detailed comparison of the life histories of *D. isaea* and *N. formosa* and showed that there were significant differences in life history strategies between these two idiobiont parasitoids (Table 6), particularly in terms of immature development time, body size, egg volume and egg load at eclosion.

In Vietnam, where *Neochrysocharis okazakii* is the most important parasitoid of *L. chinensis* on *Allium* spp., Tran & Takagi (2006), Tran *et al.* (2006, 2007) and Ueno & Tran (2015) examined the effect of temperature on development and fecundity on different host plants on the biology of *N. okazakii*. Similar detailed studies have been conducted on *N. okazakii* reared on *L. trifolii* on *Phaseolus vulgaris* in Taiwan (Chien & Chang (2009a, b, 2010, 2011, 2013). In Indonesia, *N. formosa* is more abundant than *N. okazakii* on vegetable crops (Sunari *et al.* 2016).

Table 6. Life history traits of two coexisting parasitoids *Diglyphus isaea* and *Neochrysocharis formosa* (Zhang *et al.* 2014)

Life history traits	*Diglyphus isaea*	*Neochrysocharis formosa*
Ovigeny index	0.002 (extreme synovigeny)	0.12 (relatively weak synovigeny)
Ecto/endoparasitoid	Ectoparasitoid	Endoparasitoid
Idio-/koinobiont	Idiobiont	Idiobiont
Body size	Large	Small
Egg type	Anhydropic egg	Anhydropic egg
Egg volume	Large	Small
Development time and longevity	Short immature stage and long adult longevity	Long immature stage and short adult longevity
Type of host-feeding	Non-concurrent and destructive	Non-concurrent and/or concurrent destructive
Lifetime host feeding	Similar	Similar
Gain per host-feeding event	High	Low
Lifetime fecundity	Large	Small
Host mortality	Large	Small
Age-specific fecundity curve	Slow	Fast

Closterocerus mirabilis

This species was described from Victorian material reared from *Phytomyza syngenesiae*, *L. brassicae* and *L. chenopodii* (Edwards & La Salle 2004). Other *Closterocerus* species recorded in Australia are parasitoids of small leaf-mining larvae of Coleoptera or Lepidoptera (Bouček 1988). Like all entedonines attacking agromyzids, it is an endoparasitoid (Fisher *et al.* 2005) but little known about its biology. Bjorksten *et al.* (2005) found most specimens on *P. syngenesiae*. Lardner (1991) recorded a few specimens of *Closterocerus* sp. on *L. brassicae*. At that time, *C. mirabilis* had not yet been described so it is most likely that specimens

were in fact *C. mirabilis*. However, in contrast to Fisher *et al.* (2005), he considered it to be an idiobiont ectoparasitoid.

Chrysocharis pubicornis

Chrysocharis pubicornis is an adventive koinobiont endoparasitoid recorded in New South Wales, South Australia, Tasmania and Victoria (Bouček 1988). It has also been recorded in New Zealand. Lardner (1991) only recorded a few instances of this parasitoid on *L. brassicae* in South Australia (in spring and autumn). The species is widely distributed through Europe, the Middle East, Japan, Canada and USA. Detailed studies in Japan demonstrated that *C. pubicornis* enters a summer and winter diapause (Baeza Larios & Ohno 2007; Baeza Larios *et al.* 2007). Noyes (2018) records the species from 62 agromyzid species and from 4 families of leafmining lepidopterans.

2.4.3 Braconidae: Opiinae

In Australia, three identified opiine species (Hymenopotera: Braconidae) have been reared from agromyzids in Australia, while Belokobylskij *et al.* (2004) notes that another five species of opiines (collected in Queensland, South Australia and Tasmania) are suspected of being parasitoids of agromyzids (on morphological grounds) (Table 7). However, Lambkin *et al.* (2008) reared 6 morphospecies of *Opius* spp. from *Phytomyza syngenesiae* in a 2-month period in the ACT suggesting that much more work is need on the taxonomy of opiines attacking agromyzids in Australia. Given the difficulties with the group, it will be very important for molecular barcoding to be carried out on a range of Australian samples to assist in species identification and to establish the host range for those opiine species suspected of being parasitoids of agromyzids in Australia.

Opiine parasitoids have been important parasitoids of *Liriomyza* worldwide. Apart from the limited biological studies of Lardner (1991) with *Opius (Gerius) cinerariae* on *L. brassicae*, there is no information about the biology of the three Australian species attacking agromyzids. There have been numerous studies on opiine parasitoids of agromyzids overseas which provide guidance to the Australian situation. In South America, *Phaedotroma scabriventris* (Nixon) is an important solitary, koinobiont, and larva-pupal endoparasitoid of *Liriomyza* spp. (including *L. sativae*). Females lay their eggs in 2nd or 3rd instar *Liriomyza* larvae. The biology of *Opius dissitus* has been studied extensively in several countries. Bordat *et al.* (1995a) examined the influence of temperature on the development of *O. dissitus* and compared the behaviour of *O. dissitus* with *H. varicornis* (Bordat *et al.* 1995b). Life table studies of *O. dissitus* reared on *L. sativae* were undertaken in USA (Petitt & Wietlisbach 1993) and in Bangladesh (Azad & Islam 2015). Behavioural studies were undertaken by Petitt *et al.* (1992) and by Nelson & Roitberg (1993, 1995) who looked at host discrimination by *O. dissitus* and host patch time allocation by *O. dimidiatu*s. Field studies of *O. dissitus* and *L. trifolii* in Florida on snap bean (*Phaseolus vulgaris*) examined spatial distribution of both species and a direct density dependent relationship was found between parasitism by *O. dissitus* and density of *L. trifolii* (Li *et al.* 2012). Foba *et al.* (2015) examined the interaction between the imported *Phaedrotoma scabriventris* Kenyan populations of *O. dissitus*. They concluded that there would be no adverse effects on the indigenous *O. dissitus* when *P. scabriventris* was released in the field. *Opius (Opiothorax) chromatomyiae* was described from material reared from *Phytomyza horticola* and *L. huidobrensis* in Malaysia, Indonesia and Vietnam (Belokobylskij *et al.* 2004). It has been shown to be a very important parasitoid of *L. huidobrensis* in highland areas of Indonesia (Tantowijoyo & Hoffmann 2010).

Table 7. Australian opiine species reared from agromyzids or suspected of being associated with agromyzids

Species	Rearing data	Known hosts
Opius (Adontopius) atricornis Fischer	Yes	*Calcomyza humeralis*, *Liriomyza brassicae*, *Ophiomyia lantanae*, *Ophiomyia wikstroemiae*, *Tropicomyia polyphyta*
Opius (Gerius) cinerariae Fischer	Yes	*Calcomyza humeralis*, *Liriomyza brassicae*, *Liriomyza chenopodii*, *Ophiomyia pisi*,
Phaedrotoma oleracei (Fischer)	Yes	*Calcomyza humeralis*, *Phytomyza syngenesiae*, *Ophiomyia phaseoli*
Opius (Merotrachys) penetrator Fischer	No	n.a.
Opius (Allophlebus) sanctannae Fischer	No	n.a.
Phaedrotoma australicola (Fischer)	No	n.a.
Phaedrotoma kangarooensis (Fischer)	No	n.a.
Phaedrotoma portarthurensis (Fischer)	No	n.a.

Data from Kleinschmidt (1970); Lardner (1991); Bjorksten *et al.* 2005; Belokobylskij *et al.* (2004)

2.4.4 Braconidae Alysiinae: Dacnusini

Dacnusa aerolaris is an adventive species in Australia and New Zealand where it has been reared exclusively on *P. syngenesiae* (Kelsey 1937; Wharton & Austin 1991). In Australia, there are specimen records from South Australia, Victoria, ACT, Tasmania and New South Wales (Wharton & Austin 1991). Griffiths (1966) noted that the species had only been definitely recorded from 3 *Phytomyza* spp. (*C. asteris*, *C. nigra* and *P. syngenesiae*) and considered that records from a range of other agromyzids and non-agromyzids were erroneous. This suggests that it is highly unlikely that *Dacnusa aerolaris* will parasitize *L. sativae* in Australia. No other *Dacnusa* species have been recorded in Australia and New Zealand.

In Europe, USA and Japan, *Dacnusa sibririca* is reared commercially for release in glasshouse crops targeting *L. trifolii* and *L. bryoniae*. This species has a lower temperature threshold than the other reared parasitoid, *D. isaea,* and is released early in the season to control *Liriomyza* spp. overwintering in the glasshouses (in many parts of Europe, North America and Northern Asia, *Liriomyza* spp. can only overwinter successfully in greenhouses). Abe *et al.* (2005) found that European-sourced *D. sibirica* successfully attacked *L. trifolii* and *L. bryoniae* but was encapsulated by *L. sativae*. Abe & Tokumaru (2008) suggested that this differential mortality was an important reason for the displacement of *L. trifolii* by *L. sativae* in Japan.

2.4.5 Pteromalidae

***Callitula* spp.**

Bouček (1988) found *Callitula viridicoxa* was a very common species in eastern Australia (Queensland, New South Wales, Victoria and South Australia) and that it had been reared from *Ophiomyia phaseoli,* found in the stems of cowpea (*Vigna unguiculata*) in Queensland. Bouček (1988) listed another 7 Australian *Callitula* species without biological data. He noted that *Callitula* spp. are parasitoids of small Diptera, especially Agromyzidae, and more commonly parasitized host larvae living within a grass stems or leaves. In tropical areas, the main hosts were leafmining or stem-mining species on herbaceous plants. Makinson *et al.* (2005) recorded a *Callitula* sp. from the pigeonpea pod fly, *Melanagromyza obtusa*, from the endemic *Cajanus latisepalus* in the Northern Territory but no estimate of abundance of the parasitoid was made. In South Australia, a *Callitula* sp. was reared from *Phytoliriomyza praecellens* on *Chenopodium (=Rhagodia) candolleana* and *C. parabolica* (Wood *et al.* 2010).

***Trigonogastrella* spp.**
Trigonogastrella parasitica was described from specimens in Victoria (Girault 1915b) and is recorded from *Liriomyza* spp., *Ophiomyia spp.*, *Phytomyza plantaginis* and *P. syngenesiae*. It is recorded from Queensland, NSW, Victoria and Tasmania. Bouček (1988) included another species, *T. rabiosa,* (originally described by Girault (1915b) as *Cryptoprymnoides rabiosus*) in the genus. This species has been recorded in Queensland, NSW, South Australia and Western Australia. Given the prevalence of *T. parasitica* in southern Australia, it will be important to learn more of its biology and ecology. It may prove to be an ecological analogue of the *Halticoptera* spp. which are important in the biological control of *Liriomyza* spp. in North and South America.

***Herbertia* spp.**
Very little is known about the biology and ecology of the genus *Herbertia* (Salvo & Valladares 2000). Greathead (1969) concluded that the wasps (of a pteromalid nr *Herbertia*) oviposited in the pupae of *Ophiomyia phaseoli* and *O. spencerella* and emerged from the same state of the host, so behaving as an idiobiont. It is unlikely that this genus will be of great significance for the biological control of *L. sativae*. In Australia, *Herbertia setosa* is the more common species (originally described by Girault (1915a) as *Tetracampoides setosa* from a specimen caught in Babinda, North Queensland), with one other unnamed species present (Bouček 1988).

2.4.6 Figitidae: Eucoilinae
Gronotoma domestica was described by Girault (1932) from 2 specimens. The type female was captured at a window at Indooroopilly, and a male was captured at Bowen. Nothing has been published about its ecology. Buffington (2002) noted that the location of the type specimen was unknown but Paretas-Martínez *et al.* (2013) listed the holotype as being in the Queensland Museum. *Gronotoma* and *Weldia* are parasitoids of leafmining Agromyzidae but no host records exist in Australia to date although specimens of both genera have been recorded (Paretas-Martínez *et al.* 2013). Buffington (2002) also noted that all *Gronotoma* species reared so far have been from agromyzids. This suggests that *G. domestica* and the other unnamed *Gronotoma* species known in Australia (Paretas-Martínez *et al.* 2013) will also be parasitoids of agromyzids. Buffington (2002) also noted that most *Gronotoma* spp. had been reared from *Melanagromyza* spp. However, *G. micromorpha* is an important koinobiont endoparasitoid of *L. huidobrensis* in Indonesia (La Daha 2011), and of *L. trifolii* in Japan (Abe 2001, 2006) and Guam (Johnson 1993). It is conceivable that this parasitoid might have colonised northern Australia. There are *Wolbachia*-induced thelytokous strains of *G. micromorpha* in Japan (Arakaki *et al.* 2001) and in Indonesia (La Daha 2011) so potentially this species could be a useful candidate for mass-rearing for augmentative release if it is recovered in northern Australia and if it is a thelytokous population.

2.5 Seasonality of Australian agromyzids and their parasitoids
In all the Australian studies, the occurrence of agromyzids and their parasitoids was restricted seasonally. It will be vital to gain further knowledge about the diapause and quiescence strategies the flies and their parasitoids have for survival in the Australian environment. In Europe and North America, agromyzids typically overwinter as diapausing pupae *e.g. Agromyza frontella* (Nechols *et al.* 1983; Therrien *et al.* 1985). *Phytomyza orobranchia* is used as a biological control agent for the root parasitic plant *Orobanche* spp. in the former Soviet Union. In this species, diapause is induced by a day-length of less than 14 hours, and the percentage of diapausing pupae increases with declining temperatures. Diapause can last for up to 4 years (Klein & Kroschel 2002).

In southern Australia, *P. syngenesiae* is abundant in late spring and early summer but populations decline rapidly in January (Bjorksten *et al.* 2005; Lambkin *et al.*2008) despite abundance of hosts such as *Sonchus oleraceus*. The mechanisms for the survival of *P. syngenesiae* and its parasitoids over summer and winter are not yet known. A related European species (but one where pupation occurs off the plant) *P.*

chaerophylli is multivoltine and exhibits both winter and summer diapause regulated by temperature and photoperiod (Frey 1991). Spencer (1977a) noted that two Western Australian species (*Liriomyza oleariana* and *L. scaevolae*) went through summer in the pupal stage. Both species have their puparia remaining in the leaf (unlike *L. sativae* but similar to *P. syngenesiae* and *P. plantaginis*). *Liriomyza cicerina*, an important pest of chick peas in the Middle East, emerges from winter diapause in March and April (Weigand & Tahhan 1990; Çikman & Civelek 2006) and is suspected of entering summer diapause as well (Weigand & Tahhan 1990).

Parasitoids also require survival mechanisms to overcome winter and summer temperatures as well as a lack of hosts. In Japan, the existence and mechanism of summer diapause in the eulophid, *Chrysocharis pubicornis* (a species which was also recorded from South Australia - Lardner 1991) has been demonstrated (Baeza Larios & Ohno 2007; Baeza Larios *et al.* 2007). When the drupe-feeding agromyzid *Ophiomyia lantanae* was brought to Hawaii from Mexico as a biological control agent for lantana, *Lantana camara*, Bridwell (1919) noted that *Opius lantanae* emerged from the agromyzid in two distinct time periods, 9 months apart, indicating that the parasitoid had a facultative diapause.

> *"This period of dormancy will need to be considered in future work in the introduction of this Agromyzid into other countries, for not unfrequently material supposed to have had all the insects bred out from it may still contain parasites which may escape through negligence."* Bridwell (1919)

While *Opius lantanae* was described from Hawaiian material collected soon after the release of *Ophiomyia lantanae*, it seems certain that *Opius lantanae* actually came from Mexico with its host (Bianchi 1940).

Given the climatic diversity in Australia, it is highly likely that the relative abundance of parasitoids will vary greatly between regions. It will be valuable to model the likely distribution of a number of adventive parasitoids in Australia using overseas and the limited Australian data on temperature development for key parasitoids on a range of agromyzid hosts. Appendix 4 provides summary thermal development statistics for a number of key parasitoid species relevant to Australia and the studies provide development data at a range of temperatures which will be useful for modelling potential distribution of these parasitoids. The braconid, *Opius caricovarae*, is an important parasitoid of *Liriomyza* spp. in China, and is included to show that its development is very similar to that found in Australia with *O. cinerariae* (attacking 3rd instar *L. brassicae*) by Lardner (1991). It will be important to study Australian populations of parasitoids to refine our understanding of their thermal biology in different regions in Australia. Apart from Cheah (1987), the data for *D. isaea* are very similar between studies.

3. Identifying potential reservoirs for parasitoids in key horticultural regions

Given the known host range of *L. sativae* is so large (Spencer 1973, 1990; Benavent-Corai *et al.* 2005), and also that it is likely to colonize novel hosts once it establishes widely in Australia, there will be ample opportunity for parasitoids to thrive in non-cropping situations where insecticides are not used. This will reduce the overall populations of *L. sativae* in a region and also act as parasitoid reservoirs for cropping systems.

In the current absence of *L. sativae* in the general Australian landscape and with the belief that the generalist agromyzid parasitoids will move onto other invading agromyzids, it will be essential to sample a range of other agromyzid species in key horticultural regions to determine their parasitoid assemblage at various times of the year.

3.1 Australian Agromyzidae

There are currently 2,994 Agromyzidae species described worldwide (Pape & Thompson 2018) and 151 Agromyzidae species known in Australia (Table 8, Appendix 5) (Elliott 2011; Lonsdale 2014), suggesting that Australia has a low species diversity relative to other countries with a similar climate (Spencer 1977a,

1992; Lambkin *et al.* 2008). Spencer (1977a) noted that a wide range of endemic plants in Australia had not been colonized by Agromyzidae and speculated that that "*the Agromyzidae are relatively recent arrivals to Australia - early Pleistocene or late Tertiary - and this period has not been sufficiently long for the colonization of unfamiliar hosts to have developed".* He also noted that in the Northern Hemisphere, many plants in the Fabaceae were colonized by Agromyzidae but that in Australia the endemic Fabaceae mainly had leafmines caused by micro-Lepidoptera but very few by Agromyzidae.

Table 8. Numbers of Australian agromyzid species with known hosts, broken down by Sub-family

Sub-family	Agromyzid species with known hosts	Agromyzid species with unknown hosts	Total number of agromyzid species in Australia	% species with known hosts
Agromyzinae	27	44	71	38.0%
Phytomyzinae	24	56	80	30.0%
Total	**52**	**100**	**151**	**33.8%**

In Appendix 5, which details the known species of Australian Agromyzidae, not all species are leafminers. Many species are, or are suspected of being, shoot miners or gall formers. There has been limited collecting in Australia, so it is very likely that more species will be discovered and described, while the distribution of known species will be extended. Most (66%) of Australian agromyzid species have no host plant data (Table 8; Appendix 5). Many species were described from adults caught at lights or collected by sweeping. Worldwide, the vast majority of agromyzid species are very host-specific.

Appendix 6 gives details of all known agromyzid species recorded in Queensland and is included to give direction on potential agromyzid species to target for parasitoids in the short-term, given the location of the only known infestation of *L. sativae* in Seisia. It must be stressed that many of these agromyzids do not yet have a known host.

Notes are given on important agromyzid hosts to target collecting for parasitoids in Australia.

3.2 Agromyzids common on weeds in southern Australia

In southern Australia, common agromyzids such as *L. chenopodii* (on chenopod weeds), *L. brassicae* (on brassicaceous weeds) and *Phytomyza syngenesiae* (usually on *Sonchus oleraceus*) should act as important reservoirs for populations of parasitoids (*e.g. Diglyphus isaea*) of *L. sativae* and other invasive polyphagous agromyzids (Bjorksten *et al.* 2005; Lardner 1991). Further sampling of *P. syngenesiae* and *P. plantaginis* is needed to delineate the northern boundary for *Diglyphus isaea*. Kleinschmidt (1970) recorded *P. syngenesiae* as far north as the Tropic of Capricorn. *Sonchus oleraceus* has become an important weed in cropping areas in the northern grain areas and it a known host of *L. sativae*.

3.3 *Tropicomyia polyphyta*

Tropicomyia was erected for a small group of epidermal leafminers extending throughout the Tropics of the Old World to northern Australia (Spencer 1973). Lonsdale (2014) re-defined the genus *Tropicomyia* but conceded that further work was needed in this problematic genus. Despite some earlier taxonomic confusion when it was synonymized with *Tropicomyia atomella* (Spencer 1973)*, T. polyphyta* (Kleinschmidt, 1960) is the sole Australian species in the genus. Dempewolf (2004) noted that the identification of *Tropicomyia* species was very difficult and that external characters alone will not differentiate species. In light of the extreme polyphagy of *T. polyphyta*, it would be very informative to undertake molecular barcoding of specimens from a range of hosts, together with rearing parasitoids from the various host plants supporting *T. polyphyta*.

Kleinschmidt (1960, 1970) described and provided considerable information about *T. polyphyta* (as *Melanagromyza polyphyta*). Unusually, the larvae mine in the epidermal cells, leading to a distinctive silvering of the leaf. Initially, the mine is linear in the upper epidermis but can spread to a blotch mine when the larva reaches the third (and final) instar (Kleinschmidt 1965). Spencer (1990) considered that *T. polyphyta* was restricted to Australia, Papua New Guinea and Pacific islands.
Spencer (1977a) noted:

> "*The only areas where this species is known are around Sydney and southwards to Waterfall and Clyde Mt, around Brisbane and a Darwin. This, however, in no way reflects its true distribution but mainly indicates where collecting has been undertaken- around Brisbane by Kleinschmidt and at the other localities by myself.*" Spencer (1977a).

In all, 56 plant species have been recorded as hosts of *T. polyphyta* in Australia (Appendix 7). Of these, 27 species (48%) are non-indigenous to Australia. The species come from 35 families (25 Orders). Passifloraceae (5 species), Apocynaceae (4 species), Fabaceae (4 species), Euphorbiaceae (3 species), Meliaceae (3 species) are the families most represented. Kleinschmidt (1970) noted that the indigenous host species were mined more extensively than the exotic host species in SE Queensland. The larvae have been frequently observed to be heavily parasitized but very few identifications have actually been recorded (Kleinschmidt 1970). There are 3 records of the braconid, *Opius (Adontopius) atricornis* and single records of the eulophids *Chrysonotomyia* sp. (as *Achrysocharis* sp.) and *Achrysocharoides* sp. (as *Enaysma* sp.) (Kleinschmidt 1970). In many cases (19 of 28 hosts) no adults were reared from mined leaves so the host range for *T. polyphyta* was inferred from the study of the mines and immature stages (Kleinschmidt 1970). This does mean that some of the host range recorded for *T. polyphyta* in Queensland lacks substantiation and requires additional study. Further collections and rearings of *T. polyphyta* north of Brisbane would show whether the species will act as a useful reservoir for parasitoids for *L. sativae.*

3.4 Agromyzids attacking lantana in Australia

Lantana (*Lantana camara*) was introduced into Australia in the mid 1800s as an ornamental plant, but the taxonomically complex species (Day *et al.* 2003b; Swarbrick *et al.* 1998) is a significant weed in both natural ecosystems and agricultural areas. It is currently estimated to cover 5 million ha in Australia (QDAF 2016) and is primarily found in the coastal and subcoastal areas from far-north Queensland to Sydney (Day *et al.* 2003a, b).

Three species of agromyzid flies (*Ophiomyia lantanae* (a drupe feeder*)*, *Calycomyza lantanae* (blotch leaf miner) and *O. camarae* (herringbone leafminer) have been introduced as biological control agents for *L. camara* in Australia (Day 2012). Day *et al.* (2003a) surveyed *Lantana* for presence of biological control agents from Cooktown in far north Queensland to south of Sydney. *O. lantanae* and *C. lantanae* were established along the full extent of the *Lantana* distribution but were more abundant in the northern zones. *Ophiomyia camarae* has only been recently released in Australia but is established between Cooktown and Bundaberg (Day 2012). The lantana leafminers are most abundant in late summer and autumn in the tropical areas (Day 2012). *Liriomyza brassicae* and *L. trifolii* have also been recorded from *Lantana* in California (Lonsdale 2011) but to date there is no information on parasitism for these host/agromyzid combinations. There are a number of unknowns that remain. Are these species a potential source of parasitoids for *L. sativae*? Can dispersal data from any of these species provide useful information for management of *L. sativae*? Are these agromyzids a further impediment to seeking to introduce exotic parasitoids for control of *L. sativae*?

Ophiomyia lantanae were collected on *L. urticifolia* in Mexico and sent to Hawaii in 1902 for release against *L. camara*. After the fly established successfully in Hawaii, it was subsequently released in Fiji in 1911 and Australia in 1914. It is now found on *Lantana* in many parts if the world, often without deliberate introductions (Day 2012). While widespread, it does not appear to be having a major impact on the viability

of the *Lantana* seed (Broughton 1999). Kleinschmidt (1970) recorded *Opius atricornis* from *O. lantanae* but there are very few records of parasitoids from Australia.

Calycomyza lantanae was introduced and established in Australia in 1974 (Taylor 1989) The fly prefers warm, moist areas, with most damage occurring on actively growing shoots. In Australia *C. lantanae* is found throughout the lantana infestations of Queensland and northern New South Wales. It is rarely found in the temperate regions of NSW (Taylor 1989; Day *et al.* 2003a). In South Africa, it is frequently reported to be heavily parasitized but few identified records exist (Baars & Neser 1999). The eulophid, *Cirrospilus brevicorpus* (originally described as *C. ambiguus*) was described from *L. trifolii* in Taiwan and from *C. lantanae* from Africa (Hansson & LaSalle 1996). The species was observed by John La Salle on one sample from Warraber Island in the Torres Strait in 2008. It is not known if *C. brevicorpus* attacks *C. lantanae* in North Queensland.

3.5 Agromyzids attacking *Chenopodium* spp.

Wood *et al.* (2010) found high levels of parasitism of *Phytoliriomyza praecellens* on plantings of seaberry saltbush (*Chenopdium* (syn. *Rhagodia) candolleana*) and *C. parabolica* in Virginia, SA. These saltbushes occur naturally along the coast in SA, Victoria and NSW. There has been very little research looking at this system. It is unclear if other agromyzids which attack the sub-family Chenopodioideae, such as *Liriomyza chenopodii,* also attack these saltbushes. It is also unclear if *P. praecellens* (described from South Australia) occurs in Victoria and NSW.

3.6 Agromyzids attacking grasses

Eleven *Cerodontha* spp. have been recorded in Australia but there are no records of parasitism. Spencer (1977a) only records a host for one *Cerodontha* species – *Cerodontha (Icteromyza) triplicata* which was recorded from Queensland. Lambkin *et al.* (2008) found that *Cerodontha* (*Cerodontha*) *milleri* was the most abundant agromyzid caught in Malaise and emergence traps in Tallanganda National Park, NSW. It appeared to be closely associated with the sweet vernal grass, *Anthoxanthum odouratum*, (although no mines were observed). Spencer (1973) re-examined specimens previously identified as *C. australis* in Australia and found that most of them were in fact a different species, *C. milleri*. While there are no confirmed host records for *C. milleri*, Spencer (1973, 1977a) considers that the species will be a grass-feeder. This species is frequently trapped (Lambkin *et al.* 2008; White 1970) [*n.b.* White (1970) nominated the trapped species in South Australia as *C. australis* but, on the basis of known distribution records in Australia, the specimens trapped in the drogue-net in South Australia would have almost certainly have been *C. milleri*]. Martin (2007) recorded *C. angustipennis* in spring and summer in New Zealand predominantly from tall oat grass (*Arrhenatherum elatius*), Yorkshire fog (*Holcus lanatus*), and winter grass (*Poa annua*). He did observe larvae in winter, so he hypothesized that populations post-summer were being restricted by parasitoids. He noted that *C. angustipennis* was heavily parasitized in summer by parasitoids from four families, Braconidae, Diapriidae, Eulophidae and Pteromalidae. Martin (2017) recorded *D. isaea* from a New Zealand *Cerodontha* sp. Since *Cerodontha* spp. are potentially widespread through Australia, even if in relatively low density, these species could be a potential reservoir for parasitoids of *L. sativae*.

4. Is there a need to import of exotic parasitoids into Australia?

4.1 Classical Biological control programs overseas

Classical biological control of invasive *Liriomyza* spp. has primarily been undertaken on islands which may be depauperate in terms of endemic agromyzids and parasitoids. The major examples are discussed below.

4.1.1 Hawaii

After *Liriomyza* spp. became important pests of Hawaiian horticultural crops from the 1970s, 17 parasitoid species were imported from mainland USA and Mexico between 1975 and 1978 (Johnson 1987; Funasaki *et*

al. 1988). Two of the species, *Diglyphus begini* and *Chrysocharis oscinidis* had in fact been recorded from Hawaii since 1906 and 1910, respectively (Timberlake 1924). The most successful releases were:

1. *Banacuniculus utilis* (Beardsley) (Hymenoptera: Figitidae: Eucoilinae) (it was initially known as *Cothonaspis* n. sp. at time of importation, then as *Ganaspidium hunteri* and then as *Ganaspidium utilis*) (Buffington 2010).
2. *Neochrysocharis diastatae* (Howard) (Hymenoptera: Eulophidae: Entedoninae) (known as *Chrysonotomyia punctiventris* (Crawford) at time of importation). (Johnson 1993).

Johnson & Mau (1986) found that the most important parasitoids of *L. sativae* in watermelon in Hawaii were *B. utilis*, *N. diastatae*, *Chrysocharis oscinidis* [all introduced in 1970s] and *Halticoptera circulus* (Hymenoptera; Pteromalidae: Miscogastrinae) [already present in Hawaii]. Another 5 species (totalling less than 10% of rearings) were also recorded. They noted that relative abundance of the species varied with season.

4.1.2 Guam

When *L. trifolii* began to displace *L. sativae* in Guam around 1980 (Schreiner *et al.* 1986), *B. utilis* was introduced on Guam where it became established as the most abundant parasitoid (78% parasitism) of *L. trifolii* on unsprayed beans (*Vigna unguiculata* and *Phaseolus vulgaris*) (Johnson 1993). Prior to its release, Schreiner *et al.* (1986) recorded two eulophid species - *Hemiptarsenus varicornis* (as *H. semialbiclavus*) and *Neochrysocharis formosa* (as *Chrysonotomyia formosa*) as the dominant parasitoids on beans, while 2 eucoiline species *Gronotoma micromorpha* (Perkins) and *Disorygma pacifica* (Yoshimoto) comprised only 2% of the parasitoid fauna reared from beans. *Diglyphus begini*, sourced from California, was released in 1985 but did not appear to have established (Nafus & Schreiner 1989).

4.1.3 Tonga

Liriomyza trifolii was found in Tonga in 1985 and was damaging a wide range of horticultural crops. *Banacuniculus utilis* and *Chrysocharis oscinidis* Ashmead (previously known as *C. parksi*) were obtained from Hawaii in 1988 for release. Both species were considered to be established by 1989 and chemical control was no longer considered necessary for *L. trifolii* (Johnson 1993). It would be very instructive to return to Tonga to see the current status of *Liriomyza* spp. and their parasitoid assemblage.

4.1.4 Senegal

A classical biological control program to combat *L. trifolii* in Senegal commenced in 1983 with 9 parasitoid species being sourced from Trinidad, USA (Hawaii, California and Florida), and Colombia. Only one species, the braconid *Opius dissitus* established successfully. By the time of release of the imported parasitoids, the complex of 9 species of indigenous parasitoids (including *H. varicornis*, *N. formosa* and *D. isaea*) were controlling the leafminer satisfactorily (Neuenschwander *et al.* 1987).

4.1.5 Kenya

Liriomyza huidobrensis has become the dominant agromyzid pest in Kenya in recent years with 94% of all *Liriomyza* specimens collected in a survey between November 2011 and November 2012 (Foba *et al.* 2015). Three important parasitoid species attacking *L. huidobrensis* in Peru (Mujica & Kroschel 2011; Mujica 2016) and Argentina (Valladares *et al.* 1999) are the braconid, *Phaedrotoma scabriventris* (Nixon), the pteromalid, *Halticoptera arduine* (Walker) and the eulophid, *Chrysocharis flacilla* (Walker). These three species were imported into Kenya in 2008 for control of *L. huidobrensis* and have now been evaluated and released (Musundire *et al.* 2011; Akutse *et al.* 2015; Mujica *et al.* 2016a, b, c; Muchemi *et al.* 2018a, b, c, d). Prior to this program, observed parasitism of *Liriomyza* spp. in Kenyan vegetable crops by indigenous parasitoids, including *H. varicornis*, *D. isaea* and *Opius dissitus*, was low (5%), although these data were from indiscriminately sprayed commercial crops (Foba *et al.* 2015). Subsequent surveys of crops where indiscriminate use of broad-spectrum insecticides was avoided found that the overall parasitism was 31% (Foba *et al.* 2016). *Diglyphus isaea* was the dominant parasitoid at low (14%), mid (20%) and high elevations (21%) in vegetable production systems in Kenya (Foba *et al.* 2016). *Phaedrotoma scabriventris*

had already established widely and was the second most abundant parasitoid in the mid- and low-elevation fields (Foba 2015). It is too early for the value of this classical biological control program to be assessed but success will certainly rely on farmers being convinced to reduce use of broad-spectrum insecticides (which will also assist the indigenous parasitoids to play a significant role in controlling *L. huidobrensis*).

4.1.6 New Zealand

In New Zealand, a spectacularly ill-planned biological control program was conducted against "brassica leafminers" on forage brassicas between 1969 and 1971 (McGregor 1989). It was assumed that the target leafmining pest was the agromyzid *Phytomyza syngenesiae* but in fact, once the project approached completion, the actual pest was identified as the drosphilid, *Scaptomyza flava* (Fallen) which was not attacked by agromyzid parasitoids (McGregor 1989; Martin 2004). The Commonwealth Institute of Biological Control in Pakistan supplied many consignments of adult parasitoids as well as parasitized larvae of *P. horticola* and *Liriomyza brassicae* to New Zealand for release. Among the parasitoid species released were *Hemiptarsenus varicornis* and *Diglyphus isaea*, which in fact were both known to occur in New Zealand prior to the Pakistani material being released (Valentine & Walker 1991; Ward & Edney-Browne 2015). *Chrysocharis pubicornis* was already present in New Zealand and it is not clear which *Chrysocharis* sp. was sent from Pakistan. *C. pubicornis* and *C. gemma* remain the only *Chrysocharis* species recorded in New Zealand. No establishment was recorded for *D. minoeus* (Walker), *D.* sp. nr. *chabrias* (Walker), *Tetrastichus* sp. and *Opius* sp. which were also released in the project. It would be very useful to use molecular techniques to compare populations of *D. isaea* and *H. varicornis* from New Zealand (to see if the Pakistan introductions can be distinguished) and also to compare with Australian populations.

4.2 The Australian situation

Given that 70% of Australian species of Agromyzidae have no host plant records, there would be significant difficulties for conducting host-specificity tests for any potential imports of exotic agromyzid parasitoids for a classical biological control programme in Australia. In addition, agromyzid parasitoids tend to be not host-specific and indeed some species are known to attack lepidopteran leafminers as well as Agromyzidae. Again, this lack of host-specificity will make importation of potential agromyzid parasitoids very difficult because of potential impact on non-target species of agromyzids.

Despite the lack of detailed surveying for parasitoids of agromyzids in Australia, it is clear that there is large number of generalist agromyzid parasitoids which potentially will attack *L. sativae* on its yet-to-be established host plant range in Australia. Until the indigenous fauna is assessed for capability, it would be premature to initiate the costly exercise in importing exotic parasitoids.

Fortuitously, *Diglyphus isaea,* the prime target parasitoid species for mass-rearing based on European and USA experiences, is already present in Australia. It was first found in Sydney, NSW, in 2001 and has since been found, in Victoria, Tasmania and ACT, frequently on *Phytomyza syngenesiae* infesting sow thistle, *Sonchus oleraceus*.

5. Knowledge Gaps

There are many unknowns and research questions requiring further attention and here we focus on a few that should have priority.

How widespread are Diglyphus isaea *and* Hemiptarsenus varicornis *in Australia?*

Sow thistle (*Sonchus oleraceus*) is known from Tasmania to Cairns and is becoming an important cropping weed due to glyphosate resistance. This would be a good host plant to screen for *P. syngenesia*e and *D. isaea*. Seasonal abundance in different regions of Australia needs to be established. Other known host species for *D. isaea* in Australia are *L. brassicae*, *L. chenopodii*, *P. plantaginis* and *P. syngenesiae* but other hosts need to be considered along with potential genetic variation within this parasitoid and potential entry

routes into Australia.

Hemiptarsenus varicornis is widespread (from the Torres Strait to Tasmania) and has been recorded from many agromyzid hosts. The species is also found in Asia, the Pacific, Africa and southern Europe. Seasonal abundance and genetic variation within *H. varicornis* should also be considered, along with ascertaining its full host range.

What other agromyzid species in Australia will act as reservoirs of parasitoids for L. sativae*?*

As discussed above, there seems to be potential for the 3 agromyzid species released against lantana (*Ophiomyia lantanae*, *O. camarae* and *Calycomyza lantanae*) to represent important parasitoid reservoirs, particularly in Queensland. The record from Kleinschmidt (1970) of *Opius atricornis* from *O. lantanae* needs to be validated. *Phytoliriomyza praecellens* infesting seaberry saltbush (*Chenopodium* (syn. *Rhagodia) candolleana*) and *C. parabolica* in coastal areas also need to be investigated, given that Wood *et al.* (2010) found high levels of parasitism in plantings of *C. candolleana* and *C. parabolica* in Virginia, SA. *Cerodontha* spp. (grassminers), which are widespread in Australia, could also be attacked by useful parasitoids given that Martin (2017) records *D. isaea* from a New Zealand *Cerodontha* sp.

Which parasitoids attack Tropicomyia polyphyta*?*

This highly polyphagous agromyzid species has been recorded from 33 plant families (and 55 host species). Spencer (1977a) believes it is widespread in Queensland, despite lack of records. It was the dominant agromyzid recorded in the recent study in SE Qld and Northern NSW by Maunsell *et al.* (2017).

Are any endemic parasitoids suitable for mass-rearing for augmentative release?

Endemic species such as *Zagrammosoma latilineatum*, *Closterocerus mirabilis*, *Neochrysocharis* spp., *Trigonogastrella parasitica* and *Cirrospilus brevicorpus* appear to commonly attack agromyzids in Australia and it would be worth investigating the biology of these common species further to assess their potential use for mass rearing. There may be ways of improving mass rearing through identificationof thelytokous strains, minimizing host loss due to feeding, and using new host species.

What is the taxonomy and biology of the Phaedrotoma *spp. and* Opius *spp. attacking agromyzids in Australia?*

There is a need to develop effective identification tools for non-specialist entomologists for opiine parasitoids attacking agromyzids in Australia and to establish whether *Opius (Merotrachys) penetrator*, *O. (Allophlebus) sanctannae*, *Phaedrotoma australicola, P. kangarooensis* and *P. portarthurensis* are in fact parasitoids of Agromyzidae in Australia.

What are additional host plants for agromyzids in Australia and how do they link to host records for parasitoids?

Spencer (1977a) records host plants for only 34% of identified species. Extensive sampling for agromyzids and parasitoids will provide very important host plant and host agromyzid records. Previously unknown agromyzid species and parasitoids are likely to be found. It is important that they are properly documented, including DNA barcoding.i

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7. Appendices

Appendix 1. Parasitoid species reared from *Liriomyza sativae.*

Specimens only identified to genus or reared from mixed collections of *Liriomyza* spp., which included *L. sativae*, were not included in the list. As with any compilation of species names from the scientific literature, there will be uncertainty about the validity of some species and sampling intensity varies dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem (Noyes 1994; Shaw 1994). Given the nomenclatural changes that have occurred in time, the currently accepted name is given, together with the name used in the original reference.

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Endedoninae	*Achrysocharoides zwoelferi* Delucchi, 1954	*Achrysocharoides zwoelferi*	USA	Gates *et al.* (2002)
		Asecodes deluchii (Bouček, 1971)	*Asecodes deluchii*	Japan	Amano *et al.* (2008)
			Asecodes deluchii	Vietnam	Tran (2009)
		Asecodes erxia (Walker, 1848)	*Asecodes erxia*	Japan	Amano *et al.* (2008)
		Chrysocharis ainsliei Crawford, 1912	*Chrysocharis ainsliei*	USA	Johnson *et al.* (1980a), Chandler (1982)
		Chrysocharis caribea Bouček, 1977	*Chrysocharis caribea*	Barbados	Bouček (1977)
			Chrysocharis caribea	Peru	Mujica & Kroschel (2011)
			Chrysocharis caribea	Trinidad	Bouček (1977)
			Chrysocharis caribea	Venezuela	Bouček (1977)
		Chrysocharis crassiscapus (Thomson, 1878)	*Chrysocharis mallochi*	Canada	McClanahan (1977)
		Chrysocharis flacilla (Walker, 1842)	*Chrysocharis flacilla*	Peru	Mujica & Kroschel (2011)
		*Chrysocharis girault*i Yoshimoto, 1973	*Chrysocharis girault*i	USA	Oatman & Johnson (1981)
		Chrysocharis oscinidis Ashmead, 1888	*Chrysocharis oscinidis*	China	Xu *et al.* (1999)
			Chrysocharis parksi	USA	Johnson *et al.* (1980a), Johnson (1987)
			Chrysocharis viridis	Canada	McClanahan (1977)
		Chrysocharis pentheus (Walker, 1839)	*Chrysocharis pentheus*	Bangladesh	Mazumdar & Bhuiya (2016)
			Chrysocharis pentheus	China	Xu *et al.* (1999)
			Chrysocharis pentheus	Japan	Amano *et al.* (2008)
			Chrysocharis pentheus	Taiwan	Chien & Chang (2011)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Vietnam	Tran (2009)
		Chrysocharis vonones (Walker, 1839)	*Chrysocharis brethesi*	Peru	Mujica & Kroschel (2011)
			Chrysocharis vonones	Brazil	Costa-Lima *et al.* (2014)
		Chrysonotomyia reticulata Zheng & Zhan 2000	*Chrysonotomyia reticulata*	China	Sheng & Zhan (2000)
		Closterocerus cincinnatus Girault, 1916	*Closterocerus cincinnatus*	USA	Gates *et al.* (2002)
		Closterocerus cinctipennis Ashmead	*Closterocerus cinctipennis*	USA	Harding (1965), Stegmaier (1972)
		Closterocerus purpureus (Howard, 1897)	*Chrysonotomyia purpurea*	Trinidad	Bouček (1977)
		Closterocerus trifasciatus Westwood, 1833	*Closterocerus trifasciatus*	USA	Oatman (1959)
		Closterocerus utahensis Crawford, 1912	*Closterocerus utahensis*	USA	Johnson *et al.* (1980a), Gates *et al.* (2002)
		Neochrysocharis agromyzae (Crawford, 1913)	*Achrysocharella agromyzae*	USA	Harding (1965), Stegmaier (1972)
		Neochrysocharis arizonensis (Crawford, 1913)	*Neochrysocharis arizonensis*	USA	Gates *et al.* (2002)
		Neochrysocharis beasleyi Fisher & La Salle, 1985	*Neochrysocharis beasleyi*	Vietnam	Tran (2009)
		Neochrysocharis diastatae (Howard, 1881)	*Achrysocharella diastatae*	USA	Stegmaier (1972)
			Chrysonotomyia punctiventris	China	Xu *et al.* (1999)
			Chrysonotomyia punctiventris	USA	Johnson *et al.* (1980a), Johnson (1987)
		Neochrysocharis formosa (Westwood, 1833)	*Achrysocharella fullowayi*	USA	Harding (1965)
			Achrysocharella variipes	USA	Harding (1965), Stegmaier (1972)
			Chrysonotomyia formosus	China	Xu *et al.* (1999)
			Chrysonotomyia formosus	USA	Lema & Poe (1979), Johnson (1987)
			Closterocerus formosus	Iran	Asadi *et al.* (2006), Shareki *et al.* (2016)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Bangladesh	Mazumdar & Bhuiya (2016)
			Neochrysocharis formosa	Japan	Amano *et al.* (2008)
			Chrysonotomyia formosa	USA	Tryon & Poe (1981)
			Neochrysocharis formosa	Vietnam	Tran (2009)
		Neochrysocharis okazakii Kamijo, 1978	*Neochrysocharis okazakii*	Taiwan	Chien & Chang (2011)
			Neochrysocharis okazakii	Vietnam	Tran (2009)
		Proacrias thysanoides (De Santis, 1972)	*Proacrias thysanoides*	Peru	Mujica & Kroschel (2011)
Eulophidae	Eulophinae	*Cirrospilus brevicorpus* Shafee & Rizvi, 1988	*Cirrospilus brevicorpus*	India	Shafee & Rizvi (1988)
			Cirrospilus brevicorpus	South Africa	Shafee & Rizvi (1988)
			Cirrospilus brevicorpus	Taiwan	Shafee & Rizvi (1988)
			Cirrospilus ambiguus	Australia	John La Salle (pers. comm.)
			Cirrospilus ambiguus	Vietnam	Tran (2009)
		Cirrospilus vittatus Walker, 1838	*Cirrospilus vittatus*	Iran	Asadi *et al.* (2006)
		Diaulinopsis arenaria (Erdös, 1951)	*Diaulinopsis arenaria*	China	Shi *et al.* (2000)
		Diaulinopsis callichroma Crawford, 1912	*Diaulinopsis callichroma*	Peru	Mujica & Kroschel (2011)
			Diaulinopsis callichroma	USA	Oatman (1959), Stegmaier (1972), Gates *et al.* (2002)
		Diglyphus albiscapus Erdös, 1951	*Diglyphus albiscapus*	Japan	Amano *et al.* (2008)
		Diglyphus begini (Ashmead, 1904)	*Diglyphus begini*	Canada	McClanahan (1975, 1977)
			Diglyphus begini	USA	Trumble & Nakakihara (1983), Heinz & Parrella (1990)
		Diglyphus chabrias (Walker, 1838)	*Diglyphus chabrias*	Turkey	Yefremova *et al.* (2011)
		Diglyphus crassinervis Erdös, 1958	*Diglyphus crassinervis*	Iran	Talebi *et al.* (2011)
			Diglyphus crassinervis	Turkey	Yefremova *et al.* (2011)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	USA	Johnson *et al.* (1980a), Tryon & Poe (1981), Trumble & Nakakihara (1983)
		Diglyphus isaea (Walker, 1838)	*Diglyphus isaea*	China	Xu *et al.* (1999)
			Diglyphus isaea	Iran	Asadi *et al.* (2006), Fathipour *et al.* (2006)
			Diglyphus isaea	Japan	Amano *et al.* (2008)
			Diglyphus isaea	Turkey	Yefremova *et al.* (2011)
			Diglyphus isaea	Vietnam	Tran *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Japan	Amano *et al.* (2008)
		Diglyphus pulchipes (Crawford, 1912)	*Diglyphus pulchipes*	Canada	McClanahan (1977), Stegmaier (1972)
		Diglyphus pusztensis (Erdös & Novicky, 1951)	*Diglyphus pusztensis*	Japan	Amano *et al.* (2008)
		Diglyphus sensilis Yefremova, 2011	*Diglyphus sensilis*	Turkey	Yefremova *et al.* (2011)
			Diglyphus sensilis	Turkey	Yefremova (2015)
		Diglyphus wani Liu, Zhu & Yefremova, 2018	*Diglyphus wani*	China	Ye *et al.* (2018)
		Diglyphus websteri (Crawford, 1912)	*Diglyphus websteri*	Argentina	Valladares *et al.* (1999)
			Diglyphus websteri	Peru	Mujica & Kroschel (2011)
			Diglyphus websteri	USA	Harding (1965)
		Hemiptarsenus varicornis (Girault, 1913)	*Hemiptarsenus semialbiclavus*	Guam	Johnson (1987)
			Hemiptarsenus semialbiclavus	Hawaii	Johnson (1987)
			Hemiptarsenus varicornis	Australia	John La Salle (pers. comm.)
			Hemiptarsenus varicornis	China	Xu *et al.* (1999)
			Hemiptarsenus varicornis	Indonesia	Rauf *et al.* (2000)
			Hemiptarsenus varicornis	Japan	Amano *et al.* (2008)
			Hemiptarsenus varicornis	Sri Lanka	Nagalingam *et al.* (2007)
			Hemiptarsenus varicornis	Taiwan	Chien & Chang (2007)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Vietnam	Tran (2009)
		Hemiptarsenus waileselIae Nowicki, 1929	*Hemiptarsenus waileselIae*	Iran	Talebi *et al.* (2011)
		Hemiptarsenus zilahisebessi Erdös, 1951	*Hemiptarsenus zilahisebessi*	Iran	Talebi *et al.* (2011), Shareki *et al.* (2016)
		Meruacesa liriomyzae (Bouček, 1988)	*Meruana liriomyzae*	Mauritius	Bouček (1988)
			Meruana liriomyzae	Réunion	Bouček (1988)
		Pnigalio minio (Walker, 1847)	*Pnigalio flavipes*	Canada	McClanahan (1977)
		Zagrammosoma americanum Girault, 1916	*Zagrammosoma americanum*	Canada	McClanahan (1977)
			Zagrammosoma americanum	USA	Chandler (1982)
		Zagrammosoma latilineatum Ubaidillah, 2000	*Zagrammosoma latilineatum*	Australia	John La Salle (pers. comm.)
		Zagrammosoma mirum Girault, 1916	*Zagrammosoma mirum*	USA	Oatman (1959)
		Zagrammosoma multilineatum (Ashmead, 1888)	*Zagrammosoma multilineatum*	USA	Stegmaier (1972)
		Zagrammosoma variegatum (Nasi, 1907)	*Zagrammosoma variegatum*	Iran	Hesami *et al.* (2006)
Braconidae	Opiinae	*Desmiostoma parvulum* (Wesmael 1835)	*Desmiostoma parvulum*	USA	Marsh (1979
		Oenongastra microrhopalae (Ashmead, 1896)	*Oenongastra microrhopalae*	Canada	McClanahan (1977)
		Opius (Allophlebus) aridis Gahan, 1913	*Opius aridis*	USA	Harding (1965)
		Opius (Opiognathus) liriomyzae Fischer, 1964	*Opius liriomyzae*	USA	Marsh (1979)
		Opius (Opiothorax) bruneipes Gahan, 1913	*Opius bruneipes*	USA	Harding (1965)
		Opius (Rhogadopsis) dimidiatus (Ashmead, 1889)	*Opius dimidiatus*	Canada	McClanahan (1975, 1977)
			Opius dimidiatus	China	Xu *et al.* (1999)
			Opius dimidiatus	USA	Harding (1965); Lema & Poe (1979)
		Opius caricivorae Fischer, 1964	*Opius caricivorae*	China	Chen *et al.* (2003)
			Opius caricivorae	Taiwan	Chien & Chang (2012)
		Opius chromatomyiae Belokobylskij & Wharton, 2004	*Opius chromatomyiae*	Indonesia	Wayhuni *et al.* (2017)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Braconidae	Opiinae	*Opius chromatomyiae* Belokobylskij & Wharton, 2004	*Opius chromatomyiae*	Vietnam	Tran (2009)
		Opius dissitus Muesebeck, 1963	*Opius dissitus*	China	Xu *et al.* (1999)
			Opius dissitus	Guatemala	Rodríguez-Castañeda *et al.* (2017)
		Opius dissitus Muesebeck, 1963	*Opius dissitus*	USA	Stegmaier (1972), Johnson (1987)
		Phaedrotoma biroi (Fischer, 1960)	*Phaedrotoma biroi*	China	Xing *et al.* (2017)
		Phaedrotoma scabriventris (Nixon, 1955)	*Opius (Gastrosema) scabriventris*	Brazil	Costa-Lima *et al.* (2014)
		Phaedrotoma suturalis (Gahan 1913)	*Opius suturalis*	USA	Harding (1965)
Pteromalidae	Miscogasterinae	*Halticoptera aenea* (Walker, 1833)	*Halticoptera aenea*	USA	Palumbo *et al.* (1994)
		Halticoptera arduine (Walker, 1833)	*Halticoptera arduine*	Peru	Mujica & Kroschel (2011)
		Halticoptera circulus (Walker, 1833)	*Halticoptera circulus*	Guatemala	Rodríguez-Castañeda *et al.* (2017)
			Halticoptera circulus	USA	Stegmaier (1972), Oatman & Johnson (1981)
Pteromalidae	Miscogasterinae	*Halticoptera patellana* (Dalman, 1918)	*Halticoptera patellana*	USA	Jensen & Koehler (1970), Stegmaier (1972)
			Halticoptera patellana	Canada	McClanahan (1977)
Pteromalidae	Pteromalinae	*Cyrtogaster vulgaris* Walker 1833	*Cyrtogaster vulgaris*	Iran	Lotfalizadeh & Gharali (2008)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidium hunteri*	Hawaii	Johnson (1987)
		Ganaspidium pusillae Weld, 1955	*Ganaspidium pusillae*	USA	Harding (1965)
		Gronotoma adachiae Beardsley, 1988	*Gronotoma adachiae*	Vietnam	Abe & Konishi (2012)
		Sinatra pacifica (Yoshimoto, 1962)	*Cothonaspis pacifica*	Hawaii	Johnson (1987), Buffington (2011)

Appendix 2. Parasitoid species reared from Agromyzidae in Queensland (Kleinschmidt 1970)

Agromyzid Species	Parasitoid Family	Parasitoid Species
Sub-family Agromyzinae		
Ophiomyia dianellae	Braconidae	*Opius* sp.
	Pteromalidae	Gen. and sp. indet
Ophiomyia indigoferae	Pteromalidae	Gen. and sp. indet
Ophiomyia lantanae	Braconidae	*Opius (Adontopius) atricornis*
		Ipobracon sp.
	Pteromalidae	*Syntomopus* sp.
Ophiomyia phaseoli	Braconidae	*Phaedrotoma oleracei*
	Eulophidae	*Hemiptarsenus varicornis*
		Chrysonotomyia douglassi
	Pteromalidae	*Callitula viridicoxa*
		Oxyharma subaenea
		Sphegigaster agromyzae
		Syntomopus shakespearei
	Eupelmidae	*Eupelmus grayi*
		Eupelmus sp.
	Eurytomidae	*Eurytoma larvicola*
		Eurytoma sp.
Ophiomyia pisi	Braconidae	*Opius (Gerius) cinerariae*
	Eulophidae	*Hemiptarsenus varicornis*
		Tetrastichus sp.
	Pteromalidae	Gen. and sp. indet
Ophiomyia wikstroemiae	Braconidae	*Opius (Adontopius) atricornis*
	Eulophidae	*Chrysonotomyia* sp.
	Pteromalidae	Gen. and sp. indet
	Eupelmidae	*Eupelmus* sp.

Agromyzid Species	Parasitoid Family	Parasitoid Species
Tropicomyia polyphyta	Braconidae	*Opius (Adontopius) atricornis*
	Eulophidae	*Achrysocharoides* sp.
		Chrysonotomyia sp.
Melanagromyza albisquama	no parasitoids	
Sub-family Phytomyzinae		
Calcomyza humeralis	Braconidae	*Opius (Adontopius) atricornis*
		Opius (Gerius) cinerariae
		Phaedrotoma oleracei
	Eulophidae	*Chrysonotomyia* sp.
		Hemiptarsenus varicornis
	Pteromalidae	*Sphegigaster* sp.
	Eupelmidae	*Eupelmus* sp.
Chromatomyia syngenesiae	Braconidae	*Phaedrotoma oleracei*
Chromatomyia syngenesiae	Braconidae	**Apanteles ruficrus*
	Eulophidae	*Hemiptarsenus varicornis*
		Chrysocharis sp.
		Chrysonotomyia sp.
	Pteromalidae	*Sphegigaster* sp.
		Gen. and sp. indet. A
		Gen. and sp. indet. B
Liriomyza brassicae	Braconidae	*Opius (Gerius) cinerariae*
	Eulophidae	*Hemiptarsenus varicornis*
		Achrysocharoides sp.
		Chrysonotomyia sp.
		Tetrastichus sp.
	Eupelmidae	Gen. nr *Anastalus*
Liriomyza chenopodii	Braconidae	*Opius (Gerius) cinerariae*

Agromyzid Species	Parasitoid Family	Parasitoid Species
	Eulophidae	*Hemiptarsenus varicornis*
		Achrysocharoides sp.
		Chrysonotomyia sp.
***Cerodontha australis*	no parasitoids	
Liriomyza caulophaga	no parasitoids	
Pseudonapomyza spicata	no parasitoids	

*dubious record
** likely to be in fact *Cerodontha milleri* (Spencer 1977)

Appendix 3. Hymenopteran Parasitoids of Agromyzidae collected in Australia by State/Region

Super-Family	Family	Sub-Family	Species	Qld	Torres Strait	SA	ACT/NSW	Vic	Tas	WA	NT	Total
Ichneumonoidea	**Braconidae**	Alysiinae: Dacnusini	*Dacnusa areolaris*					1				**1**
		Opiinae	*Opius (Gerius) cinerariae*	1		1		1				**3**
			Opius (Adontopius) atricornis	1		1						**2**
			Opius sp.	1	1							**2**
			Opius sp. (1)			1						**1**
			Opius sp. (2)			1						**1**
			Opius sp. 1				1					**1**
			Opius sp. 2				1					**1**
			Opius sp. 3				1					**1**
			Opius sp. 4				1					**1**
			Opius sp. 5				1					**1**
			Opius sp. 6				1					**1**
			Phaedrotoma oleracei	1								**1**
Chalcidoidea	**Eulophidae**	Entedoninae	*Apleurotropis* sp.					1				**1**
			Asecodes sp.				1					**1**
			Chrysocharis pubicornis			1	1	1	1			**4**
			Chrysocharis sp.	1								**1**
			Chrysonotomyia douglassi	1								**1**

Super-Family	Family	Sub-Family	Species	Qld	Torres Strait	SA	ACT/NSW	Vic	Tas	WA	NT	Total
Chalcidoidea	**Eulophidae**	Entedoninae	*Chrysonotomyia* sp.	1		1						**2**
			Closterocerus mirabilis				1	1				**2**
			Closterocerus sp.		1	1						**2**
			Neochrysocharis okazakii		1			1				**2**
			Neochrysocharis sp.				1	1				**2**
			Neochrysocharis sp. (*?okazakii*)		1							**1**
			Entedoninae genus A					1				**1**
			Entedoninae genus B					1				**1**
			Entedoninae genus C					1				**1**
Chalcidoidea	**Eulophidae**	Eulophinae										
			Cirrospilus brevicorpus (=*ambiguus*)		1							**1**
			Cirrospilus sp.			1						**1**
			Diglyphus isaea				1	1	1			**4**
			Hemiptarsenus varicornis	1	1	1	1	1	1	1		**7**
			Stenomesius sp.					1				**1**
			Zagrammosoma latilineatum		1	1		1				**3**
			Zagrammosoma sp.			1						**1**
Chalcidoidea	**Eulophidae**	Tetrastichinae										
			Aprostocetus sp. (1)			1						**1**
			Aprostocetus sp. (2)			1						**1**
			Aprostocetus sp. (3)			1						**1**

Super-Family	Family	Sub-Family	Species	Qld	Torres Strait	SA	ACT/NSW	Vic	Tas	WA	NT	Total
Chalcidoidea	**Eulophidae**	Tetrastichinae	*Quadrastichus* sp. (*?liriomyzae*)		1							**1**
			Tetrastichus sp.	1								**1**
Chalcidoidea	**Eulophidae**	Unknown		1								**1**
			Gen. and sp. indet D	1								**1**
Chalcidoidea	**Pteromalidae**	Herbertiinae										
			Herbertia setosa	1			1					**2**
			Herbertia sp.		1							**1**
Chalcidoidea	**Pteromalidae**	Miscogasterinae										
			Ammeia pulchella	1		1	1					**3**
			Halticoptera sp. 1	1								**1**
Chalcidoidea	**Pteromalidae**	Pteromalinae										
			Callitula aenea	1								**1**
			Callitula albipes	1								**1**
			Callitula flavipes			1						**1**
			Callitula grotiusi		1							**1**
			Callitula nigriviridis				1					**1**
			Callitula prima			1						**1**
			Callitula punctata	1								**1**
			Callitula silvensis	1								**1**
			Callitula sp.		1	1					1	**3**
			Callitula uliginosa	1								**1**
			Callitula variventris	1								**1**

Super-Family	Family	Sub-Family	Species	Qld	Torres Strait	SA	ACT/NSW	Vic	Tas	WA	NT	Total
Chalcidoidea	**Pteromalidae**	Pteromalinae	*Callitula viridicoxa*	1		1	1	1				**4**
			Merismomorpha acutiventris	1								**1**
			Merismomorpha asilus	1								**1**
			Merismomorpha faunus	1								**1**
			Merismomorpha flavipetiole	1								**1**
			Merismomorpha fulvicoxa	1								**1**
			Merismomorpha nigra	1								**1**
			Merismomorpha petiolata	1								**1**
			Merismomorpha sicarius	1								**1**
			Oxyharma pulchra	1								**1**
			Oxyharma subaenea	1								**1**
			Sphegigaster agromyzae	1								**1**
			Sphegigaster sp.	1								**1**
			Sphegigaster voltairei	1								**1**
			Syntomopus shakespearei	1			1			1		**3**
			Syntomopus sp.	1								**1**
			Trigonogastrella parasitica	1			1	1	1			**6**
			Trigonogastrella rabiosa	1			1			1		**3**
			Trigonogastrella sp.			1		1				**2**
Chalcidoidea	**Pteromalidae**	Unknown	Gen. and sp. indet C	1								**1**
			Pteromalidae species				1					**1**
			Gen. and sp. indet. A	1								**1**

Super-Family	Family	Sub-Family	Species	Qld	Torres Strait	SA	ACT/NSW	Vic	Tas	WA	NT	Total
Chalcidoidea	**Pteromalidae**	Unknown	Gen. and sp. indet. B	1								**1**
Chalcidoidea	**Tetracampidae**	Tetracampinae	*Epiclerus haeckeli*	1								**1**
			Epiclerus aligherini	1								**1**
			Epiclerus australiensis	1								**1**
Chalcidoidea	**Eurytomidae**	Eurytominae	*Eurytoma larvicola*	1								**1**
			Plutarchia bicarinativentris	1								**1**
			Eurytoma sp.	1								**1**
Chalcidoidea	**Ormyridae**		*Ormyrus* sp.								1	**1**
Chalcidoidea	**Eupelmidae**	Eupelminae	*Eupelmus* sp.	1								**1**
			Eupelmus grayi	1								**1**
			Gen. nr *Anastatus*	1								**1**
Proctotrupoidea	**Diapriidae**	**Proctotrupoidea**	Unknown species					1				**1**
			Grand Total	**50**	**11**	**20**	**19**	**17**	**3**	**3**	**2**	**132**

Appendix 4. Lower development threshold (°C) and thermal constant (day-degrees) for *Hemiptarsenus varicornis*, *Diglyphus isaea*, *Neochrysocharis formosus*, *N. okazakii, Opius cinerariae* and *O. caricovarae* on a range of agromyzid hosts

Parasitoid species [Country]	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (day degrees)	Host Plant	Reference
Hemiptarsenus varicornis [Japan]		*Chromatomyia horticola*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Hemiptarsenus varicornis [Australia]		*Liriomyza brassicae*	9.3	192.3	rape (*Brassica napus)*	Lardner (1991)
Hemiptarsenus varicornis [Japan]	♀	*L. bryoniae*	8.4	149.5	kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis [Japan]	♂	*L. bryoniae*	8.2	146.2	kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis [Japan]		*L. sativae*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Hemiptarsenus varicornis [Taiwan]		*L. trifolii*	11.6	156.3	kidney bean (*Phaseolus vulgaris*)	Chien *et al.* (2004)
Hemiptarsenus varicornis [Japan]	♀	*L. trifolii*	11.8	135.1	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Hemiptarsenus varicornis [Japan]	♂	*L. trifolii*	11.7	126.6	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Hemiptarsenus varicornis [Japan]	♀	*L. trifolii*	8.5	150.8	kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis [Japan]	♂	*L. trifolii*	8.9	137.6	kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Diglyphus isaea [Japan]		*C. horticola*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Diglyphus isaea [Great Britain]	♀	*P. syngenesiae*	12.8	154.6	lettuce (*Lactuca sativa*)	Cheah (1987)

Parasitoid species [Country]	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (day degrees)	Host Plant	Reference
Diglyphus isaea [Great Britain]	♂	*P. syngenesiae*	12.9	152.4	lettuce (*Lactuca sativa*)	Cheah (1987)
Diglyphus isaea [Italy]	♀	*L. huidobrensis*	9.3	165.0	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
Diglyphus isaea [Italy]	♂	*L. huidobrensis*	9.3	157.4	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
Diglyphus isaea [Iran]	♀	*L. sativae*	9.4	161.3	cucumber (*Cucumis sativa*)	Haghani *et al.* (2007b)
Diglyphus isaea [Iran]	♂	*L. sativae*	9.2	153.8	cucumber (*Cucumis sativa*)	Haghani *et al.* (2007b)
Diglyphus isaea [Japan]		*L. sativae*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Diglyphus isaea [Italy]	♀	*L. trifolii*	9.2	161.8	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
Diglyphus isaea [Italy]	♂	*L. trifoli*	9.5	151.4	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
Diglyphus isaea (native) {Japan]	♀	*L. trifolii*	8.5	185.2	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Diglyphus isaea (native) [Japan]	♂	*L. trifoli*	8.4	172.4	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Diglyphus isaea (introduced) [Japan]	♀	*L. trifolii*	9.1	175.4	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Diglyphus isaea (introduced) [Japan]	♂	*L. trifolii*	8.8	169.5	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Diglyphus isaea [The Netherlands]	♀	*L. bryoniae*	8.8	175.4	tomato (*Solanum lycopersicum*)	Minkenberg (1989)
Diglyphus isaea [The Netherlands]	♂	*L. bryoniae*	7.9	158.7	tomato (*Solanum lycopersicum*)	Minkenberg (1989)

Parasitoid species [Country]	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (day degrees)	Host Plant	Reference
Diglyphus isaea [The Netherlands]	♀	*L. trifolii*	8.7	166.7	tomato (*Solanum lycopersicum*)	Minkenberg (1989)
Diglyphus isaea [The Netherlands]	♂	*L. trifolii*	9.0	166.7	tomato (*Solanum lycopersicum*)	Minkenberg (1989)
Neochrysocharis formosa [Japan]		*C. horticola*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Neochrysocharis formosa [China]		*L. sativae*	12.1	197.3	kidney bean (*Phaseolus vulgaris*)	Song et al. (2005)
Neochrysocharis formosa [China]		*L. sativae*	14.3	154.2	kidney bean (*Phaseolus vulgaris*)	Wang *et al.* (2009)
Neochrysocharis formosa [Japan]		*L. sativae*	*	*	kidney bean (*Phaseolus vulgaris*)	Matsuda *et al.* (2009)
Neochrysocharis formosa [Taiwan]		*L. trifolii*	13.0	165.0	kidney bean (*Phaseolus vulgaris*)	Chien *et al.* (2005)
Neochrysocharis formosa [Japan]	♀	*L. trifolii*	9.7	227.3	kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006b)
Neochrysocharis okazakii [China]		*L. sativae*	8.1	188.7	bean	Chen & Ye (2002)
Neochrysocharis okazakii [Vietnam]	♀	*L. chinensis*	11.3	172.4	onion (*Allium fistulosum*)	Tran *et al.* (2012)
Neochrysocharis okazakii [Vietnam]	♂	*L. chinensis*	11.5	166.7	onion (*Allium fistulosum*)	Tran *et al.* (2012)
Neochrysocharis okazakii [Vietnam]	♀	*L. trifolii*	11.3	178.6	kidney bean (*Phaseolus vulgaris*)	Tran *et al.* (2012)

Parasitoid species [Country]	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (day degrees)	Host Plant	Reference
Neochrysocharis okazakii [Vietnam]	♂	*L. trifolii*	11.6	166.7	kidney bean (*Phaseolus vulgaris*)	Tran *et al.* (2012)
Neochrysocharis okazakii [Taiwan]		*L. trifolii*	11.4	192.3	kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2009b)
Opius cinerariae [Australia]	♀	*L. brassicae* I instar	9.1	270.3	rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♂	*L. brassicae* I instar	9.1	250.0	rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♀	*L. brassicae* II instar	9.2	263.2	rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♂	*L. brassicae* II instar	9.4	238.1	rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♀	*L. brassicae* III instar	7.9	294.1	rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♂	*L. brassicae* III instar	7.9	270.3	rape (*Brassica napus)*	Lardner (1991)
Opius caricovarae [Taiwan]		*L. huidobrensis*	7.0	285.7	kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2012)
Opius caricovarae [Taiwan]		*L. sativae*	7.6	263.2	kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2012)

* = Matsuda *et al.* (2009) only tested insects at 20°C and 25°C so meaningful thresholds could not be calculated

Appendix 5. Known species of Agromyzidae in Australia with host plant status (Spencer 1963, 1977; Elliott 2011; Lonsdale 2014)

Species in Sub-family Agromyzinae	Ex[1]	H[2]	NH[3]	Species in Sub-family Phytomyzinae	Ex[1]	H[2]	NH[3]
Agromyza illustris Spencer, 1977			NH	*Amauromyza caliginosa* (Spencer, 1963)			NH
Agromyza mellita Spencer, 1977			NH	*Amauromyza queenslandica* Spencer, 1977			NH
Agromyza venusta Spencer, 1977			NH	*Calycomyza humeralis* (Roser, 1840)	Ex	H	
Japanagromyza tephrosiae (de Meijere, 1917)	Or[4]		NH	*Calycomyza lantanae* (Frick, 1956)	Ex	H	
Japanagromyza eucalypti Spencer, 1963		H		*Cerodontha (Butomomyza) poemyzina* (Spencer, 1963)			NH
Japanagromyza fortis Spencer, 1977			NH	*Cerodontha (Cerodontha) augustensis* Spencer, 1977			NH
Japanagromyza howensis Spencer, 1977			NH	*Cerodontha (Cerodontha) australis* Malloch, 1925	Ex[5]		NH
Japanagromyza kalshoveni (de Meijere, 1934)	Or	H		*Cerodontha (Cerodontha) delectabilis* Spencer, 1977			NH
Melanagromyza albisquama (Malloch, 1927)	Or	H		*Cerodontha (Cerodontha) fulva* Spencer, 1977			NH
Melanagromyza anceps Spencer, 1977			NH	*Cerodontha (Cerodontha) milleri* Spencer, 1973			NH
Melanagromyza apii Hering, 1951		H		*Cerodontha (Cerodontha) robusta* Malloch, 1925			NH
Melanagromyza avicenniae Spencer, 1977		H		*Cerodontha (Cerodontha) vittigera* Malloch, 1927			NH
Melanagromyza blanda Spencer, 1977			NH	*Cerodontha (Cerodontha) voluptabilis* Spencer, 1977			NH
Melanagromyza decapitata Spencer, 1977			NH	*Cerodontha (Icteromyza) piliseta* (Becker, 1903)	Or		NH
Melanagromyza declinata Sasakawa, 1963	Or		NH	*Cerodontha (Icteromyza) triplicata* (Spencer, 1963)		H	
Melanagromyza dipodii Spencer, 1977		H		*Chromatomyia syngenesiae* Hardy, 1849	Ex	H	
Melanagromyza epistomella Spencer, 1977			NH	*Liriomyza brassicae* (Riley, 1885)	Ex	H	
Melanagromyza grata Spencer, 1977			NH	*Liriomyza cassiniae* Spencer, 1977		H	
Melanagromyza hastata Spencer, 1977			NH	*Liriomyza caulophaga* (Kleinschmidt, 1961)	Or	H	
Melanagromyza humida Spencer, 1977			NH	*Liriomyza chenopodii* (Watt, 1924)		H	
Melanagromyza metallica (Thomson, 1869)	Or	H		*Liriomyza compositella* Spencer, 1961	Or		NH
Melanagromyza multiformis Spencer, 1977			NH	*Liriomyza electa* Spencer, 1977			NH
Melanagromyza obtusa (Malloch, 1914)	Ex	H		*Liriomyza helichrysi* Spencer, 1963		H	
Melanagromyza placata Spencer, 1977			NH	*Liriomyza impolita* Spencer, 1977			NH
Melanagromyza praesignis Spencer, 1977			NH	*Liriomyza inopinata* Spencer, 1977			NH
Melanagromyza proboscidella Spencer, 1977			NH	*Liriomyza languida* Spencer, 1977			NH
Melanagromyza prodigiosa Spencer, 1977			NH	*Liriomyza lepida* Spencer, 1977			NH
Melanagromyza pseudograta Spencer, 1977		H		*Liriomyza meracula* Spencer, 1977			NH

Species in Sub-family Agromyzinae	Ex[1]	H[2]	NH[3]	Species in Sub-family Phytomyzinae	Ex[1]	H[2]	NH[3]
Melanagromyza pseudoplacata Spencer, 1977			NH	*Liriomyza obscurata* Spencer, 1963		H	
Melanagromyza pubiseta (Hendel, 1923)			NH	*Liriomyza oleariana* Spencer, 1977		H	
Melanagromyza seneciophila Spencer, 1963			NH	L*iriomyza primitiva* Spencer, 1977			NH
Melanagromyza sojae (Zehntner, 1900)	Ex	H		*Liriomyza sativae* Blanchard, 1938	Ex	H	
Melanagromyza specifica Spencer, 1963		H		*Liriomyza scaevolae* Spencer, 1977		H	
Melanagromyza trifilis Spencer, 1977			NH	*Liriomyza tenera* Spencer, 1977			NH
Melanagromyza trispina (Malloch, 1927)			NH	*Phytobia colorata* Spencer, 1977			NH
Melanagromyza verdescens Spencer, 1963			NH	*Phytobia fausta* Spencer, 1977			NH
Ophiomyia alysicarpi (Bezzi, 1928)		H		*Phytobia incerta* Spencer, 1963			NH
Ophiomyia angustilunula Spencer, 1963		H		*Phytobia insulana* Spencer, 1977			NH
Ophiomyia atralis (Spencer, 1961)		H		*Phytobia liepae* Spencer, 1977			NH
Ophiomyia camarae Spencer, 1963	Ex	H		*Phytobia macalpinei* Spencer, 1977			NH
Ophiomyia cassiae (Spencer, 1963)			NH	*Phytobia malabarensis* Spencer, 1977			NH
Ophiomyia centrosematis (de Meijere, 1940)		H		*Phytobia manifesta* Spencer, 1977			NH
Ophiomyia compositicaulis Spencer, 1977		H		*Phytobia optabilis* Spencer, 1977			NH
Ophiomyia conspicua (Spencer, 1961)	Or	H		*Phytobia sasakawai* Spencer, 1989			NH
Ophiomyia cornuta de Meijere, 1910	Or	H		*Phytoliriomyza australensis* Spencer, 1963	Or		NH
Ophiomyia cymbonoti Spencer, 1977			NH	*Phytoliriomyza australina* (Spencer, 1963)			NH
Ophiomyia deceptiva Spencer, 1977			NH	*Phytoliriomyza cognata* Spencer, 1977			NH
Ophiomyia dianellae (Kleinschmidt, 1960)		H		*Phytoliriomyza collessi* Spencer, 1977			NH
Ophiomyia dulcis Spencer, 1977			NH	P*hytoliriomyza flavopleuralis* Spencer, 1977			NH
Ophiomyia fera Spencer, 1977			NH	*Phytoliriomyza lacunosa* Spencer, 1977			NH
Ophiomyia gemina (Spencer, 1977)			NH	*Phytoliriomyza mollis* Spencer, 1977			NH
Ophiomyia indigoferae (Kleinschmidt, 1961)		H		*Phytoliriomyza monstruosa* Spencer, 1977			NH
Ophiomyia kaputarensis Spencer, 1977			NH	*Phytoliriomyza nigricans* Spencer, 1977			NH
Ophiomyia lantanae Froggatt, 1919	Ex	H		*Phytoliriomyza pallidicentralis* (Malloch, 1927)			NH
Ophiomyia lucidella Spencer, 1977			NH	*Phytoliriomyza pittosporocaulis* (Hering, 1962)		H	
Ophiomyia micra Spencer, 1963			NH	*Phytoliriomyza pittosporophylli* (Hering, 1962)		H	
Ophiomyia mussauensis Spencer, 1966	Or		NH	*Phytoliriomyza polita* Spencer, 1977			NH
Ophiomyia nuginiensis (Spencer, 1977)	Or		NH	*Phytoliriomyza praecellens* Spencer, 1977		H	

Species in Sub-family Agromyzinae	Ex[1]	H[2]	NH[3]	Species in Sub-family Phytomyzinae	Ex[1]	H[2]	NH[3]
Ophiomyia otfordensis Spencer, 1977			NH	*Phytoliriomyza queenslandica* Spencer, 1977		H	
Ophiomyia paramonovi (Spencer, 1963)			NH	*Phytoliriomyza rieki* Spencer, 1977			NH
Ophiomyia parvula Spencer, 1977			NH	*Phytoliriomyza striatella* Spencer, 1977			NH
Ophiomyia phaseoli (Tryon, 1895)	Ex	H		*Phytoliriomyza tricolor* (Malloch, 1927)			NH
Ophiomyia pisi (Kleinschmidt, 1960)		H		*Phytoliriomyza variana* Spencer, 1977			NH
Ophiomyia placida (Spencer, 1963)			NH	*Phytomyza anthocercidis* Spencer, 1977		H	
Ophiomyia pullata Spencer, 1977			NH	*Phytomyza clematidicolla* Spencer, 1963		H	
Ophiomyia rotata Spencer, 1965	Or		NH	*Phytomyza placita* Spencer, 1977			NH
Ophiomyia solanicola Spencer, 1963		H		*Phytomyza plantaginis* Goureau, 1851	Ex	H	
Ophiomyia subtilis Spencer, 1977			NH	*Phytomyza pulchella* Spencer, 1977		H	
Ophiomyia vegeta Spencer, 1977			NH	*Phytomyza ranunculicaulis* Spencer, 1977		H	
Ophiomyia wikstroemiae (Kleinschmidt, 1960)		H		*Phytomyza venerabilis* Spencer, 1977			NH
Tropicomyia polyphyta (Kleinschmidt, 1960)		H		*Phytomyza vitalbae* Kaltenbach, 1872	Ex	H	
				Pseudonapomyza memorata Spencer, 1977			NH
				Pseudonapomyza parilis Spencer, 1977			NH
				Pseudonapomyza probata Spencer, 1977			NH
				Pseudonapomyza pudica Spencer, 1977			NH
				Pseudonapomyza rara Spencer, 1977			NH
				Pseudonapomyza salubris Spencer, 1977			NH
				Pseudonapomyza spinosa Spencer, 1973	Ex	H	

[1]Ex – exotic introduction to Australia [2]H – known host

[3]NH – no known host [4]Or - also present elsewhere in the Oriental region to the north

[5]*Cerodontha (Cerodontha) australis* Malloch, 1925 – Spencer (1977) considered this species to be New Zealand in origin and to have been introduced into Australia.

Appendix 6. Queensland distribution records of Agromyzidae, extracted largely from Spencer (1963, 1977) and Kleinschmidt (1970) – updated 2017–12–01

The main collectors of specimens:

D.H. Colless - D.H.C.
I.F.B. Common - I.F.B.C.
G.A. Holloway - G.A.H.
Z. Liepa - Z.L.
D.K. McAlpine - D.K.M.
S.J. Paramonov - S.J.P.
E.F. Riek - E.F.R.

Species	Queensland records (Kleinschmidt 1960, 1970; Spencer 1963; 1977)	NQ	Host
Sub-family Agromyzinae (35 species)			
Agromyza mellita sp.n.	Holotype ♂, Qld. Ingham, Light Trap, 15.iii.1961 (K.L. Harley); paratype ♀, Dawson River, near Duaringa, 8.v.1970 (Z.L.), both in ANIC. This species closely resembles *papuensis* (Sasakawa 1963) from the Southern Highlands, Papua New Guinea. However, the frons and antennae are paler and the epistoma is significantly broader (little more than linear in *papuensis).* The male genitalia of the 2 species seem to be little differentiated, but exact comparison is not possible, owing to crushing and distortion in the slide preparation of the holotype of *papuensis.* It is believed that the host of *papuensis* is *Setaria palmifolia* (Koen.) Stapf on which leaf-mines were not uncommon in the New Guinea Highlands in October/November, 1973 (Spencer, 1977b). The host of *mellita* may well prove also to be ***Setaria****.*	Y	? *Setaria*
Agromyza venusta sp.n.	Holotype ♂, N. Qld. 3 km W. of Kuranda, 7.v.1967; paratypes: I♂, same data; I♂, 4.8 km W. of Kuranda, Mareeba Rd., 3.v.1967 (all D.H.C.), in ANIC. This species generally resembles *mellita,* also from Qld., but is distinctly darker. The male genitalia are of the characteristic form of grass-feeders in the 2 species but with significant differences in detail.	Y	? grasses
Japanagromyza eucalypti	Qld: Claudie River, near Mt Lamond, 1♀, 18.xii.1971 (D.K.M. and G.A.H.; N.S.W. Iluka rain forest, Clarence River, 1♂, 22.ii.1965 (D.K.M.); 16 km S. of Forster on east bank of Wallis Lake, 1♂, 3.iii.1968 (G.A.H.).	Y	Y
Japanagromyza fortis	Holotype ♂, Qld. Earl Hill, N. of Cairns, 8.v.1967; paratypes: 1♂, Gillies Highway, 2 km W. of Little Mulgrave, 18.iv.1967 (both D.H.C.); 1♀, Palm Is., 20.xii.1930–6.i.1931 (I.M. Mackerras), all in ANIC.	Y	N
Japanagromyza kalshoveni	Qld. Claudie River, near Mt Lamond, 1♀, 18.xii.1971 (G.A.H.); N.T. Katherine, Manbulloo Station,1♂, 1♀.vii.1929 (L.M. Mackerras and T.G. Campbell). "Although there seems little reasonable doubt that the specimens seen do represent *kalshoveni*, it is to be hoped that confirmation can in due course be obtained from examination of the genitalia of a male from Java, or alternatively that leaf-mines can be found in Australia on the host, *Antidesma bunius*" (Spencer 1977a).	Y	? *Antidesma bunius* (Herbert River cherry)
Melanagromyza albisquama	Qld: Junction of Goldmine & Davies Creeks, Kuranda-Mareeba Rd., 1♂, 1♀, 3.v.1967 (D.H.C.); 14.4 km. N.E. of Ingham, I♂, 8.vii.1971 (Z.L.); Big Mitchell Creek, Mareeba-Molloy Rd., 3♂, 4.v.1967 (D.H.C.); Tinaroo Falls Dam, I♂, 8.vii.1971 (Z.L.); CSIRO Farm, Woodstock, near Townsville, 2♂, 1♀, 17.vi.1963 (N.E. Grylls). In Mauritius, *M. albisquama* (as *desmodii* Spencer) has been bred from pods of *Desmodium frutescens* Walp. (Spencer 1965). *M. albisquama* is widespread in northern Australia. The synonymy with *leguminum* establishes that the **host or hosts are in the Fabaceae but the actual species remain to be discovered.** The closest relative appears to	Y	? in Fabaceae

Species	Queensland records (Kleinschmidt 1960, 1970; Spencer 1963; 1977)	NQ	Host
	be *pseudograta* sp.n. Distribution of *albisquama* extends throughout the Old World tropics from Micronesia to the Cape Verde Islands.		
Melanagromyza anceps	Holotype ♂, Qld: W. Garnet River, 19 km S. of Proserpine, 18.xii.1961 (D.K.M. and R. Lossin), in AM.	Y	N
Melanagromyza blanda	Holotype ♂, N. Qld: Bramston Beach, near Innisfail, 10.iv.1967 (open savannah); paratypes 10, 1♂, 1♀ same data (D.H.C.), in ANIC.	Y	N
Melanagromyza decapitata	Holotype ♂, N. Qld: N. Maria Creek, near Silkwood, 14.xii.1961 (D.K.M.), in AM.	Y	N
Melanagromyza epistomella	Holotype ♂, Qld: 4.8 km N.W. of Mt Mowbullan, Bunya Mts, 1,020 m, 7–8.i.1970, MV lamp (G.A.H.); paratype ♀, same data, both in AM.	N	N
Melanagromyza grata	Holotype ♂, N. Qld: Mt Garnet Rd., 20.93 km W. of Ravenshoe, 2.v.1967; paratypes: 1♀, same data (D.H.C.); Emu Park, N.E. of Rockhampton, 1♂, 7.v.1970 (Z.L.); N.T: 54–74 km NW. of Dorisvale HS, 14°13'S, 13°55 'E, 1♂, 14.viii.1968 (M. Mendum), all in ANIC.	Y	N
Melanagromyza humida	Holotype ♂, N. Qld: Big Mitchell Creek, Mareeba·Molloy Rd., 4.v.1967 (D.H.C.); paratype ♀ same data, both in ANIC.	Y	N
Melanagromyza metallica	Widespread in Queensland, extending south to NSW, but less common than was previously believed. Qld: 7–14 km W. of Herberton, via Watsonville, I♂, 1.v.1967 (D.H.C.); Bramston Beach, near Innisfail, I♂, 30.iv.1967 (open savannah) (D.H.C.); Iron Range, 3 ♂, 6♀, 16.viii.1971 (R. Jenkins); Kuranda, I♂, 20.viii.1951 (A.H. Wetherly); Woombye, near Nambour, 5 ♂, 3♀, 11–16.x.1965 (D.H.C.). Spencer (1977b) observed that *metallica* is by far the most common species in Papua New Guinea, with *Bidens pilosa* as the main host. It is widely distributed in the 'Old World' tropics from Micronesia to Africa.		*Ageratum conyzoides, Bidens pilosa* (Asteraceae)
Melanagromyza multiformis	Holotype ♂, N. Qld: Claudie River, near Mt Lamond, 29.v.1966 (D.K.M.), in AM.	Y	N
Melanagromyza obtusa	Known hosts are *Cajanus indicus* Spreng. (India, Malaysia), *Flemingia* sp. (Java) and *Phaseolus radiatus* L. = *Vigna radiata* (Sri Lanka). The larva feeds internally in young seed pods and the reddish puparium (Fig. 39) remains free within the pod. In India *obtusa* is considered a serious pest on *Cajanus.* Its economic importance and biology are discussed in some detail by Spencer (1973). Qld: Speewah Rd., 8 km S. of Kuranda, 1♂, 12.i.1967 (DK.M. and G.A.H.); Claudie River, near Mt Lamond, 2♀, 13.xii.1971 (D.K.M., G.A.H. and D.P. Sands).	Y	? *Cajanus, Vigna radiata* (mung bean)
Melanagromyza pseudograta	Holotype ♂, N. Qld: Mt Garnet Rd., 20.93 km W. of Ravenshoe, 2.v.1967; paratypes: 5♀, same data (D.H.C.). Qld: 14.49 km N. of Dayboro, I♂, 23.v.1966 (Z.L.); near Rosewood (pine forest), I♂, 29.xi1.1961 (R. Lindsay); 40.25 km NW. of Bowen, I♂, 1.x.1950 (ER). (Spencer 1977a tentatively identified the host of this species as *Glycine clandestina*).	Y	N
Melanagromyza pseudoplacata	Holotype ♂, Qld: Claudie River., 1.6 km W. of Mt Lamond, 17.xii.1971 (D.K.M., G.A.H. and D.P. Sands) in AM.	Y	N
Melanagromyza sojae	Qld: Mt Garnet Rd., 20.93 km W. of Ravenshoe, 4♂, 5♀, 2.v.1967 (D.H.C.); Junction of Goldmine and Davies Creeks, Kuranda-Mareeba Rd., 2♂, 3.v.1967 (D.H.C.); Oonoonba, Townsville, 1♂, 5.viii.1968 (P. Ferrar). Hosts include many genera in the Fabaceae, particularly cultivated *Cajanus, Glycine, Medicago* and *Phaseolus.* Damage caused by larval feeding is normally slight, as young plants are rarely attacked, in contrast to	Y	Y

Species	Queensland records (Kleinschmidt 1960, 1970; Spencer 1963; 1977)	NQ	Host
	Ophiomyia phaseoli. Oviposition takes place in leaves but the larva feeds primarily in the stem where pupation takes place.		
Melanagromyza specifica	Qld. Holotype ♂, Queensland: near Brisbane, swept on damp vegetation by gravel pit below Mt. Coot-tha, 22.i.1961 (K.A.S.), in ANIC.	N	N
Ophiomyia pisi (Kleinschmidt, 1960), comb. nov.	Holotype ♀ from Toowong, Qld. October 1957 in QM. Also Toowong, April 1953, November 1957. Ormiston Sept. 1953 (A. R. Brimblecombe).	N	Y
Ophiomyia atralis	N. Qld. Junction of Goldmine and Davies Creeks, Kuranda-Mareeba Rd., 1♂, 3.v.1967; 7–14 km W. of Herberton, via Watsonville, 1♂, 1.v.1967 (both D.H.C.).	Y	N
Ophiomyia centrosematis	Qld: near Noccundra, 4♂, 2♀, 13.xi.1949 (S.J.P.).		
Ophiomyia conspicua	Qld: Crediton Creek, near Eungella, 1♂, 12.xii.1961 (D.K.M.); 11–23 km W. of Herberton, via Watsonvi11e, 1♀, l.v.1967 (D.H.C.).	Y	N
Ophiomyia deceptiva	Holotype ♂, Qld. Mt Walsh National Park, Biggenden, Aug. 1972 paratype ♂, same data (both H. Frauca), in ANIC.	N	N
Ophiomyia dianellae	Holotype ♀ from Coolangatta, Qld. in QM (Apr. 1956); also Jan. 1959, June 1960); Deception Bay (Mar. 1957); Kenmore (Mar. 1957) (A.R. Brimblecombe)	N	Y
Ophiomyia indigoferae	Holotype ♂ from Eight Mile Plains, Brisbane, Qld. in QM (May 1955 R.C. Colbran)	N	Y *Indigofera suffruticosa*
Ophiomyia lantanae	Seed-feeding fly (*Ophiomyia lantanae*) attacking *Lantana camara*. Found from Cape Tribulation to Eden in New South Wales and also around Darwin and Perth. Introduced into Queensland in 1914 and 1917 (Spencer 1973).	Y	Y
Ophiomyia camarae	Herringbone leafminer – released against *Lantana* in last few years. Doing well at Cooktown.	Y	Y
Ophiomyia mussauensis	N. Qld: Mt Garnet Rd., 21 km W. of Ravenshoe, 1♂, 2.v.1967 (D.H.C.).	Y	N
Ophiomyia nuginiensis	N. Qld: Rocky Creek, 11 km N. of Atherton, 1♂, 3.v.1967; Tinaroo Falls Dam,1♂, 27.iv.1967 (both D.H.C.).	Y	N
Ophiomyia parvula	Holotype ♂, Qld: near Rosewood (Brigalow scrub), 29.xii.1961 (R. Lindsay), in ANIC.	N	N
Ophiomyia phaseoli	First recorded in Queensland, from Bulimba, Brisbane, in 1888 by Tryon (1895). Recorded hosts in Queensland are confined to the leguminous genera *Phaseolus* (for which a definite preference is shown), *Dolichos*, *Vigna*, *Glycine* and *Crotalaria*. With the exception of *Crotalaria* (tribe Crotalarieae), these genera are all included in one tribe (Phaseoleae), showing that the fly has a limited choice of hosts. These records show that the species occurs in all districts from the southern border north to Cairns and west to the dry inland districts of Dalby and Goondiwindi. It is uncommon in the Granite Belt, which supplies the State with large quantities of summer beans, since the climate there is unsuitable for overwintering.	Y	Y
Ophiomyia placida (Spencer, 1963), comb. nov.	Holotype ♂ from Lake Placid, Old. in AM. Qld. Claudie River, near Mt Lamond, I♂, 31.v.66 (D.K.M.).	Y	N
Ophiomyia rotata	N. Qld. Big Mitchell Creek, Mareeba-Molloy Rd., 1♂, 4.v.67 (D.H.C.).	Y	N
Ophiomyia wikstroemiae	Holotype ♂ from Kenmore, Qld. in OM.	N	Y

Species	Queensland records (Kleinschmidt 1960, 1970; Spencer 1963; 1977)	NQ	Host
			Wikstroemia indici
Tropicomyia polyphyta	Holotype ♀ from Kenmore, Qld in QM. Spencer (1977) noted: "*The only areas where this species is known are around Sydney and southwards to Waterfall and Clyde Mt, around Brisbane and at Darwin. This, however, in no way reflects its true distribution but mainly indicates where collecting has been undertaken- around Brisbane by Kleinschmidt and at the other localities by myself.*" Two specimens caught on *Erythrina* sp. near Brisbane also almost certainly represent *T. polyphyta* rather than *Ophiomyia cassiae* (Kleinschmidt 1970). It is a highly polyphagous species with extensive records in Australia [56 plant species from 35 families from 24 Orders] (Kleinschmidt (1960, 1970); Spencer 1977, 1990; Maunsell et al. 2017). Kleinschmidt 1970 noted that parasitism was very extensive although not many identified records of parasitoids are known.	Y	Y
Sub-family Phytomyzinae (26 species)			
Amauromyza queenslandica	Holotype ♂, N. Qld: The Crater, near Herberton, 16.xii.61 (McAlpine & Lossin), in AM.	Y	
Calycomyza humeralis	Qld. Junction of Goldmine and Davies Creeks, Kuranda-Mareeba Rd., 1♂, 3.v.67; Woombyee, near Nambour, 1♂, 11–16.x.65; 32.2 km Glen lnnes to Grafton Highway, 1♀, 2.iii.70 (all D.H.C.).	Y	Y
Calycomyza lantanae	Qld. First released in Australia as a biological control agent for *Lantana camara* in 1974. It is more common in tropical Queensland (Cairns to Brisbane) because it prefers warm, moist areas.	Y	Y
Cerodontha (Icteromyza) piliseta	N. Qld: The Boulders, Babinda, I♂, 1.v.67 (D.H.C.); The Boulders, 6.4 km N.W. of Babinda, I♂, 8.vii.71 (Z.L.); 11–22 km W. of Herberton via Watsonville, 1♀, l.v.67; Mt Garnet Rd., 20.9 km W. of Ravenshoe, 1♀, 2.v.67 (both D.H.C.).	Y	? grasses
Cerodontha (Icteromyza) triplicata	Qld. Clayton Gully, 4 km E. of Cunningham's Gap, 1♀, l.vi.66 (Z.L.); 8–12.8 km Mt Lewis Rd. off Mossman-Mt Molloy Rd., 1♂, 1♀, 22.iv.67; North Maria Creek, near Southwood, I♀, 14.xii.61 (D.K.M.); Woombye, near Nambour, 1♂, 11–16.x.65 (D.H.C.).	N	*Juncus pallidus*
Cerodontha (Cerodontha) robusta	Qld. 11 km S.E. of Mt Nebo, c. 457 m, 1♀, 24.v.66 (Z.L.).	N	? grasses
Cerodontha (Butomomyza) poemyzina	Qld: Claudie River, near Mt Lamond, 1♀, 18.xii.71 (G.A.H.); Woombye, near Nambour, 1♀, 11–16.x.65 (D.H.C.).	Y	? grasses
Liriomyza brassicae	Qld. Woombye, near Nambour, 1♂, 2♀, 11–16.x.65 (D.RC) (Spencer 1977). Brisbane, (Spencer 1963).	N	Y
Liriomyza chenopodii	Qld. Woombye, near Nambour, I♂, 11–16.x.65 (D.H.C.).	N	Y
Liriomyza caulophaga	Qld: Near Rosewood (Brigalow scrub), 2♀, 29.xii.61 (R. Lindsey) (Spencer 1977). Qld: Nudgee and near Nocundra (Spencer 1963)	N	Y
Liriomyza sativae	Qld: Seisia 2015	Y	Y
Phytoliriomyza australensis	Qld, Atherton Tableland, head of Clohesy River, 20.v.1956 (Spencer 1963)	Y	
Phytoliriomyza australina	Qld. Coomera River, Lamington National Park, 365 m, 1♀, 28.v.66 (Z.L, Upper Mulgrave River, 16.1 km Goldsborough Rd., 1♀, 9.v.67 (D.H.C.).	Y	N

Species	Queensland records (Kleinschmidt 1960, 1970; Spencer 1963; 1977)	NQ	Host
Phytoliriomyza cognata	Holotype ♂, Qld. Crediton Creek, near Eungella, 14.xii.61 (D.K.M.), in AM.		
Phytoliriomyza flavopleuralis	Holotype ♀, N. Qld. Big Mitchell Creek, Mareeba-Mt Molloy Rd., 4.v.67 (D.H.C.), in ANIC.	Y	
Phytoliriomyza queenslandica	Holotype ♂, Qld. Lamington National Park, 914 m, 5.xi.61; paratypes: 1♂,1♀, same data (I.F.B.C. and M.S. Upton), in ANIC. Recorded on *Pittosporum multiflorum* and *P. oreillyanum* (Maunsell et al. 2015, 2016). Two parasitoids of *T. polyphyta* recorded by Maunsell *et al.* (2015) were a pteromalid (*Herbertia* sp.) and a braconid (Opiinae Braconidae sp. B.)	Y	Y
Phytoliriomyza striatella	N. Qld. 2♂, 2♀, junction of Goldmine and Davies Creeks, Kuranda - Mareeba Rd., 3.v.67; 2♀, 11.2–22.5 km W. of Herberton, via Watsonville, 1.v.67 (all D.H.C.)	Y	N
Phytobia colorata	Holotype ♂, N. Qld. Claudie River, near Mt Lamond, 2.vi.66 (D.K.M.), in AM.	Y	N
Phytobia incerta	N. Qld: Kuranda Range State Forest, 11.2–12.8 km Black Mt Rd., 1♂, 20.iv.67 (D.H.C.); Claudie River, near Mt Lamond, 2♀, 3.v. and 4.vi.66 (D.K.M.). ♀ N. Queensland, Kuranda, 20.xii.1958 (D.K.M.), AM (Spencer 1963a) (as *Agromyza testacea* sp.n.)	Y	N
Phytomyza clematidicolla	Qld. Big Mitchell Creek, Mareeba-Molloy Rd., 1♂, 4.v.67 (D.H.C.).	Y	Y *Clematis* spp.
Phytomyza plantaginis	Qld. Toowong ex *Plantago lanceolata* 31.viii.2011 (PMR)	N	Y
Phytomyza syngenesiae	Qld. Brisbane, 2♂, 2♀, 21.x.48, ex *Sonchus megalocarpus* Hook f. (E.F.R.); Catin, 'mining milk thistle', 3♂, 3♀, 22.x.48; Toowomba, ex same host, 2♂, 2♀, 21.x.48 (all E.F.R.). Kleinschmidt (1970) notes that it has only been found as far north as the Tropic of Capricorn (i.e. close to Bowen).	N	Y
Phytomyza venerabilis	♂, Qld. Yeppoon, N.E. of Rockhampton, 7.v.70 (Z.L.)		
Pseudonapomyza memorata	Holotype ♂, N. Qld. Mt Garnet Rd., 20.9 km W. of Ravenshoe, 2.v.67 (D.H.C.) in ANIC.	Y	N
Pseudonapomyza probata	Holotype ♂, Qld: Gatton, 3.i.62 (R. Lindsay); paratypes: Qld. Bundaberg (mangroves), 1♂, 6.vi.72 (H. Frauca); 11–22 km W. of Herberton, via Watsonville, 1♂, 1♀, l.v.67 (D.H.C.); Mackay, 1♂, 12.v.70 (Z.L.); Big Mitchell Creek, Mareeba-Molloy Rd., 1♀, 4.v.67; junction of Goldmine & Davies Creeks, Kuranda-Mareeba Rd., 4♀, 3.v.67; Mt Garnet Rd., 20.9 km W. of Ravenshoe, 2♂, 1♀, 2.v.67 (all D.H.C.); Emu Park, N.E. of Rockhampton, 1♀, 7.v.70 (Z.L.); Yeppon, N.E. of Rockhampton, 1♀, 7.v.70 (Z.L.); Tinaroo Falls Dam (open savannah), 1♂, 1♀, 27.iv.67 (D.H.C.); Woombye, near Nambour, 1♀, 11–16.x.65 (D.H.C.).	Y	N
Pseudonapomyza rara	Holotype ♂, N. Qld. Tinaroo Falls Dam (open savannah) 27.iv.67; paratype ♀, same data (D.H.C.), in ANIC.	Y	N
Pseudonapomyza spinosa	Qld. Mt Garnet Rd., 20.9 km W. of Ravenshoe, 2♀, 2.v.67 (D.H.C.); Yeppoon, N.E. of Rockhampton, 1♂ (genitalia examined), 3♀, 7.v.70 (Z.L.).	Y	*Brachiaria miliformis, Eleusine indica* (Poaceae)

Appendix 7. Host range for *Tropicomyia polyphyta* in Australia Kleinschmidt 1960, 1965, 1970; Spencer 1963, 1977a; Maunsell *et al.* 2017)

Order	Family	Genus	Species	Common Name
Apiales	Araliaceae	*Cephalaria*	*cephalobotrys*	climbing panax
Apiales	Pennantiaceae	*Pennantia*	*cunninghammii*	brown beech
Berberidopsidales	Berberidopsidaceae	*Berberidopsis*	*beckleri*	montane tape vine
Celastrales	Celastraceae	*Celastrus*	*subspicatus*	large-leaved staff vine
Celastrales	Celastraceae	*Denhamia*	*celastroides*	orange boxwood
Cornales	Cornaceae	*Alangium*	*villosum* subsp. *polysmoides*	muskwood
Cornales	Hydrangeaceae	*Hydrangea*	*macrophylla*	French hydrangea
Dioscoreales	Luzuriagaceae	*Eustrephus*	*latifolius* var. *angustifolius*	wombat berry
Dipsacales	Caprifoliaceae	*Lonicera*	*periclymenum*	European honeysuckle
Ericales	Sapotaceae	*Planchonella*	*australis*	black apple
Ericales	Theaceae	*Camellia*	*japonica*	Japanese camellia
Fabales	Fabaceae	*Erythrina*	*crista-galli*	cockspur coral tree
Fabales	Fabaceae	*Macroptilium*	*lathyroides*	phasey bean
Fabales	Fabaceae	*Senna*	*pendula* var. *glabrata*	Easter Cassia
Fabales	Fabaceae	*Vigna*	*unguiculata* subsp. *sesquipedalis*	asparagus bean
Gentianales	Apocynaceae	*Hoya*	sp.	wax plant
Gentianales	Apocynaceae	*Marsdenia*	*rostrata*	milk vine
Gentianales	Apocynaceae	*Plumeria*	*rubra*	frangipani
Gentianales	Apocynaceae	*Tylophora*	*barbata*	bearded *Tylophora*
Gentianales	Rubiaceae	*Atractocarpus*	*benthamianus*	native gardenia
Gentianales	Rubiaceae	*Coffea*	*arabica*	coffee
Laurales	Atherospermataceae	*Doryphora*	*sassafras*	sassafras
Laurales	Monimiaceae	*Wilkiea*	*huegeliana*	veiny *Wilkiea*
Laurales	Monimiaceae	*Wilkiea*	*austroqueenslandica*	smooth *Wilkiea*

Order	Family	Genus	Species	Common Name
Lecythidales	Lecythidaceae	*Barringtonia*	*acutangula*	freshwater mangrove
Liliales	Ripogonaceae	*Ripogonum*	sp.	supplejack
Liliales	Smilacaceae	*Smilax*	*australis*	lawyer vine
Malpighiales	Berberidopsidaceae	*Streptothamnus*	*moorei*	coast redberry vine
Malpighiales	Euphorbiaceae	*Acalypha*	*wilkesiana*	copper leaf
Malpighiales	Euphorbiaceae	*Croton*	*verreauxii*	green native cascarilla
Malpighiales	Euphorbiaceae	*Euphorbia*	*pulcherrima*	poinsettia
Malpighiales	Phyllanthaceae	*Breynia*	*oblongifolia*	coffee bush
Malpighiales	Salicaceae	*Salix*	*babylonica*	weeping willow
Malvales	Sterculiaceae	*Brachychiton*	sp.	kurrajong
Myrtales	Myrtaceae	*Angophora*	*floribunda*	rough barked apple
Polygales	Malpighiaceae	*Heteropterys*	*nitida*	
Proteales	Combretaceae	*Combretum*	sp.	bush willow
Proteales	Combretaceae	*Quisqualis*	*indica*	Rangoon creeper
Proteales	Proteaceae	*Helicia*	*glabriflora*	pale oak
Ranunculales	Menispermaceae	*Stephania*	*japonica*	snake vine
Rosales	Moraceae	*Maclura*	*cochinchinensis*	cockspur thorn
Rosales	Rhamnaceae	*Alphitonia*	*excelsa*	red ash
Rubiales	Gardenieae	*Gardenia*	*jasminoides*	gardenia
Sapindales	Meliaceae	*Anthocarapa*	*nitidula*	incense cedar
Sapindales	Meliaceae	*Melia*	*azederach*	white cedar
Sapindales	Meliaceae	*Synoum*	*glandulosum* subsp. *glandulosum*	scentless rosewood
Sapindales	Rutaceae	*Citrus*	*aurantifolia*	lime
Sapindales	Rutaceae	*Murraya*	*paniculata*	mock-orange
Scrophulariales	Bignoniaceae	*Kigelia*	*africana*	sausage tree
Solanales	Solanaceae	*Capsicum*	*annuum*	bell pepper

Order	Family	Genus	Species	Common Name
Solanales	Solanaceae	*Cestrum*	*parqui*	green cestrum
Violales	Passifloraceae	*Passiflora*	*caerulea*	blue passionflower
Violales	Passifloraceae	*Passiflora*	*edulis*	passionfruit
Violales	Passifloraceae	*Passiflora*	*foetida*	stinking passion vine
Violales	Passifloraceae	*Passiflora*	*suberosa*	corky passion vine
Violales	Passifloraceae	*Passiflora*	*cinnabarina*	red passionflower

Appendix 4: Parasitoids of the polyphagous agromyzid leafmining flies, *Liriomyza huidobrensis*, *L. trifolii* and *L. sativae* (Diptera: Agromyzidae)

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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Contents

Executive Summary 3
1. Introduction 5
2. Parasitoids reared from *Liriomyza* spp. worldwide 5
3. Composition of the parasitoid assemblages worldwide 6
4. Composition of the parasitoid assemblages in Australia 9
5. Research needs for biological control 10
5. Conclusions 10
6. References 11
7. Appendices 14

Executive Summary

This review complements the review "Prospects for biological control of the vegetable leafminer, *Liriomyza sativae*, in Australia" prepared for the project (MT16004) in 2018. It examines the worldwide records of parasitoid wasps reared from *L. huidobrensis* and *L. trifolii* to see whether the existing assemblage of parasitoid species recorded from agromyzids in Australia would be adequate for control of both species.

Internationally, problems with the polyphagous *Liriomyza* pest species (*L. huidobrensis*, *L. sativae* and *L. trifolii*) are universally associated with the destruction of their parasitoids by excessive use of non-selective insecticides. *Liriomyza huidobrensis* and *L. trifolii* are more challenging to control than *L. sativae* because of their higher levels of resistance to insecticides. In the USA and southern China, *L. trifolii* has displaced, (or is displacing) *L. sativae*. Similarly, *L. huidobrensis* has now become the dominant agromyzid in Kenya at all elevations and is the most widespread pest of potatoes in South Africa. In Europe, *L. huidobrensis* and *L. trifolii* are significant pests of glasshouse production of vegetables and flowers.

Accurate identification of the parasitoids of agromyzid leafminers is difficult, with very few taxonomists available to confirm diagnoses. Consequently, reared parasitoids are sometimes only identified to genus, and some questionable identifications have also been published. Increased use of molecular testing is required to ensure that records can be verified. There is also growing evidence of cryptic species among some species complexes such as *Diglyphus isaea* and *Neochrysocharis formosa*. In these cases, molecular diagnosis is essential.

For this desktop study, we searched the scientific literature for records of parasitoids of *L. huidobrensis* and *L. trifolii*, noting the host plant and country for each record. For *L. trifolii*, we found 610 records while for *L. huidobrensis*, we found 657 records. These data were combined with the data previously gathered for *L. sativae*. Analysis of the combined parasitoid data showed that the three *Liriomyza* spp. have a similar profile of parasitoid families, comprising Eulophidae (63%); Braconidae (19%); Pteromalidae (9%); and Figitidae (8%). Detailed data tables are included in the Appendices, together with four relevant papers (two with translations) which are not readily available to researchers and extension workers.

Around the world, *D. isaea* and *N. formosa* are the most common parasitoids found attacking *L. trifolii* and *L. huidobrensis.* The presence of these two adventive species in Australia is very encouraging. At this stage, there are five adventive species of parasitoids of agromyzids in Australia. With more field surveys, more adventive species will likely be found.

In Australia, the parasitoid assemblage is deficient in the Figitidae and the Miscogastrinae, the subfamily of *Halticoptera* - which is the most common pteromalid genus attacking the polyphagous *Liriomyza* species in North and South America. However, further surveys in Queensland may reveal more species in these two groups as a few specimens of the figitid, *Gronotoma domestica*, and the pteromalid, *Halticoptera* sp., have been recorded in that state.

Establishing the presence of candidate parasitoids is only a first step in implementing a successful biological control program for pest *Liriomyza* spp. in horticultural and nursery crops in Australia. Movement patterns of parasitoids from reservoirs of non-crop plants to crops are unknown, as is the dispersal of agromyzids and their parasitoids through wider cropping areas, adjoining non-crop areas and sequentially planted crops (Ridland *et al.* 2020).

In summary, the prime research needs for biological control from an Australian horticultural industry perspective identified initially in the review for *L. sativae* remain equally as relevant for *L. huidobrensis* and *L. trifolii* and include:

- Delineating the current distribution of *D. isaea* and *N. formosa;*
- Clarifying the taxonomy and biology of the complex of opiine braconid parasitoids already found attacking agromyzids in Australia;

- Clarifying the taxonomy and biology of the eulophid parasitoids found attacking agromyzids in Australia;
- Establishing whether there are climatic biotypes for *Hemiptarsenus varicornis* and *Zagrammosoma latilineatum* – two eulophid species found from southern Australia to Cape York (and Indonesia);
- Determining what other agromyzid species in Australian horticultural regions will act as reservoirs of parasitoids for *L. huidobrensis*, *L. sativae* and *L. trifolii.*

We believe the abundant endemic parasitoid fauna in Australia can help control polyphagous *Liriomyza* spp. should they become established in horticultural regions. The composition and impact of the endemic parasitoid assemblage in Australia on populations of invasive *Liriomyza* spp. needs to be assessed adequately in the field before the importation of additional exotic parasitoid species is contemplated. The primary challenge will be to ensure these parasitoids are not disrupted by excessive use of inappropriate insecticides and reservoirs of parasitoids and non-target agromyzids are identified and managed effectively. This would also apply equally if exotic parasitoids were released.

1. Introduction

This review complements the review "Prospects for biological control of the vegetable leafminer, *Liriomyza sativae*, in Australia" prepared for the project (MT16004) in 2018. It examines the records of parasitoid wasps recorded worldwide from *L. huidobrensis, L. trifolii* and *L. sativae* to see whether the existing assemblage of parasitoid species recorded from agromyzids in Australia would be adequate for control of the three *Liriomyza* species.

Internationally, problems with the polyphagous *Liriomyza* pest species (*L. huidobrensis*, *L. sativae* and *L. trifolii*) are universally associated with the destruction of their parasitoids by excessive use of non-selective insecticides. When using parasitoids to control agromyzid leafminers as part of an IPM program, it is necessary to (i) avoid the use of broad-spectrum insecticides, (ii) develop economic thresholds to reduce numbers of sprays applied, (iii) understand the impact of host plant on parasitoid preferences, and (iv) understand the role of non-crop hosts as reservoirs of parasitoids (ideally from non-pest agromyzid species) (Ridland *et al.* 2020).

Liriomyza huidobrensis and *L. trifolii* are considered more challenging to control than *L. sativae* because of their higher levels of resistance to insecticides (van der Staay 1992; Ferguson 2004; Reitz *et al.* 2013; Weintraub *et al.* 2017). In the USA and southern China, *L. trifolii* has (or is) displacing *L. sativae* (Gao *et al.* 2012, 2017). Similarly, *L. huidobrensis* has now become the dominant agromyzid in Kenya at all elevations (Foba *et al.* 2015) and is the most widespread pest of potatoes in South Africa (Visser & Weintraub 2001a,b; Potatoes South Africa 2017). In Europe, *L. huidobrensis* and *L. trifolii* are significant pests of glasshouse production of vegetables and flowers. *Liriomyza trifolii* was introduced to Kenya with chrysanthemum cuttings from Florida in 1976 (Spencer 1989) and was subsequently recorded in South Africa in 1983 (Bolton 1986). Infested chrysanthemum cuttings from Kenya and Colombia are considered the source for *L. trifolii* arriving in Europe in the early 1980s (Spencer 1989). Again, in Indonesia, chrysanthemum nurseries are believed to be the initial infestation site for *L. trifolii* (Yasa *et al.* 2020).

With the ongoing dispersal of polyphagous *Liriomyza* species (*L. sativae*, *L. trifolii* and *L. huidobrensis*) through the world, studies in many countries show that these invading populations are rapidly exploited by endemic agromyzid parasitoids (Murphy & LaSalle 1999). The few published Australian field studies on the occurrence of agromyzid flies and their parasitoids show there is a substantial community of parasitoid wasps attacking agromyzids in Australia, with species mainly from Eulophidae, Pteromalidae and Braconidae (Ridland *et al.* 2020). The most abundant species from these studies are two cosmopolitan species, *Hemiptarsenus varicornis* and *Diglyphus isaea*, and four Australian indigenous species, *Zagrammosoma latilineatum, Trigonogastrella parasitica*, *Closterocerus mirabilis* and *Opius cinerariae*, for which little is known about their biology and ecology.

2. Parasitoids reared from *Liriomyza* spp. worldwide

Since 1987, several lists of parasitoids reared from polyphagous *Liriomyza* spp. have been published (Table 1). With any compilation of species names from the scientific literature, there will be uncertainty about the validity of some records. As well, sampling intensity can vary dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem (Grenouillet *et al.* 1993; Noyes 1994; Shaw 1994; Ridland *et al.* 2020).

In the scientific literature in the 1950s and 1960s, there were difficulties in identifying *Liriomyza* spp. correctly (Spencer 1965, 1973). Accurate identification of the parasitoids of agromyzid leafminers is also difficult and their taxonomy remains in flux, with very few taxonomists available to confirm diagnoses. It is highly likely that many species are yet to be formally described. Consequently, reared parasitoids are sometimes only identified to genus, and some questionable identifications have been published, which can be perpetuated in subsequent lists of parasitoids. Increased use of molecular testing is required to ensure that records in regions can be verified. As well, it is vital that voucher specimens are also lodged in suitable insect collections where they will be databased and be available for future reference (Ridland *et al.* 2020).

There is also growing direct evidence of cryptic species among species such as *D. isaea* (Sha *et al.* 2007). Indirectly, the regional variability observed in *N. formosa* (Fisher & La Salle 2005; Hernández *et al.* 2009) also suggests that there might be an unresolved species complex in this widespread species. Molecular diagnosis and morphological analysis using samples from many locations will be needed to resolve these taxonomic problems.

Table 1 Comprehensive lists of parasitoids recorded from polyphagous *Liriomyza* spp. worldwide

Reference	Description
Waterhouse & Norris (1987)	List of parasitoids of *L. huidobrensis*, *L. sativae* and *L. trifolii.* They differentiate between parasitoids identified to species or only to genus. They also provide lists of parasitoids reared from *Liriomyza* spp.' or *'Liriomyza*. sp.' (implying *L. sativae*, *L. trifolii* or *L. huidobrensis* or mixed populations of two or three of these). Note that they include records of *L. langei* from California and Hawaii as *L. huidobrensis*.
Grenouillet *et al.* (1993)	A world list of all known parasites and predators of the 26 economically important species of *Liriomyza* is given. Relatively few records of parasitoids of *L. huidobrensis*, since the review was completed before the major expansion of *L. huidobrensis* worldwide. Because of space limitations, the majority of the references consulted were not cited by the authors.
Barranco Vega (2003)	Lists of 73 parasitoid species reared from *L. bryoniae*, *L. strigata*, *L. huidobrensis* and *L. trifolii*. No references given and nomenclature is outdated.
Liu *et al.* (2011)	This review provides a list of 143 parasitoids recorded from *Liriomyza* spp. but without references for individual species. Nomenclature is outdated for many species.
Weintraub *et al.* (2017)	This is a very comprehensive record of parasitoids of *L. huidobrensis*, with host plant records for nearly all records. Primary references are cited.
Noyes (2019)	The Universal Chalcidoidea Database provides an exhaustive catalogue of Chalcidoidea, with extensive references and up-to-date nomenclature. *Liriomyza huidobrensis* (30 species of Eulophidae; 8 species of Pteromalidae), *L. sativae* (39 species of Eulophidae; 4 species of Pteromalidae) and *L. trifolii* (45 species of Eulophidae; 6 species of Pteromalidae; 1 species of Tetracampidae).
Ridland *et al.* (2020)	Seventy-one parasitoid species recorded from *L. sativae* worldwide. Primary references are cited.

For this review, we went back to the primary literature sources but used some of the previous lists (Table 1) as guides. For *L. huidobrensis*, we used the data from Weintraub *et al.* (2017) as the foundation with supplementary data added where found. For *L. trifolii*, all data was retrieved from primary sources. Data for *L. sativae* was based on Ridland *et al.* (2020). Host plant records were included in the database where available for all three species but were not used for the analyses presented here. Host/country records for parasitoids of each *Liriomyza* species. were pooled to avoid inflation of the importance of a particular species if the scientific effort in a country (*e.g.* Japan, Turkey and China) was much greater than in other countries. For example, if 10 records of *D. isaea* were recorded attacking *L. trifolii* in Turkey, these data were pooled to give one record of *D. isaea* in Turkey. Parasitoids reared from mixed collections of *Liriomyza* spp. were not included in the lists.

3. Composition of the parasitoid assemblages worldwide

The composition of the parasitoid assemblages for all three *Liriomyza* species is very similar in terms of family and sub family composition (Table 2). The majority of species (63%) are from the family Eulophidae, with representation equally between the Eulophinae and the Endedoninae subfamiles. The Braconidae is

the next most abundant group (19%), with the family Pteromalidae (9%) and the Eucoilinae subfamily in the Figitidae (8%) also abundant parasitoids in some regions for all three *Liriomyza* species.

Table 2 Hymenopteran parasitoids reared from *L. huidobrensis* (Lh), *L. trifolii* (Lt) and *L. sativae* (Ls) worldwide summarized by family and sub-family. Data for all parasitoid species are listed in Appendix 4.

Family/	Lh		Lt		Ls		∑[1]		Pooled[2]		Overlap[3]	
Sub-Family	n	%	n	%	n	%	n	%	n	%	n	%
Eulophidae	**49**	**66%**	**61**	**66%**	**50**	**69%**	**160**	**67%**	**92**	**63%**	**68**	**43%**
Eulophinae	27	36%	29	32%	26	36%	82	34%	44	30%	38	46%
Endedoninae	21	28%	28	30%	24	33%	73	31%	44	30%	29	40%
Tetrastichinae	1	1%	4	4%	0	0%	5	2%	4	3%	1	20%
Braconidae	**13**	**18%**	**13**	**14%**	**12**	**17%**	**38**	**16%**	**27**	**19%**	**11**	**29%**
Opiinae	10	14%	10	11%	12	17%	32	13%	22	15%	10	31%
Alysiinae	2	3%	3	3%	0	0%	5	2%	4	3%	1	20%
Braconinae	1	1%		0%	0	0%	1	0%	1	1%	0	0%
Pteromalidae	**7**	**9%**	**8**	**9%**	**5**	**7%**	**20**	**8%**	**13**	**9%**	**7**	**35%**
Miscogastrinae	6	8%	4	4%	4	6%	14	6%	8	6%	6	43%
Pteromalinae	1	1%	4	4%	1	1%	6	3%	5	3%	1	17%
Figitidae	**4**	**5%**	**9**	**10%**	**5**	**7%**	**18**	**8%**	**11**	**8%**	**7**	**39%**
Eucoilinae	4	5%	9	10%	5	7%	18	8%	11	8%	7	39%
Tetracampidae	**1**	**1%**	**1**	**1%**	**0**	**0%**	**2**	**1%**	**2**	**1%**	**0**	**0%**
Tetracampinae	0	0%	1	1%	0	0%	1	0%	1	1%	0	0%
Platynocheilinae	1	1%	0	0%	0	0%	1	0%	1	1%	0	0%
∑	**74**		**92**		**72**		**238**		**145**		**93**	

[1]Sum of all named species reared from each *Liriomyza* host species
[2]Pooled species from all three *Liriomyza* spp. (NB it is not the sum of the species from each *Liriomyza* spp. because of overlap of parasitoid species between *Liriomyza* spp.)
[3]Overlapping species (∑-pooled) (those reared from more than one *Liriomyza* host species)

Parasitoids of agromyzids are typically generalists, attacking a wide range of agromyzid species (Murphy & La Salle 1999; Ridland *et al.* 2020). The level of overlap of parasitoid species and the target *Liriomyza* species was examined in Table 3. About 1/3 of the parasitoid species have been recorded from all three *Liriomyza* spp., while a further 13% of species been recorded from two of the target *Liriomyza* spp. (Table 3). Seventy-nine species were only recorded from one of the *Liriomyza* species. This largely reflects the recording of native parasitoids from the area of origin of a particular *Liriomyza* species.

With the international movement of all three *Liriomyza* spp. being a relatively new phenomenon, endemic parasitoid species will often attack the invasive agromyzid species (Murphy & LaSalle 1999), which boosts the level of species overlap. The accidental arrival of parasitoids with their invasive hosts has been recognised as an increasingly common phenomenon due to the increasing volume of global trade (Mason *et al.* 2017; Weber *et al.* 2017). For example, two Asian parasitoids of spotted wing drosophila, *Drosophila suzukii*, (*Leptopilina japonica* and *Ganaspis brasilensis* G1 clade) being investigated for possible release in North America have now been found to be established in British Columbia (Abram *et al.* 2020). As well, *L. japonica* has now been recorded parasitising *D. suzukii* infesting cherries in Italy (Puppato *et al.* 2020).

The family and sub-family breakdown of the parasitoid assemblages (Table 2) is reflected in Table 4, where 4 genera of Eulophidae [*Diglyphus* (Eulophinae), *Chrysocharis* (Endedoninae), *Neochrysocharis* (Endedoninae) and *Hemiptarsenus* (Eulophinae)], one genus of Braconidae [*Opius* (Opiinae)] and one genus

of Pteromalidae [*Halticoptera* (Miscogasterinae)] are the six most commonly recorded genera parasitizing the three *Liriomyza* spp. (accounting for 59% of records).

Table 3 Overlap of parasitoid species with agromyzid host. Data for all parasitoid species are listed in Appendix 4.

	L. huidobrensis		*L. trifolii*		*L. sativae*	
Species overlap	*n*	%	*n*	%	*n*	%
Lh/Lt/Ls	27	36%	27	29%	27	38%
Lh/Lt	11	15%	11	12%		
Lh/Ls	12	16%			12	17%
Lt/Ls			16	17%	16	22%
Lh only	24	32%				
Lt only			38	41%		
Ls only					17	24%
∑	**74**		**92**		**72**	

Lh = *Liriomyza huidobrensis,* Lt = *L. trifolii,* Ls= *L. sativae*

Table 4 Most commonly recorded parasitoid genera by country records in descending order (pooling all records for a particular country for each *Liriomyza* sp.)

Family	**Sub-Family**	**Genus**	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**	***%*[1]**
Eulophidae	Eulophinae	*Diglyphus*	48	35	21	104	18%
Braconidae	Opiinae	*Opius*	25	29	14	68	12%
Eulophidae	Endedoninae	*Chrysocharis*	30	19	18	67	11%
Eulophidae	Endedoninae	*Neochrysocharis*	14	34	15	63	11%
Eulophidae	Eulophinae	*Hemiptarsenus*	14	19	11	44	7%
Pteromalidae	Miscogastrinae	*Halticoptera*	17	12	6	35	6%
Eulophidae	Endedoninae	*Closterocerus*	5	6	5	16	3%
Eulophidae	Eulophinae	*Pnigalio*	6	8	1	15	3%
Eulophidae	Eulophinae	*Zagrammosoma*	4	6	5	15	3%
Eulophidae	Endedoninae	*Asecodes*	3	5	4	12	2%
Figitidae	Eucoilinae	*Gronotoma*	5	4	2	11	2%
Braconidae	Alysiinae	*Dacnusa*	9	2		11	2%
Braconidae	Opiinae	*Phaedrotoma*	6	1	3	10	2%
Eulophidae	Endedoninae	*Chrysonotomyia*	2	7	1	10	2%
Eulophidae	Eulophinae	*Diaulinopsis*	3	4	3	10	2%

[1] % of all country records (588)

The five most commonly recorded parasitoid species parasitizing *L. huidobrensis*, *L. sativae* and *L. trifolii* worldwide (Table 5) are *Diglyphus isaea*, *Neochrysocharis* formosa, *Hemiptarsenus varicornis*, *Chrysocharis pentheus* and *Opius* sp.

Table 5 Most commonly recorded parasitoid species by country records in descending order (pooling all records for a particular country for each *Liriomyza* sp.). (Full data set in Appendix 5).

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*	∑
Eulophidae	Eulophinae	*Diglyphus isaea*	15	12	5	32
Eulophidae	Endedoninae	*Neochrysocharis formosa*	7	16	7	30
Eulophidae	Eulophinae	*Hemiptarsenus varicornis*	6	11	9	26
Braconidae	Opiinae	*Opius* sp.[1]	13	6	1	20
Eulophidae	Endedoninae	*Chrysocharis pentheus*	5	5	5	15
Eulophidae	Eulophinae	*Diglyphus crassinervis*	5	6	2	13
Braconidae	Opiinae	*Opius dissitus*	4	5	3	12
Pteromalidae	Miscogastrinae	*Halticoptera circulus*	5	5	2	12
Eulophidae	Endedoninae	*Neochrysocharis okazakii*	3	5	3	11
Eulophidae	Endedoninae	*Chrysocharis* sp.[1]	6	4		10
Braconidae	Opiinae	*Opius dimidiatus*	2	4	3	9
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi*	3	5	1	9
Eulophidae	Eulophinae	*Diglyphus begini*	4	2	3	9
Eulophidae	Endedoninae	*Chrysocharis caribea*	2	2	4	8
Eulophidae	Endedoninae	*Chrysocharis oscinidis*	1	4	3	8
Eulophidae	Eulophinae	*Diglyphus* sp.[1]	6	2		8
Pteromalidae	Miscogastrinae	*Halticoptera* sp.[1]	7	1		8

[1]records of parasitoids only identified to genus are included in the data for *L. huidobrensis* and *L. trifolii*

In South America, the braconid, *Phaedrotoma scabriventris*, the eulophid, *Chrysocharis flacilla*, and the pteromalid, *Halticoptera arduine* are the most abundant species parasitising *L. huidobrensis* in Peru (Mujica & Kroschel 2011; Mujica 2016) and Argentina (Valladares *et al.* 1999). Note that the genus *Phaedrotoma* was split from *Opius* in 1997 (Li et al. 2013). All three species have recently been imported into Kenya for the biological control of *L. huidobrensis* (Muchemi *et al.* 2018a,b,c,d). To date, *P. scabriventris* appears to have established well (Foba *et al.* 2016). The long-term results of these Kenyan releases will provide insights into the value of importing and releasing exotic parasitoids in a classical biological control program for *L. huidobrensis*.

4. Composition of the parasitoid assemblages in Australia

In Australia, *D. isaea* and *N. formosa* are both recent adventive species which have been recorded parasitizing *Liriomyza* spp. in south-eastern Australia (Ridland *et al.* 2020). *Chrysocharis pubicornis* is a pupal parasitoid of agromyzids and is the only representative of the genus in Australia (Bouček 1988). It is not considered an important parasitoid of *Liriomyza* spp., unlike *C. pentheus*, which is a common larval-pupal endoparasitoid of *Liriomyza* spp. in Asia (Cai & Saito 2011; Chien & Chang 2013), and *C. flacilla* which is a common larval-pupal parasitoid of *L. huidobrensis* in South America (Salvo & Valladares 1997). *Opius*

spp. and *Phaedrotoma* spp. are present in Australia, however, it will be important to clarify the taxonomy and biology of the complex of opiine braconid parasitoids already found attacking agromyzids.

In Australia, the parasitoid assemblage lacks species in the Eucoilinae and the Miscogastrinae, the subfamily of *Halticoptera* - which is the most common pteromalid genus attacking the polyphagous *Liriomyza* species in North and South America. Bouček (1988) listed only two species from the Miscogastrinae in Australia: the cosmopolitan *Ammeia pulchella* and the indigenous *Rhicnocoelia incisa*. Two other Australian species, *Lamprotatus damia* and *Lamprotatus nicon* are considered to be *incertae sedis* (Latin for 'of uncertain placement') given that both type species are missing. Importantly, Bouček (1988) noted that there was at least one species of *Halticoptera* in Australia and a second species in Papua New Guinea. He considered that they were probably introduced species, but noted that no Australo-Papuan species of *Halticoptera* have been named so far. Further collecting in Queensland may reveal more species in these two sub-families as a few specimens of the eucoiline, *Gronotoma domestica*, and the pteromalid, *Halticoptera* sp., have been recorded in that state (Bouček 1988).

5. Research needs for biological control

In summary, the prime research needs for biological control from an Australian horticultural industry perspective identified initially in the review for biological control of *L. sativae* remain equally as crucial for *L. huidobrensis* and *L. trifolii* and include:

- Delineating the current distribution of *D. isaea* and *N. formosa*;
- Clarifying the taxonomy and biology of the complex of opiine braconid parasitoids already found attacking agromyzids in Australia;
- Clarifying the taxonomy and biology of the eulophid parasitoids found attacking agromyzids in Australia;
- Establishing whether there are climatic biotypes for *Hemiptarsenus varicornis* and *Zagrammosoma latilineatum* – two eulophid species found from southern Australia to Cape York (and Indonesia);
- Determining what other agromyzid species in Australian horticultural regions will act as reservoirs of parasitoids for *L. huidobrensis*, *L. sativae* and *L. trifolii*;
- Studying the biology, ecology and distribution of endemic parasitoids in Australia (*Zagrammosoma latilineatum*, *Closterocerus mirabilis* (Family Eulophidae), *Trigonogastrella parasitica* (Family Pteromalidae; Sub-family Pteromalinae), and *Opius cinerariae* (Family Braconidae; Sub-family Opiinae)) to assess their potential role in controlling *L. huidobrensis*, *L. sativae* and *L. trifolii* in open-air production as well as their potential suitability for use in augmentative biological control in protected cropping;
- Undertaking field surveys in Queensland, which may reveal more species in the Eucoilinae and Miscogastrinae - given a few specimens of *Gronotoma domestica* (Family Figitidae; Sub-family Eucoilinae) and *Halticoptera* sp. (Family Pteromalidae; Sub-family Miscogastrinae) have been recorded in that state.

5. Conclusions

With increased surveying in Australia, it is highly likely that the numbers of identified species of both agromyzids and their parasitoids will increase substantially.

Establishing the presence of candidate parasitoids is only a first step in implementing a successful biological control program for polyphagous *Liriomyza* spp. in horticultural and nursery crops in Australia. Movement patterns of parasitoids from reservoirs of non-crop plants to crops are unknown, as is the dispersal of agromyzids and their parasitoids through wider cropping areas, adjoining non-crop areas and sequentially planted crops (Ridland *et al.* 2020).

We believe the abundant endemic parasitoid fauna in Australia can help control polyphagous *Liriomyza* spp. should they become established in horticultural regions. The composition and impact of the endemic parasitoid assemblage in Australia on populations of invasive *Liriomyza* spp. needs to be assessed adequately in the field before the importation of additional exotic parasitoid species is contemplated. The primary challenge will be to ensure these parasitoids are not disrupted by excessive use of inappropriate insecticides and that the reservoirs of parasitoids and non-target agromyzids are identified and managed effectively (Ridland *et al.* 2020). This would also apply equally if exotic parasitoids were released.

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7. Appendices

Appendix 7.1 Parasitoid species reared from *Liriomyza huidobrensis*

Appendix 7.2 Parasitoid species reared from *Liriomyza trifolii*

Appendix 7.3 Parasitoid species reared from *Liriomyza sativae*

Appendix 7.4 Overlap of parasitoid species recorded from the 3 *Liriomyza* spp.

Appendix 7.5 Parasitoid species by country records in descending order (pooling all records for a particular country for each *Liriomyza* sp.)

Appendix 7.6 Grenouillet C, Martinez M & Rasplus JY. 1993. Liste des parasitoïdes et des prédateurs des *Liriomyza* d'importance economique dans le monde (Diptera: Agromyzidae). *Colloque sur les mouches mineuses des plantes cultivees*, 24-26 Mar 1993, Montpellier, France, 143-156. [original and translation]

Appendix 7.7 Bolton M. 1986. Die Amerikaanse blaarmyner: 'n belangrike plaag by tamaties. *Roodeplaat Bulletin* **11**, 8-10. [original and translation]

Appendix 7.8 Visser D & Weintraub P. 2001. Outbreak of the potato leafminer, comparisons between Israel and South Africa. *Chips* **15**(2), 38–39; Potato leafminer - lessons Israel. *Chips* **16**(4), 39–40.

Appendix 7.1 Parasitoid species reared from *Liriomyza huidobrensis*

Specimens reared from mixed collections of *Liriomyza* spp., which included *L. huidobrensis*, were not included in the list. This table is based on data from Weintraub *et al.* (2017) with supplementary data added. As with any compilation of species names from the scientific literature, there will be uncertainty about the validity of some species and sampling intensity varies dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem (Grenouillet *et al.* 1993; Noyes 1994; Shaw 1994). Given the nomenclatural changes that have occurred in time, the currently accepted name is given, together with the name used in the original reference. All references for the Appendix are appended.

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Braconidae	Alysiinae	*Dacnusa sasakawai* Takada, (1977)	*Dacnusa sasakawai*	Japan	Shindo *et al.* (2005)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	Austria	Stolz & Blümel (1998)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	China	Pan *et al.* (2019)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	Italy	Burgio *et al.* (2005)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	Portugal	Godinho & Mexia (2000); Mexia *et al.* (2004)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	Réunion	Vayssieres *et al.* (2001)
Braconidae	Alysiinae	*Dacnusa sibirica* Telenga, 1935	*Dacnusa sibirica*	The Netherlands	van der Linden (1990, 1991)
Braconidae	Alysiinae	*Dacnusa* sp.	*Dacnusa* sp.	Canada	Bahlai *et al.* (2006)
Braconidae	Alysiinae	*Dacnusa* sp.	*Dacnusa* sp.	China	Pan *et al.* (2019)
Braconidae	Alysiinae	*Oenonogastra* sp.	*Oenonogastra* sp.	Costa Rica	Carballo *et al.* (1990); Rodriguez (1997); Weintraub *et al.* (2017)
Braconidae	Braconinae	*Bracon intercessor* Nees, 1834	*Bracon intercessor*	Turkey	Civelek *et al.* (2002)
Braconidae	Opiinae	*Opius caricivorae* Fischer, 1964	*Opius caricivorae*	Taiwan	Chien & Chang (2012)
Braconidae	Opiinae	*Opius chromatomyiae* Belokobylskij & Wharton, (2004)	*Opius chromatomyiae*	Indonesia	Hidrayani *et al.* (2005)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	China	Chen *et al.* (2000)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	Guatemala	Weintraub *et al.* (2017)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	China	Song *et al.* (2003)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Réunion	Olivera & Bordat (1996)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Kenya	Foba *et al.* (2015)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Guatemala	Weintraub *et al.* (2017)
Braconidae	Opiinae	*Opius mandibularis* Gahan, 1915	*Opius mandibularis*	Guatemala	Weintraub *et al.* (2017)
Braconidae	Opiinae	*Opius meracus* Fischer, 1960	*Opius meracus*	Turkey	Civelek *et al.* (2002)
Braconidae	Opiinae	*Opius pallipes* Wesmael, 1835	*Opius pallipes*	Malaysia	Nor Ahya & Nur Liyana (2018)
Braconidae	Opiinae	*Opius pallipes* Wesmael, 1835	*Opius pallipes*	The Netherlands	van der Linden (1990)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Brazil	Pereira *et al.* (2002); Guimarães *et al.* (2009); Dequech *et al.* (2010)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Canada	Bahlai *et al.* (2006)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	China	Chen *et al.* (2000); Luo *et al.* (2001); Chen *et al.* (2011)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Columbia	Hincapie *et al.* (1993)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Costa Rica	Carballo *et al.* (1990); Rodriguez (1997); Weintraub *et al.* (2017)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Indonesia	Prijono *et al.* (2004)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Israel	Weintraub *et al.* (2017)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Japan	Shindo *et al.* (2005)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Jordan	Al-Ghabeish & Allawi (2001)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Malaysia	Sivapragasm *et al.* (1999)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Peru	Delgado & Aguilar (1980); Redolfi *et al.* (1985)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Philippines	Baucas *et al.* (2003)
Braconidae	Opiinae	*Opius* sp.	*Opius* sp.	Sri Lanka	Nugaliyada (2000)
Braconidae	Opiinae	*Phaedrotoma luteoclypealis* van Achterberg & Salvo, (1997)	*Phaedrotoma luteoclypealis*	Argentina	van Achterberg & Salvo (1997); Salvo *et al.* (2005)
Braconidae	Opiinae	*Phaedrotoma mesoclypealis* van Achterberg & Salvo, (1997)	*Phaedrotoma mesoclypealis*	Argentina	van Achterberg & Salvo (1997); Salvo *et al.* (2005)
Braconidae	Opiinae	*Phaedrotoma scabriventris* (Nixon, 1955)	*Phaedrotoma scabriventris*	Argentina	
Braconidae	Opiinae	*Phaedrotoma scabriventris* (Nixon, 1955)	*Phaedrotoma scabriventris*	Argentina	van Achterberg & Salvo (1997); Salvo & Valladares (2002); Salvo *et al.* (2005)
Braconidae	Opiinae	*Phaedrotoma scabriventris* (Nixon, 1955)	*Phaedrotoma scabriventris*	Peru	Delgado & Aguilar (1980)
Braconidae	Opiinae	*Phaedrotoma* sp.	*Phaedrotoma* sp.	Argentina	van Achterberg & Salvo (1997)
Diapriidae	Diapriinae	*Trichopria* sp.	*Trichopria* sp.	Portugal	Mexia *et al.* (2004)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes delucchii*	Indonesia	Hidrayani *et al.* (2005)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes delucchii*	Philippines	Baucas *et al.* (2003)
Eulophidae	Endedoninae	*Asecodes* sp.	*Asecodes* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Endedoninae	*Chrysocharis ainsliei* Crawford, 1912	*Chrysocharis ainsliei*	Peru	Delgado & Aguilar (1980)
Eulophidae	Endedoninae	*Chrysocharis bedius* (Walker, 1842)	*Chrysocharis bedius*	Brazil	Dequech *et al.* (2010)
Eulophidae	Endedoninae	*Chrysocharis bedius* (Walker, 1842)	*Chrysocharis* sp. cerca *bedius*	Peru	Sanchez & Redolfi (1985)
Eulophidae	Endedoninae	*Chrysocharis bedius* (Walker, 1842)	*Chrysocharis bedius*	Reunion	Vayssieres *et al.* (2001)
Eulophidae	Endedoninae	*Chrysocharis c.f. aluta*	*Chrysocharis c.f. aluta*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, (1977)	*Chrysocharis caribea*	Argentina	Salvo & Valladares (1997); Salvo *et al.* (2005)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, (1977)	*Chrysocharis caribea*	Peru	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis flacilla* (Walker, 1842)	*Chrysocharis flacilla*	Argentina	Arce de Hamity & Neder de Román (1984); Salvo & Valladares (1997); Salvo *et al.* (2005)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Eulophidae	Endedoninae	*Chrysocharis flacilla* (Walker, 1842)	*Chrysocharis flacilla*	Chile	Neder de Román & Arce de Hamity (1984)
Eulophidae	Endedoninae	*Chrysocharis flacilla* (Walker, 1842)	*Chrysocharis phytomyzae, Chrysocharis flacilla*	Peru	Redolfi *et al.* (1985), Iannacone (1998); Delgado & Aguilar (1980); Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis ignota* Hansson, (1987)	*Chrysocharis ignota*	Guatemala	MacVean & Perez (1997)
Eulophidae	Endedoninae	*Chrysocharis orbicularis* (Nees, 1834)	*Chrysocharis orbicularis*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis oscinidis*	Canada	Bahlai *et al.* (2006)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	China	Chen *et al.* (2000); Pan *et al.* (2019)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Israel	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Japan	Shindo *et al.* (2005)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Malaysia	Sivapragasm *et al.* (1999); Nor Ahya & Nur Liyana (2018)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Taiwan	Chien & Chang (2013)
Eulophidae	Endedoninae	*Chrysocharis pubicornis* (Zetterstedt, 1838)	*Chrysocharis pubicornis*	China	Song *et al.* (2003); Pan et a. (2019)
Eulophidae	Endedoninae	*Chrysocharis pubicornis* (Zetterstedt, 1838)	*Chrysocharis pubicornis*	Japan	Shindo *et al.* (2005)
Eulophidae	Endedoninae	*Chrysocharis pubicornis* (Zetterstedt, 1838)	*Chrysocharis pubicornis*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	Argentina	Salvo & Valladares (1995)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	China	Pan *et al.* (2019)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	Costa Rica	Carballo *et al.* (1990); Hidalgo & Carballo (1991); Rodriguez (1997); Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	Peru	Delgado & Aguilar (1980); Redolfi *et al.* (1985); Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis* sp.	*Chrysocharis* sp.	Portugal	Mexia *et al.* (2004)
Eulophidae	Endedoninae	*Chrysocharis tristis* Hansson, (1987)	*Chrysocharis tristis*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis vonones* (Walker, 1839)	*Chrysocharis vonones*	Argentina	Salvo *et al.* (2005); Salvo & Valladares (1997)
Eulophidae	Endedoninae	*Chrysocharis vonones* (Walker, 1839)	*Chrysocharis brethesi*	Peru	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Chrysonotomyia* sp.	*Chrysonotomyia* sp.	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005)
Eulophidae	Endedoninae	*Chrysonotomyia* sp.	*Chrysonotomyia* sp.	Peru	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Closterocerus cinctipennis* Ashmead, 1888	*Closterocerus cinctipennis*	Peru	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Closterocerus pulcher* (Howard, 1897)	*Closterocerus pulcher*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Closterocerus* sp.	*Closterocerus* sp.	Brazil	Guimarães *et al.* (2010)
Eulophidae	Endedoninae	*Closterocerus* sp.	*Closterocerus* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Endedoninae	*Closterocerus* sp.	*Closterocerus* sp.	Peru	Delgado & Aguilar (1980)
Eulophidae	Endedoninae	*Neochrysocharis beasleyi* Fisher & La Salle, (2005)	*Neochrysocharis beasleyi*	Indonesia	Fisher & La Salle (2005)
Eulophidae	Endedoninae	*Neochrysocharis beasleyi* Fisher & La Salle, (2005)	*Neochrysocharis beasleyi*	Vietnam	Fisher & La Salle (2005)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Neochrysocharis diastatae*	Guatemala	Weintraub *et al.* (2017)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	China	Chen *et al.* (2000); Pan *et al.* (2019)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Indonesia	Hidrayani *et al.* (2005)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Israel	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Kenya	Foba *et al.* (2016)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Malaysia	Sivapragasm *et al.* (1999)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Turkey	Civelek *et al.* (2002)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Japan	Shindo *et al.* (2005)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Philippines	Baucas *et al.* (2003)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Closterocerus okazakii*	Taiwan	Chien & Chang (2013)
Eulophidae	Endedoninae	*Neochrysocharis* sp.	*Neochrysocharis* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Endedoninae	*Pediobius metallicus* (Nees, 1834)	*Pediobius metallicus*	China	Song *et al.* (2003); Pan *et al.* (2019)
Eulophidae	Endedoninae	*Pediobius metallicus* (Nees, 1834)	*Pediobius metallicus*	Israel	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Pediobius metallicus* (Nees, 1834)	*Pediobius metallicus*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Endedoninae	*Proacrias* sp.	*Proacrias* sp.	Easter Islands	Ripa *et al.* (1995)
Eulophidae	Endedoninae	*Proacrias thysanoides* (De Santis, 1972)	*Chrysonotomyia thysanoides*	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005)
Eulophidae	Endedoninae	*Proacrias thysanoides* (De Santis, 1972)	*Chrysonotomyia thysanoides*	Peru	Weintraub *et al.* (2017)
Eulophidae	Endedoninae	*Proacrias xenodice* (Walker, 1842)	*Chrysonotomyia xenodice*	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005); Videla *et al.* (2006)
Eulophidae	Endedoninae	*Proacrias xenodice* (Walker, 1842)	*Chrysonotomyia xenodice*	Easter Islands	Ripa *et al.* (1995)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Cirrospilus ambiguus* Hansson & LaSalle, (1996)	*Cirrospilus ambiguus*	Indonesia	Rauf & Shepard (2001)
Eulophidae	Eulophinae	*Diaulinopsis callichroma* Crawford, 1912	*Diaulinopsis callichroma*	Peru	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diaulinopsis* sp.	*Diaulinopsis* sp.	Argentina	Salvo & Valladares (1995)
Eulophidae	Eulophinae	*Diaulinopsis* sp.	*Diaulinopsis* sp.	Peru	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus albiscapus*	*Diglyphus albiscapus*	Japan	Shindo *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Argentina	Salvo *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Columbia	Cure & Cantor (2003)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Peru	Galantini Vignez & Redolfi de Huiza (1992); Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus bimaculatus* Zhu, LaSalle & Huang, (2000)	*Diglyphus bimaculatus*	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Israel	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Portugal	Godinho & Mexia (2000); Mexia *et al.* (2004)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Turkey	Civelek *et al.* (2002); Çıkman *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	China	Chen *et al.* (2000)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	Columbia	Andrade *et al.* (1989)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	Costa Rica	Carballo *et al.* (1990)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1833)	*Diglyphus isaea*	Portugal	Mexia *et al.* (2004)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Austria	Stolz & Blümel (1998)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	China	Chen *et al.* (2000); Luo *et al.* (2000); Luo *et al.* (2001); Chen *et al.* (2011); Pan *et al.* (2019)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Costa Rica	Rodriguez (1997)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Israel	Weintraub & Horowitz (1996); Weintraub (1999)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Italy	Calabretta *et al.* (1995); Burgio *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Japan	Shindo *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Kenya	Foba *et al.* (2016)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Lebanon	Noujeim *et al.* (2013)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Philippines	Baucas *et al.* (2003)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Portugal	Godinho & Mexia (2000)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Sri Lanka	Nugaliyada (2000)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	The Netherlands	van der Linden (1990)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Turkey	Civelek *et al.* (2002)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Portugal	Mexia *et al.* (2004)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Turkey	Çıkman *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus pachyneurus* Graham, 1963	*Diglyphus pachyneurus*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Diglyphus pedicellus* Gordh & Hendrickson, 1979	*Diglyphus pedicellus*	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus poppoea* Walker, 1848	*Diglyphus poppoea*	Portugal	Godinho & Mexia (2000); Mexia *et al.* (2004)
Eulophidae	Eulophinae	*Diglyphus pulchripes* (Crawford, 1912)	*Diglyphus pulchripes*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	Argentina	Arce de Hamity & Neder de Roman (1984)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	China	Chen *et al.* (2000)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	Columbia	Hincapie *et al.* (1993)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	Costa Rica	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	Peru	Iannacone & Reyes (2001)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	South Africa	Visser & Weintraub (2001)
Eulophidae	Eulophinae	*Diglyphus* sp. *(near intermedius)*	*Diglyphus* sp. *(near intermedius)*	Costa Rica	Hidalgo & Carballo (1991)
Eulophidae	Eulophinae	*Diglyphus wani* Liu, Zhu & Yefremova, (2018)	*Diglyphus wani*	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Diglyphus websteri*	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005); Videla *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Diglyphus websteri*	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Solenotus websteri* , *Diglyphus websteri*	Peru	Campos (1977), Redolfi *et al.* (1985); Sanchez & Redolfi (1985); Iannacone (1998); Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Hemiptarsenus fulvicollis* Westwood, 1833	*Hemiptarsenus fulvicollis*	Portugal	Mexia *et al.* (2004)
Eulophidae	Eulophinae	*Hemiptarsenus ornatus* (Nees, 1834)	*Hemiptarsenus ornatus*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Hemiptarsenus* sp.	*Hemiptarsenus* sp.	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Hemiptarsenus* sp.	*Hemiptarsenus* sp.	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Hemiptarsenus unguicellus* (Zetterstedt, 1838)	*Hemiptarsenus unguicellus*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	China	Chen *et al.* (2000); Chen *et al.* (2011)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Indonesia	Shepard *et al.* (1998); Prijono *et al.* (2004)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Kenya	Foba *et al.* (2016)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Malaysia	Sivapragasm *et al.* (1999); Nor Ahya & Nur Liyana (2018)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Philippines	Baucas *et al.* (2003)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Sri Lanka	Nugaliyada (2000)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Israel	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Meruacesa* sp.	*Meruacesa* sp.	Kenya	Foba *et al.* (2016)
Eulophidae	Eulophinae	*Pnigalio incompletus* (Bouček, 1971)	*Pnigalio incompletus*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Pnigalio katonis* (Ishii, 1953)	*Pnigalio katonis*	China	Song *et al.* (2003)
Eulophidae	Eulophinae	*Pnigalio katonis* (Ishii, 1953)	*Pnigalio katonis*	Philippines	Baucas *et al.* (2003)
Eulophidae	Eulophinae	*Pnigalio soemius* (Walker, 1839)	*Pnigalio soemius*	Israel	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Pnigalio* sp.	*Pnigalio* sp.	China	Pan *et al.* (2019)
Eulophidae	Eulophinae	*Pnigalio* sp.	*Pnigalio* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Eulophinae	*Zagrammosoma latilineatum* Ubaidillah, (2000)	*Zagrammosoma latilineatum*	Indonesia	Ubaidillah *et al.* (2000)
Eulophidae	Eulophinae	*Zagrammosoma multilineatum* (Ashmead, 1888)	*Zagrammosoma multilineatum*	Peru	Weintraub *et al.* (2017)
Eulophidae	Eulophinae	*Zagrammosoma* sp.	*Zagrammosoma* sp.	Peru	Delgado & Aguilar (1980)
Eulophidae	Eulophinae	*Zagrammosoma* sp.	*Zagrammosoma* sp.	Philippines	Baucas *et al.* (2003)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae* Hansson & LaSalle, (1996)	*Quadrastichus liriomyzae*	Philippines	Baucas *et al.* (2003)
Eulophidae	Tetrastichinae	*Quadrastichus* sp.	*Quadrastichus* sp.	Guatemala	Weintraub *et al.* (2017)
Eulophidae	Tetrastichinae	*Quadrastichus* sp.	*Quadrastichus* sp.	Indonesia	Rauf & Shepard (2001)
Eulophidae	Tetrastichinae	*Quadrastichus* sp.	*Quadrastichus* sp.	Israel	Weintraub *et al.* (2017)
Figitidae	Eucoilinae	*Alloxysta* sp.	*Alloxysta* sp.	Easter Islands	Ripa *et al.* (1995)
Figitidae	Eucoilinae	*Ganaspidium* sp.	*Ganaspidium* sp.	Peru	Redolfi *et al.* (1985); Sánchez & Redolfi (1985); Iannacone (1998); Weintraub *et al.* (2017)
Figitidae	Eucoilinae	*Gronotoma adachiae* Beardsley, 1988	*Gronotoma adachiae*	China	Abe & Konishi (2012)
Figitidae	Eucoilinae	*Gronotoma micromorpha* (Perkins, 1910)	*Gronotoma micromorpha*	Indonesia	Prijono *et al.* (2004); Tantowijoyo & Hoffmann (2010); La Daha (2011)
Figitidae	Eucoilinae	*Gronotoma* sp.	*Gronotoma sp*	China	Pan *et al.* (2019)
Figitidae	Eucoilinae	*Gronotoma* sp.	*Gronotoma* sp.	Guatemala	MacVean & Perez (1997)
Figitidae	Eucoilinae	*Gronotoma* sp.	*Gronotoma* sp.	Jordan	Al-Ghabeish & Allawi (2001)
Figitidae	Eucoilinae	*Moneucoela* sp.	*Moneucoela* sp.	Guatemala	MacVean & Perez (1997)
Figitidae	Eucoilinae	*Sinatra pacifica*	*Disorygma pacifica*	Guatemala	MacVean & Perez (1997)
Figitidae	Eucoilinae	*Tribliographa* sp.	*Tribliographa* sp.	Columbia	Andrade *et al.* (1989)
Figitidae	Eucoilinae	*Zaeucoila clavatus* (Diaz, 1976)	*Agrostocynips clavatus*	Argentina	Salvo & Valladares (1995); Salvo *et al.* (2005); Videla *et al.* (2006)
Figitidae	Eucoilinae	*Zaeucolia* sp.	*Zaeucolia* sp.	Guatemala	Weintraub *et al.* (2017)
Pteromalidae		Pteromalidae species	Pteromalidae species	Argentina	Salvo & Valladares (1995)
Pteromalidae		Pteromalidae species	Pteromalidae species	Israel	Weintraub *et al.* (2017)
Pteromalidae		Pteromalidae species	Pteromalidae species	Portugal	Mexia *et al.* (2004)
Pteromalidae	Miscogastrinae	*Halticoptera arduine* (Walker, 1843)	*Halticoptera arduine*	Argentina	de Santis (1987)
Pteromalidae	Miscogastrinae	*Halticoptera arduine* (Walker, 1843)	*Halticoptera arduine*	Argentina	Neder de Román (2000)
Pteromalidae	Miscogastrinae	*Halticoptera arduine* (Walker, 1843)	*Halticoptera arduine*	Chile	Neder de Román (2000)
Pteromalidae	Miscogastrinae	*Halticoptera arduine* (Walker, 1843)	*Halticoptera arduine*	Peru	Redolfi *et al.* (1985); Sanchez & Redolfi (1985); Iannacone (1998); Iannacone & Reyes (2001); Weintraub *et al.* (2017)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Austria	Stolz & Blümel (1998)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Canada	Bahlai *et al.* (2006)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	China	Chen *et al.* (2000)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	China	Song *et al.* (2003)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Guatemala	Weintraub *et al.* (2017)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Jordan	Al-Ghabeish & Allawi (2001)
Pteromalidae	Miscogastrinae	*Halticoptera helioponi* De Santis ,1976	*Halticoptera helioponi*	Argentina	Salvo & Valladares (1995); Salvo & Valladares (2002); Salvo *et al.* (2005); Videla *et al.* (2006)
Pteromalidae	Miscogastrinae	*Halticoptera patellana* (Dalman, 1818)	*Halticoptera patellana*	Peru	Delgado & Aguilar (1980)
Pteromalidae	Miscogastrinae	*Halticoptera peruviana* De Santis, (1987)	*Halticoptera peruviana*	Argentina	de Santis (1987)

Family	Sub-family	Full Name	Parasitoid name in text	Country	Reference
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Argentina	Salvo *et al.* (2005)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Columbia	Hincapie *et al.* (1993)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Easter Islands	Ripa *et al.* (1995)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Guatemala	MacVean & Perez (1997)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Jordan	Al-Ghabeish & Allawi (2001)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Peru	Delgado & Aguilar (1980); Weintraub *et al.* (2017)
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	*Halticoptera* sp.	Portugal	Mexia *et al.* (2004)
Pteromalidae	Miscogastrinae	*Thinodytes cyzicus* (Walker, 1839)	*Thinodytes cyzicus*	China	Chen *et al.* (2000)
Pteromalidae	Miscogastrinae	*Thinodytes* sp.	*Thinodytes* sp.	Argentina	Salvo *et al.* (2005)
Pteromalidae	Pteromalinae	*Heteroschema* sp.	*Heteroschema* sp.	Peru	Delgado & Aguilar (1980)
Pteromalidae	Pteromalinae	*Notoglyptus tzeltales* Heydon, 1988	*Notoglyptus tzeltales*	Guatemala	MacVean & Perez (1997)
Pteromalidae	Pteromalinae	*Sphegigaster* sp.	*Sphegigaster* sp.	Indonesia	Rauf & Shepard (2001)
Pteromalidae	Pteromalinae	*Trichomalopsis* sp.	*Trichomalopsis* sp.	China	Chen *et al.* (2000)
Tetracampidae	Platynocheilinae	*Platynocheilus cuprifrons* (Nees, 1834)	*Platynocheilus cuprifrons*	Israel	Weintraub *et al.* (2017)
Tetracampidae	Tetracampinae	*Epiclerus* sp.	*Epiclerus* sp.	Portugal	Mexia *et al.* (2004)

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Appendix 7.2 Parasitoid species reared from *Liriomyza trifolii*

Specimens reared from mixed collections of *Liriomyza* spp., which included *L. trifolii*, were not included in the list. As with any compilation of species names from the scientific literature, there will be uncertainty about the validity of some species and sampling intensity varies dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem (Grenouillet *et al.* 1993; Noyes 1994; Shaw 1994). Given the nomenclatural changes that have occurred in time, the currently accepted name is given, together with the name used in the original reference. All references for the Appendix are appended.

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Braconidae	Alysiinae	*Dacnusa nipponica* Takada, 1977	*Dacnusa nipponica*	Japan	Saito *et al.* (1996); Cai & Saito (2011)
Braconidae	Alysiinae	*Dacnusa sasakawai* Takada, 1977	*Dacnusa sasakawai*	Japan	Amano *et al.* (2008); Cai & Saito (2011)
Braconidae	Alysiinae	*Pseudopezomachus masii* Nixon, 1940	*Pseudopezomachus masii*	Egypt	Hegazi *et al.* (2014); Neama & Hegazi (2014)
Braconidae	Opiinae	*Opius ambiguus* Wesmael, 1835	*Opius ambiguus*	Turkey	Çıkman & Uygun (2003)
Braconidae	Opiinae	*Opius basalis* Fischer, 1958	*Opius basalis*	Turkey	Çikman (2006); Çikman et al. (2006); Neama & Hegazi (2014); Ali et al. (2020)
Braconidae	Opiinae	*Opius bruneipes* Gahan, 1913	*Opius bruneipes*	USA	Neuenschwander *et al.* (1987); Schuster *et al.* (1991); Schuster & Wharton (1993); Hernández *et al.* (2009)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidatus*	China	Liu *et al.* (2012); Wang *et al.* (2017)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	Mexico	Valenzuela-Escoboza *et al.* (2010)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	Senegal	Neuenschwander *et al.* (1987)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	USA	Stegmaier (1972); Schuster *et al.* (1991); Schuster & Wharton (1993)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Egypt	Bassiony *et al.* (2017)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Indonesia	Yasa *et al.* (2020)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Mexico	Holguín-Peña *et al.* (2019)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Mexico	Valenzuela-Escoboza *et al.* (2010)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Réunion	Olivera & Bordat (1996)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	USA	Neuenschwander *et al.* (1987); Schuster *et al.* (1991); Schuster & Wharton (1993); Hernández *et al.* (2009); Li *et al.* (2012)
Braconidae	Opiinae	*Opius exigua* Wesmael, 1835	*Opius exiguus*	India	Sharma & Kumar (2017)
Braconidae	Opiinae	*Opius exigua* Wesmael, 1835	*Opius exiguus*	Turkey	Çıkman & Uygun (2003)
Braconidae	Opiinae	*Opius gafsaensis* Fischer, 1964	*Opius gafsaensis*	Turkey	Çıkman & Uygun (2003)
Braconidae	Opiinae	*Opius nr brownsvillensis*	*Opius nr brownsvillensis*	USA	Hernández *et al.* (2009)
Braconidae	Opiinae	*Opius osogovoensis* Fischer, 1964	*Opius osogovoensis*	Turkey	Çıkman & Uygun (2003)
Braconidae	Opiinae	*Opius pallipes* Wesmael, 1835	*Opius pallipes*	Egypt	Bayoumy *et al.* (2018)
Braconidae	Opiinae	*Opius pallipes* Wesmael, 1835	*Opius pallipes*	Turkey	Çıkman & Uygun (2003)
Braconidae	Opiinae	*Opius sp.*	*Opius* sp.	Egypt	Ali *et al.* (2020)
Braconidae	Opiinae	*Opius sp.*	*Opius* sp.	Japan	Amano *et al.* (2008)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Braconidae	Opiinae	*Opius sp.*	*Opius* sp.	Spain	Cabello *et al.* (1994)
Braconidae	Opiinae	*Opius sp.*	*Opius* sp.	Taiwan	Lin & Wang (1992); Chien & Ku (1998)
Braconidae	Opiinae	*Opius sp.*	*Opius* sp.	Venezuela	Chirinos *et al.* (2014)
Braconidae	Opiinae	*Opius sp. 1*	*Opius* sp. *1*	USA	Hernández *et al.* (2009)
Braconidae	Opiinae	*Opius sp. 2*	*Opius* sp. *2*	USA	Hernández *et al.* (2009)
Braconidae	Opiinae	*Opius spp.*	*Opius* sp.	USA	Schuster & Price (1985); Hernández *et al.* (2009)
Braconidae	Opiinae	*Opius thoracosema sp. 3*	*Opius thoracosema* sp. *3*	USA	Hernández *et al.* (2009)
Braconidae	Opiinae	*Phaedrotoma biroi* (Fischer, 1960)	*Opius biroi*	China	Xing *et al.* (2017)
Braconidae	Tersilochinae	*Allophrys sp.*	*Allophrys* sp.	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Apleurotropis kumatai* (Kamijo, 1977)	*Apleurotropis kumatai*	Japan	Cai & Saito (2011)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes delucchii*	India	Sharma & Kumar (2017)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes delucchii*	Japan	Arakaki & Kinjo (1998)
Eulophidae	Endedoninae	*Asecodes erxias* (Walker, 1848)	*Asecodes erxias*	India	Sharma & Kumar (2017)
Eulophidae	Endedoninae	*Asecodes erxias* (Walker, 1848)	*Asecodes erxias; Teleopterus erxias*	Japan	Saito *et al.* (1996); Nishino & Uchida (1999)
Eulophidae	Endedoninae	*Asecodes sp.*	*Asecodes* sp.	USA	Hernández *et al.* (2009)
Eulophidae	Endedoninae	*Chrysocharis ainsliei* Crawford, 1912	*Chrysocharis ainsliei*	USA	Chandler (1982); Trumble & Nakakihara (1983)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribae*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribae*	USA	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Chrysocharis liriomyzae* Delucchi, 1954	*Chrysocharis liriomyzae*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Chrysocharis orbicularis* (Nees, 1834)	*Chrysocharis orbicularis*	Italy	Rizzo & Massa (2002)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis oscinidis*	China	Liu *et al.* (2012)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis parksi*	Mexico	Valenzuela-Escoboza *et al.* (2010)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis oscinidis*	Tonga	Johnson (1993)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis parksi*	USA	Parrella et al. (1982); Trumble & Nakakihara (1983); Schuster & Price (1985; Neuenschwander et al. (1987); Schuster et al. (1991);
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	China	Chen *et al.* (2006); Liu *et al.* (2012); Wang *et al.* (2017)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Egypt	Neama & Hegazi (2014); Ali *et al.* (2020)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Japan	Nishino *et al.* (1997); Arakaki & Kinjo (1998); Nishino & Uchida (1999); Amano *et al.* (2008); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Taiwan	Lin & Wang (1992); Chien & Ku (1998)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Turkey	Civelek & La Salle (2005)
Eulophidae	Endedoninae	*Chrysocharis pubicornis* (Zetterstedt, 1838)	*Chrysocharis pubicornis*	Japan	Cai & Saito (2011)
Eulophidae	Endedoninae	*Chrysocharis sp.*	*Chrysocharis* sp.	Egypt	Abul Fadl & El-Khawas (2009)
Eulophidae	Endedoninae	*Chrysocharis sp.*	*Chrysocharis* sp.	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Endedoninae	*Chrysocharis sp.*	*Chrysocharis* sp.	Mexico	Holguín-Peña *et al.* (2019)
Eulophidae	Endedoninae	*Chrysocharis sp.*	*Chrysocharis* sp.	USA	Hernández *et al.* (2009)
Eulophidae	Endedoninae	*Chrysonotomyia ricini* Parshuram & Agnihotri, 2017	*Chrysonotomyia ricini*	India	Murugan & Dhandapani (2006); Parshuram & Agnihotri (2017)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Endedoninae	*Chrysonotomyia smaragdulus* (Graham, 1963)	*Chrysonotomyia smaragdula*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Chrysonotomyia sp.*	*Chrysonotomyia sp.*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Endedoninae	*Chrysonotomyia sp.*	*Chrysonotomyia sp.*	USA	Chandler (1982)
Eulophidae	Endedoninae	*Chrysonotomyia* sp. near *leptocera*	*Chrysonotomyia* sp. near *leptocera*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Chrysonotomyia sp.F*	*Chrysonotomyia* sp.*F*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Cirrospilus sp. near cinctiventris*	*Cirrospilus sp. near cinctiventris*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Closterocerus agromyzae* Narayanan, Subba Rao and Ramachandra, 1960	*Closterocerus agromyzae*	India	Reji *et al.* (2003)
Eulophidae	Endedoninae	*Closterocerus cinctipennis* Ashmead, 1888	*Closterocerus cinctipennis*	Mexico	Valenzuela-Escoboza *et al.* (2010); Holguín-Peña *et al.* (2019)
Eulophidae	Endedoninae	*Closterocerus cinctipennis* Ashmead, 1888	*Closterocerus cinctipennis*	USA	Stegmaier (1966); Hernández *et al.* (2009)
Eulophidae	Endedoninae	*Closterocerus cinctipennis* Ashmead, 1888	*Closterocerus cintipennis*	Venezuela	Chirinos *et al.* (2014)
Eulophidae	Endedoninae	*Closterocerus purpureus* (Howard, 1897)	*Closterocerus purpureus*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Closterocerus utahensis* Crawford, 1912	*Closterocerus utahensis*	USA	Palumbo *et al.* (1994)
Eulophidae	Endedoninae	*Neochrysocharis agromyzae* (Crawford, 1913)	*Derostenus agromyzae*	USA	Stegmaier (1966)
Eulophidae	Endedoninae	*Neochrysocharis ambitiosus* Hansson, 1990	*Neochrysocharis ambitiosa*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Neochrysocharis arvensis* Graham, 1963	*Neochrysocharis arvensis*	Turkey	Gençer (2004)
Eulophidae	Endedoninae	*Neochrysocharis chlorogaster* (Erdös, 1966)	*Neochrysocharis chlorogaster*	Turkey	Ulubilir & Yabas (2000)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Neochrysocharis punctiventris*	China	Wang *et al.* (2017)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Chrysonotomyia punctiventris; Neochrysocharis punctiventris*	USA	Trumble & Nakakihara (1983); Schuster & Price (1985); Lynch & Johnson (1987); Neuenschwander *et al.* (1987); Schuster *et al.* (1991); Schuster & Wharton (1993)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Walker, 1833)	*Neochrysocharis formosa*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chysonotomyia formosa*	China	Liu *et al.* (2012); Wang *et al.* (2017)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Egypt	Ali *et al.* (2020)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosa*	Guam	Schreiner *et al.* (1986)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	India	Sharma & Kumar (2017)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Indonesia	Yasa *et al.* (2020)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Closterocerus formosus*	Iran	Talebi *et al.* (2005); Asadi *et al.* (2006); Dousti *et al.* (2008); Shahreki *et al.* (2016)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chysonotomyia formosa*; *Neochrysocharis formosa*	Italy	Del Bene (1989); Rizzo & Massa (2002)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Japan	Saito *et al.* (1996); Arakaki & Kinjo (1998); Nishino & Uchida (1999); Amano *et al.* (2008); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Philippines	Baucas *et al.* (2003); Arida *et al.* (2013)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia* sp., *formosa group*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosa*	Spain	Cabello *et al.* (1994)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia* sp.	Taiwan	Lin & Wang (1992); Chien & Ku (1998)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosa*; *Closterocerus formosus*; *Neochrysocharis formosa*	Turkey	Ulubilir & Yabas (2000); Gençer (2004); Çikman *et al.* (2006); Çikman & Çömlekçioğlu (2006); Çikman (2012)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Derostenus variipes* ; *Neochrysocharis formosa*	USA	Stegmaier (1966); Hernández *et al.* (2009)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Venezuela	Chirinos *et al.* (2014)
Eulophidae	Endedoninae	*Neochrysocharis indicus* Khan, Agnihotri and Sushil, 2005	*Closterocerocus indicus*; *Closterocerus indica*; *Chrysocharis indicus*	India	Bhat *et al.* (2009); Gupta & Poorani (2009); Sharma & Kumar (2017)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*; *Chysonotomyia okazakii*	China	Chen *et al.* (2006); Liu *et al.* (2012)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Indonesia	Yasa *et al.* (2020)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Japan	Saito *et al.* (1996); Arakaki & Kinjo (1998); Nishino & Uchida (1999); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Philippines	Baucas *et al.* (2003); Arida *et al.* (2013)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Chrysonotomyia okazakii*	Taiwan	Lin & Wang (1992); Chien & Ku (1998)
Eulophidae	Endedoninae	*Neochrysocharis pictipes* (Crawford, 1912)	*Neochrysocharis albipes*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Neochrysocharis sericea* (Erdös, 1954)	*Neochrysocharis sericea*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Neochrysocharis sp.*	*Neochrysocharis* sp.	Egypt	Abul Fadl & El-Khawas (2009); Ali *et al.* (2020)
Eulophidae	Endedoninae	*Neochrysocharis sp.*	*Neochrysocharis* sp.	Japan	Amano *et al.* (2008); Cai & Saito (2011)
Eulophidae	Endedoninae	*Neochrysocharis sp.*	*Neochrysocharis* sp.	Mexico	Valenzuela-Escoboza *et al.* (2010)
Eulophidae	Endedoninae	*Neochrysocharis sp.*	*Neochrysocharis* sp.	USA	Patel & Schuster (1992)
Eulophidae	Endedoninae	*Pediobius metallicus* (Nees, 1834)	*Pediobius metallicus*	Turkey	Çikman (2006)
Eulophidae	Endedoninae	*Pediobius metallicus* (Nees, 1834)	*Pediobius acantha*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Endedoninae	*Pediobius sp.*	*Pediobius* sp.	Japan	Arakaki & Kinjo (1998)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	Iran	Talebi *et al.* (2005); Asadi *et al.* (2006)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	Italy	Del Bene (1989)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	Spain	Cabello *et al.* (1994)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vitatus*	Turkey	Çikman (2006); Çikman & Çömlekçioğlu (2006)
Eulophidae	Eulophinae	*Cirrospilus brevicorpus* Shafee & Rizvi, 1988	*Cirrospilus ambiguus*	Taiwan	Hansson & LaSalle (1996); Chien & Ku (1998)
Eulophidae	Eulophinae	*Cirrospilus sp.*	*Cirrospilus* sp.	Egypt	Abul Fadl & El-Khawas (2009)
Eulophidae	Eulophinae	*Cirrospilus sp. near cinctiventris*	*Cirrospilus* sp. *near cinctiventris*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Cirrospilus variegatus group*	*Cirrospilus variegatus group*	USA	Hernández *et al.* (2009)
Eulophidae	Eulophinae	*Diaulinopsis arenaria* (Erdös, 1951)	*Diaulinopsis arenaria*	Iran	Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Diaulinopsis arenaria* (Erdös, 1951)	*Diaulinopsis arenaria*	Jordan	Al-Ghabeish & Allawi (2001)
Eulophidae	Eulophinae	*Diaulinopsis callichroma* Crawford, 1912	*Diaulinopsis callichroma*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Diaulinopsis callichroma* Crawford, 1912	*Diaulinopsis callichroma*	USA	Schuster & Price (1985); Schuster *et al.* (1991); Schuster & Wharton (1993)
Eulophidae	Eulophinae	*Diglyphus albiscapus* Erdos, 1951	*Diglyphus albiscapus*	China	Zhu *et al.* (2000)
Eulophidae	Eulophinae	*Diglyphus albiscapus* Erdös, 1951	*Diglyphus albiscapus*	Japan	Saito *et al.* (1996); Arakaki & Kinjo (1998); Nishino & Uchida (1999); Amano *et al.* (2008); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Colombia	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	USA	Parrella *et al.* (1982); Trumble & Nakakihara (1983); Hara (1986); Neuenschwander *et al.* (1987); Heinz & Parrella (1990); Schuster *et al.* (1991); Schuster & Wharton (1993)
Eulophidae	Eulophinae	*Diglyphus chabrias* (Walker, 1838)	*Diglyphus chabrias*	Spain	Cabello *et al.* (1994)
Eulophidae	Eulophinae	*Diglyphus chabrias* (Walker, 1838)	*Diglyphus chabrias*	Turkey	Gençer (2004)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Egypt	Abul Fadl & El-Khawas (2009)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Iran	Talebi *et al.* (2005); Asadi *et al.* (2006); Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Italy	Hansson & Navone (2017)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Jordan	Al-Ghabeish & Allawi (2001)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Turkey	Çikman *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus horticola* Khan, 1985	*Diglyphus horticola*	India	Sharma & Kumar (2017)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	USA	Stegmaier (1972); Trumble & Nakakihara (1983); Schuster & Price (1985); Hara (1986); Neuenschwander *et al.* (1987); Schuster *et al.* (1991); Patel & Schuster (1992); Schuster & Wharton (1993)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	China	Chen *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Egypt	Abul Fadl & El-Khawas (2009); Neama & Hegazi (2014); Bassiony *et al.* (2017); Bayoumy *et al.* (2018); Ali *et al.* (2020)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	India	Sharma & Kumar (2017)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Iran	Talebi *et al.* (2005); Asadi *et al.* (2006); Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Italy	Del Bene (1989); Rizzo & Massa (2002); Hansson & Navone (2017)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Japan	Saito *et al.* (1996); Nishino & Uchida (1999); Amano *et al.* (2008); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Philippines	Baucas *et al.* (2003); Arida *et al.* (2013)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Spain	Cabello *et al.* (1994)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Turkey	Ulubilir & Yabas (2000); Çıkman & Uygun (2003); Gençer (2004); Çikman (2006); Çikman *et al.* (2006); Çikman & Çömlekçioğlu (2006); Gençer (2009); Çikman & La Salle (2011); Çikman (2012)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	USA	Parrella *et al.* (1982); Hara (1986); Hernández *et al.* (2009)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Japan	Saito *et al.* (1996); Cai & Saito (2011)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Turkey	Çıkman & Uygun (2003); Çikman (2006); Çikman & Çömlekçioğlu (2006); Çikman (2012)
Eulophidae	Eulophinae	*Diglyphus poppoea* Walker, 1848	*Diglyphus poppoea*	Portugal	Godinho & Mexia (2000)
Eulophidae	Eulophinae	*Diglyphus pulchripes* (Crawford, 1912)	*Diglyphus pulchripes*	Iran	Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus pulchripes* (Crawford, 1912)	*Diglyphus pulchripes*	USA	Stegmaier (1972)
Eulophidae	Eulophinae	*Diglyphus pusztensis* (Erdös and Novicky, 1951)	*Diglyphus pusztensis*	Japan	Nishino & Uchida (1999)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	Egypt	Ali *et al.* (2020)
Eulophidae	Eulophinae	*Diglyphus* sp.	*Diglyphus* sp.	USA	Schuster & Price (1985)
Eulophidae	Eulophinae	*Elachertus sp. L*	*Elachertus* sp. *L*	Trinidad	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Hemiptarsenus ornatus* (Nees, 1834)	*Hemiptarsenus dropion*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Hemiptarsenus ornatus* (Nees, 1834)	*Hemiptarsenus dropion*	Italy	Del Bene (1989)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Eulophinae	*Hemiptarsenus sp.*	*Hemiptarsenus* sp.	Turkey	Gençer (2004)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	China	Chen *et al.* (2006); Liu *et al.* (2012); Wang *et al.* (2017)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemitarsenus semiablaclavus*	Ethiopia	Abate (1987)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus semialbiclavus*	Guam	Schreiner *et al.* (1986)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	India	Srinivasan *et al.* (1995); Bhat *et al.* (2009); Sharma & Kumar (2017)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Indonesia	Yasa *et al.* (2020)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus* sp. *near semialbiclavus*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Japan	Saito *et al.* (1996); Amano *et al.* (2008); Arakaki & Kinjo (1998); Cai & Saito (2011)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Philippines	Baucas *et al.* (2003); Arida *et al.* (2013)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus semialbiclava*	Senegal	Neuenschwander *et al.* (1987)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Spain	Cabello *et al.* (1994)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Taiwan	Lin & Wang (1992); Chien & Ku (1998)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Egypt	Ali *et al.* (2020)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Iran	Talebi *et al.* (2005); Asadi *et al.* (2006); Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	South Korea	Cheol *et al.* (2002)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Spain	Cabello *et al.* (1994)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Turkey	Çikman (2006); Çikman & Çömlekçioğlu (2006); Çikman *et al.* (2006)
Eulophidae	Eulophinae	*Meruacesa liriomyzae* (Bouček 1988)	*Meruna liriomyzae*	Kenya	Bouček (1988)
Eulophidae	Eulophinae	*Pnigalio cristatus* (Ratzeburg, 1848)	*Ratzeburgiola cristatus*	Iran	Dousti *et al.* (2008)
Eulophidae	Eulophinae	*Pnigalio incompletus* (Bouček , 1971)	*Ratzeburgia incompleta*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Pnigalio katonis* (Ishii, 1953)	*Pnigalio katonis*	Japan	Saito *et al.* (1996); Amano *et al.* (2008); Saito *et al.* (2008); Cai & Saito (2011)
Eulophidae	Eulophinae	*Pnigalio katonis* (Ishii, 1953)	*Pnigalio katonis*	Philippines	Baucas *et al.* (2003); Arida *et al.* (2013)
Eulophidae	Eulophinae	*Pnigalio soemius* (Walker, 1839)	*Pnigalio soemias*	Israel	Freidberg & Gijswijtz (1983)
Eulophidae	Eulophinae	*Pnigalio sp.*	*Pnigalio* sp.	Italy	Del Bene (1989)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Eulophidae	Eulophinae	*Pnigalio sp.*	*Pnigalio* sp.	USA	Hernández *et al.* (2009)
Eulophidae	Eulophinae	*Pnigalio sp.- nr. pectinicornis*	*Pnigalio* sp.- *nr. pectinicornis*	Iran	Talebi *et al.* (2005)
Eulophidae	Eulophinae	*Sympiesis acalle* (Walker, 1848)	*Sympiesis acalle*	Iran	Shahreki *et al.* (2016)
Eulophidae	Eulophinae	*Sympiesis gordius* (Walker, 1839)	*Sympiesis gordi*	Turkey	Çıkman & Uygun (2003)
Eulophidae	Eulophinae	*Sympiesis sp.*	*Sympiesis* sp.	Turkey	Ulubilir & Yabas (2000)
Eulophidae	Eulophinae	*Zagrammosoma americanum* Girault, 1916	*Zagrammosoma americanum*	USA	Chandler (1982)
Eulophidae	Eulophinae	*Zagrammosoma lineaticeps* (Girault, 1915)	*Mirzagrammosoma lineaticeps*	USA	Stegmaier (1966)
Eulophidae	Eulophinae	*Zagrammosoma sp.*	*Zagrammosoma* sp.	Egypt	Neama & Hegazi (2014); Ali *et al.* (2020)
Eulophidae	Eulophinae	*Zagrammosoma sp.*	*Zagrammosoma* sp.	USA	Hernández *et al.* (2009)
Eulophidae	Eulophinae	*Zagrammosoma talitzkii* Bouček, 1961	*Cirrospilus talitzkii*	Turkey	Çikman & La Salle (2011)
Eulophidae	Eulophinae	*Zagrammosoma variegatum* (Masi, 1907)	*Zagrammosoma variegatum*	India	Kapadia (1995)
Eulophidae	Tetrastichinae	*Aprostocetus sp.*	*Aprostocetus* sp.	Turkey	Çikman & Çömlekçioğlu (2006)
Eulophidae	Tetrastichinae	*Baryscapus impeditus* (Nees, 1834)	*Baryscapus impeditus*	Iran	Dousti *et al.* (2008)
Eulophidae	Tetrastichinae	*Neotrichoporoides szelenyii* (Erdös, 1951)	*Neotrichoporoides szelenyii*	Iran	Shahreki *et al.* (2016)
Eulophidae	Tetrastichinae	*Oomyzus sp.*	*Oomyzus* sp.	Japan	Saito *et al.* (1996)
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae* Hansson & LaSalle, 1996	*Quadrastichus liriomyzae*	Japan	Hansson & LaSalle (1996); Arakaki & Kinjo (1998)
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae* Hansson & LaSalle, 1996	*Quadrastichus liriomyzae*	Philippines	Baucas *et al.* (2003)
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae* Hansson & LaSalle, 1996	*Quadrastichus liriomyzae*	Taiwan	Hansson & LaSalle (1996); Chien & Ku (1998)
Eulophidae	Tetrastichinae	*Quadrastichus plaquoi* Reina & La Salle, 2004	*Quadrastichus plaquoi*	India	Reina & La Salle (2004); Sharma & Kumar (2017)
Eulophidae	Tetrastichinae	Quadrastichus sp.	*Quadrastichus* sp.	Japan	Cai & Saito (2011)
Figitidae	Eucoilinae	*Banacuniculus nigrimanus* (Kieffer, 1907)	*Ganaspidium nigrimanus*	Mexico	Valenzuela-Escoboza *et al.* (2010)
Figitidae	Eucoilinae	*Banacuniculus nigrimanus* (Kieffer, 1907)	*Ganaspidium nigrimanus*	USA	Hernández *et al.* (2009)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidium utilis*	Guam	Nafus & Schreiner (1989)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidium hunteri*	Tonga	Johnson (1993)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidium hunteri*	USA	Hara (1986); Lynch& Johnson (1987)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidius utilis*	Venezuela	Chirinos *et al.* (2014)
Figitidae	Eucoilinae	*Ganaspidium pusillae* Weld, 1955	*Ganaspidium pusillae*	USA	Hernández *et al.* (2009)
Figitidae	Eucoilinae	*Ganaspidium sp.*	*Ganaspidium* sp.	USA	Schuster & Price (1985)
Figitidae	Eucoilinae	*Gronotoma fetura* (Quinlan, 1986)	*Eucoilidea fetura*	Senegal	Neuenschwander *et al.* (1987)
Figitidae	Eucoilinae	*Gronotoma guamensis* (Yoshimoto, 1962)	*Eucoilidea guamensis*	Guam	Schreiner *et al.* (1986)
Figitidae	Eucoilinae	*Gronotoma micromorpha* (Perkins, 1910)	*Eucoilidea micromorpha*	Guam	Schreiner *et al.* (1986)

Family	Sub-family	Parasitoid (current)	Parasitoid (in reference)	Country	Reference
Figitidae	Eucoilinae	*Gronotoma micromorpha* (Perkins, 1910)	*Gronotoma micromorpha*	Japan	Arakaki & Kinjo (1998)
Figitidae	Eucoilinae	*Kleidotoma sp.*	*Kleidotoma* sp.	Japan	Saito *et al.* (1996)
Figitidae	Eucoilinae	*Nordlanderia plowa* Quinlan, 1986	*Nordlanderia plowa*	Senegal	Neuenschwander *et al.* (1987)
Figitidae	Eucoilinae	*Sinatra pacifica* (Yoshimoto, 1962)	*Cothonaspis pacifica*	Guam	Schreiner *et al.* (1986)
Figitidae	Eucoilinae	*Sinatra pacifica* (Yoshimoto, 1962)	*Disorygma pacifica*	Mexico	Valenzuela-Escoboza *et al.* (2010)
Figitidae	Eucoilinae	*Sinatra pacifica* (Yoshimoto, 1962)	*Disorygma pacifica*	USA	Hernández *et al.* (2009)
Figitidae	Eucoilinae	*Zaeucoila robusta* (Ashmead, 1894)	*Agrostocynips robusta*	USA	Hernández *et al.* (2009)
Pteromalidae	Herbertiinae	*Herbertia Hernández* Burks, 1959	*Herbertia indica*	India	Bhat *et al.* (2009)
Pteromalidae	Miscogastrinae	*Halticoptera aenea* (Walker, 1833)	*Halticoptera aenea*	USA	Palumbo *et al.* (1994)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Egypt	Neama & Hegazi (2014); Bayoumy *et al.* (2018); Ali *et al.* (2020)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Japan	Saito *et al.* (1996)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Senegal	Neuenschwander *et al.* (1987)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Trinidad	Neuenschwander *et al.* (1987)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	USA	Stegmaier (1966); Trumble & Nakakihara (1983); Schuster & Price (1985); Neuenschwander *et al.* (1987); Schuster *et al.* (1991); Schuster & Wharton (1993)
Pteromalidae	Miscogastrinae	*Halticoptera longipetiolus* Hedqvist, 1975	*Halticoptera longipetiolus*	Turkey	Doganlar (2006)
Pteromalidae	Miscogastrinae	*Halticoptera nr. circulus*	*Halticoptera nr. circulus*	USA	Hernández *et al.* (2009)
Pteromalidae	Miscogastrinae	*Halticoptera patellana* (Dalman, 1818)	*Halticoptera patellana*	Turkey	Çikman (2012)
Pteromalidae	Miscogastrinae	*Halticoptera sp.*	*Halticoptera* sp.	Egypt	Neama & Hegazi (2014); Ali *et al.* (2020)
Pteromalidae	Miscogastrinae	*Halticoptera sp. 1*	*Halticoptera* sp. *1*	Taiwan	Lin & Wang (1992)
Pteromalidae	Miscogastrinae	*Halticoptera sp. 2*	*Halticoptera* sp. *2*	Taiwan	Lin & Wang (1992)
Pteromalidae	Pteromalinae	*Cyrtogaster vulgaris* Walker, 1833	*Cyrtogaster vulgaris*	Turkey	Çikman & Çömlekçioğlu (2006); Çikman *et al.* (2006)
Pteromalidae	Pteromalinae	*Sphegigaster brevicornis* (Walker, 1833)	*Sphegigaster brevicornis*	Turkey	Çikman *et al.* (2006)
Pteromalidae	Pteromalinae	*Sphegigaster hamugurivora* Ishii, 1953	*Sphegigaster hamugurivora*	Japan	Saito *et al.* (1996); Arakaki & Kinjo (1998)
Pteromalidae	Pteromalinae	*Sphegigaster sp.*	*Sphegigaster* sp.	Japan	Arakaki & Kinjo (1998)
Pteromalidae	Pteromalinae	*Trichomalopsis oryzae* Kamijo & Grissell, 1982	*Trichomalopsis oryzae*	Japan	Cai & Saito (2011)
Tetracampidae	Tetracampinae	*Epiclerus nomocerus* (Masi, 1934)	*Epiclerus nomocerus*	France	Franco & Panis (1991)

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Appendix 7.3 Parasitoid species reared from *Liriomyza sativae*

Specimens reared from mixed collections of *Liriomyza* spp., which included *L. sativae*, were not included in the list. This table is based on data from Ridland *et al.* (2020) with supplementary data added. As with any compilation of species names from the scientific literature, there will be uncertainty about the validity of some species and sampling intensity varies dramatically between countries, which usually reflects the scientific input into the problem rather than the actual size of the problem (Noyes 1994; Shaw 1994). Given the nomenclatural changes that have occurred in time, the currently accepted name is given, together with the name used in the original reference. All references for the Appendix are appended.

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Endedoninae	*Achrysocharoides zwoelferi* Delucchi, 1954	*Achrysocharoides zwoelferi*	USA	Gates *et al.* (2002)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes deluchii*	Indonesia	Tantowijoyo & Hoffmann (2010); Herlinadewi et al. (2013)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes deluchii*	Japan	Amano *et al.* (2008)
Eulophidae	Endedoninae	*Asecodes delucchii* (Bouček, 1971)	*Asecodes deluchii*	Vietnam	Tran (2009)
Eulophidae	Endedoninae	*Asecodes erxias* (Walker, 1848)	*Asecodes erxias*	Japan	Amano *et al.* (2008)
Eulophidae	Endedoninae	*Chrysocharis ainsliei* Crawford, 1912	*Chrysocharis ainsliei*	USA	Johnson *et al.* (1980); Chandler (1982)
Eulophidae	Endedoninae	*Chrysocharis ainsliei* Crawford, 1912	*Chrysocharis ainsliei*	USA	Oatman (1959)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribea*	Barbados	Bouček (1977)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribea*	Peru	Mujica & Kroschel (2011)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribea*	Trinidad	Bouček (1977)
Eulophidae	Endedoninae	*Chrysocharis caribea* Bouček, 1977	*Chrysocharis caribea*	Venezuela	Bouček (1977)
Eulophidae	Endedoninae	*Chrysocharis crassiscapus* (Thomson, 1878)	*Chrysocharis mallochi*	Canada	McClanahan (1977)
Eulophidae	Endedoninae	*Chrysocharis flacilla* (Walker, 1842)	*Chrysocharis flacilla*	Peru	Mujica & Kroschel (2011)
Eulophidae	Endedoninae	*Chrysocharis giraulti* Yoshimoto, 1973	*Chrysocharis giraulti*	USA	Oatman & Johnson (1981)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis viridis*	Canada	McClanahan (1977)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis oscinidis*	China	Xu *et al.* (1999)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis parksi*	USA	Johnson *et al.* (1980); Johnson (1987)
Eulophidae	Endedoninae	*Chrysocharis oscinidis* Ashmead, 1888	*Chrysocharis parksi*	USA	Oatman (1959)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Bangladesh	Mazumdar & Bhuiya (2016)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	China	Xu *et al.* (1999)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Japan	Amano *et al.* (2008)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Taiwan	Chien & Chang (2011)
Eulophidae	Endedoninae	*Chrysocharis pentheus* (Walker, 1839)	*Chrysocharis pentheus*	Vietnam	Tran (2009)
Eulophidae	Endedoninae	*Chrysocharis vonones* (Walker, 1839)	*Chrysocharis vonones*	Brazil	Costa-Lima *et al.* (2014)
Eulophidae	Endedoninae	*Chrysocharis vonones* (Walker, 1839)	*Chrysocharis brethesi*	Peru	Mujica & Kroschel (2011)
Eulophidae	Endedoninae	*Chrysonotomyia reticulata* Zheng & Zhan 2000	*Chrysonotomyia reticulata*	China	Sheng & Zhan (2000)
Eulophidae	Endedoninae	*Closterocerus cincinnatus* Girault, 1916	*Closterocerus cincinnatus*	USA	Gates *et al.* (2002)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Endedoninae	*Closterocerus cinctipennis* Ashmead, 1888	*Closterocerus cinctipennis*	USA	Harding (1965), Stegmaier (1972)
Eulophidae	Endedoninae	*Closterocerus purpureus* (Howard, 1897)	*Chrysonotomyia purpurea*	Trinidad	Bouček (1977)
Eulophidae	Endedoninae	*Closterocerus trifasciatus* Westwood, 1833	*Closterocerus trifasciatus*	USA	Oatman (1959)
Eulophidae	Endedoninae	*Closterocerus utahensis* Crawford, 1912	*Closterocerus utahensis*	USA	Johnson *et al.* (1980a), Gates *et al.* (2002)
Eulophidae	Endedoninae	*Neochrysocharis agromyzae* (Crawford, 1913)	*Achrysocharella agromyzae*	USA	Harding (1965), Stegmaier (1972)
Eulophidae	Endedoninae	*Neochrysocharis arizonensis* (Crawford, 1913)	*Neochrysocharis arizonensis*	USA	Gates *et al.* (2002)
Eulophidae	Endedoninae	*Neochrysocharis arizonensis* (Crawford, 1913)	*Derostenus arizonensis*	USA	Oatman (1959)
Eulophidae	Endedoninae	*Neochrysocharis beasleyi* Fisher & La Salle, 1985	*Neochrysocharis beasleyi*	Vietnam	Tran (2009)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Chrysonotomyia punctiventris*	China	Xu *et al.* (1999)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Achrysocharella diastatae*	USA	Stegmaier (1972)
Eulophidae	Endedoninae	*Neochrysocharis diastatae* (Howard, 1881)	*Chrysonotomyia punctiventris*	USA	Johnson *et al.* (1980a), Johnson (1987)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Bangladesh	Mazumdar & Bhuiya (2016)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosus*	China	Xu *et al.* (1999)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Indonesia	Tantowijoyo & Hoffmann (2010); Herlinadewi et al. (2013)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Closterocerus formosus*	Iran	Asadi *et al.* (2006); Shareki *et al.* (2016)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Japan	Amano *et al.* (2008)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Achrysocharella fullowayi*	USA	Harding (1965)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Achrysocharella variipes*	USA	Harding (1965); Stegmaier (1972)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosus*	USA	Lema & Poe (1979); Johnson (1987)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Chrysonotomyia formosa*	USA	Tryon & Poe (1981)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Derostenus variipes*	USA	Oatman (1959)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Vietnam	Tran (2009)
Eulophidae	Endedoninae	*Neochrysocharis formosa* (Westwood, 1833)	*Neochrysocharis formosa*	Indonesia	Herlinadewi *et al.* (2013)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Taiwan	Chien & Chang (2011)
Eulophidae	Endedoninae	*Neochrysocharis okazakii* Kamijo, 1978	*Neochrysocharis okazakii*	Vietnam	Tran (2009)
Eulophidae	Endedoninae	*Proacrias thysanoides* (De Santis, 1972)	*Proacrias thysanoides*	Peru	Mujica & Kroschel (2011)
Eulophidae	Eulophinae	*Cirrospilus brevicorpus* Shafee & Rizvi, 1988	*Cirrospilus ambiguus*	Malaysia	Sivapragasam et al. (1999)
Eulophidae	Eulophinae	*Cirrospilus brevicorpus* Shafee & Rizvi, 1988	*Cirrospilus ambiguus*	Vietnam	Tran (2009)
Eulophidae	Eulophinae	*Burkseus vittatus* (Walker, 1838)	*Cirrospilus vittatus*	Iran	Asadi *et al.* (2006)
Eulophidae	Eulophinae	*Diaulinopsis arenaria* (Erdös, 1951)	*Diaulinopsis arenaria*	China	Shi *et al.* (2000)
Eulophidae	Eulophinae	*Diaulinopsis callichroma* Crawford, 1912	*Diaulinopsis callichroma*	Peru	Mujica & Kroschel (2011)
Eulophidae	Eulophinae	*Diaulinopsis callichroma* Crawford, 1912	*Diaulinopsis callichroma*	USA	Oatman (1959), Stegmaier (1972), Gates *et al.* (2002)
Eulophidae	Eulophinae	*Diglyphus albiscapus* Erdös, 1951	*Diglyphus albiscapus*	Japan	Amano *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Canada	McClanahan (1975, 1977)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	Colombia	de la Cruz *et al.* (1989)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Diglyphus begini*	USA	Trumble & Nakakihara (1983), Heinz & Parrella (1990)
Eulophidae	Eulophinae	*Diglyphus begini* (Ashmead, 1904)	*Solenotus begini*	USA	Oatman (1959)
Eulophidae	Eulophinae	*Diglyphus chabrias* (Walker, 1838)	*Diglyphus chabrias*	Turkey	Yefremova *et al.* (2011)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Iran	Talebi *et al.* (2011)
Eulophidae	Eulophinae	*Diglyphus crassinervis* Erdös, 1958	*Diglyphus crassinervis*	Turkey	Yefremova *et al.* (2011)
Eulophidae	Eulophinae	*Diglyphus intermedius* (Girault, 1916)	*Diglyphus intermedius*	USA	Johnson *et al.* (1980a), Tryon & Poe (1981), Trumble & Nakakihara (1983)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	China	Xu *et al.* (1999)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Iran	Asadi *et al.* (2006), Fathipour *et al.* (2006)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Japan	Amano *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Turkey	Yefremova *et al.* (2011)
Eulophidae	Eulophinae	*Diglyphus isaea* (Walker, 1838)	*Diglyphus isaea*	Vietnam	Tran *et al.* (2005)
Eulophidae	Eulophinae	*Diglyphus minoeus* (Walker, 1838)	*Diglyphus minoeus*	Japan	Amano *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus pulchripes* (Crawford, 1912)	*Diglyphus pulchripes*	Canada	McClanahan (1977), Stegmaier (1972)
Eulophidae	Eulophinae	*Diglyphus pulchripes* (Crawford, 1912)	*Solenotus pulchripes*	USA	Oatman (1959)
Eulophidae	Eulophinae	*Diglyphus pusztensis* (Erdös & Novicky, 1951)	*Diglyphus pusztensis*	Japan	Amano *et al.* (2008)
Eulophidae	Eulophinae	*Diglyphus sensilis* Yefremova, 2011	*Diglyphus sensilis*	Turkey	Yefremova *et al.* (2011)
Eulophidae	Eulophinae	*Diglyphus wani* Liu, Zhu & Yefremova, 2018	*Diglyphus wani*	China	Ye *et al.* (2018)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Diglyphus websteri*	Argentina	Valladares *et al.* (1999)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Diglyphus websteri*	Peru	Mujica & Kroschel (2011)
Eulophidae	Eulophinae	*Diglyphus websteri* (Crawford, 1912)	*Solenotus websteri; Diglyphus websteri*	USA	Oatman (1959); Harding (1965)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Australia	Elia Pirtle (unpublished data)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	China	Xu *et al.* (1999)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus semialbiclavus*	Guam	Johnson (1993)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Indonesia	Rauf *et al.* (2000); Herlinadewi et al. (2013)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Japan	Amano *et al.* (2008)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Sri Lanka	Nagalingam *et al.* (2007)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Taiwan	Chien & Chang (2007)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus semialbiclavus*	USA	Johnson (1987)
Eulophidae	Eulophinae	*Hemiptarsenus varicornis* (Girault, 1913)	*Hemiptarsenus varicornis*	Vietnam	Tran (2009)
Eulophidae	Eulophinae	*Hemiptarsenus wailesellae* Nowicki, 1929	*Hemiptarsenus wailesellae*	Iran	Talebi *et al.* (2011)
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi* Erdös, 1951	*Hemiptarsenus zilahisebessi*	Iran	Talebi *et al.* (2011), Shareki *et al.* (2016)
Eulophidae	Eulophinae	*Meruacesa liriomyzae* (Bouček, 1988)	*Meruana liriomyzae*	Mauritius	Bouček (1988)
Eulophidae	Eulophinae	*Meruacesa liriomyzae* (Bouček, 1988)	*Meruana liriomyzae*	Réunion	Bouček (1988)

Family	Sub-family	Full Name	Name in Reference	Country	Reference
Eulophidae	Eulophinae	*Pnigalio minio* (Walker, 1847)	*Pnigalio flavipes*	Canada	McClanahan (1977)
Eulophidae	Eulophinae	*Zagrammosoma americanum* Girault, 1916	*Zagrammosoma americanum*	Canada	McClanahan (1977)
Eulophidae	Eulophinae	*Zagrammosoma americanum* Girault, 1916	*Zagrammosoma americanum*	USA	Chandler (1982)
Eulophidae	Eulophinae	*Zagrammosoma mirum* Girault, 1916	*Zagrammosoma mirum*	USA	Oatman (1959)
Eulophidae	Eulophinae	*Zagrammosoma multilineatum* (Ashmead, 1888)	*Zagrammosoma multilineatum*	USA	Stegmaier (1972)
Eulophidae	Eulophinae	*Zagrammosoma variegatum* (Nasi, 1907)	*Zagrammosoma variegatum*	Iran	Hesami *et al.* (2006)
Pteromalidae	Miscogastrinae	*Halticoptera aenea* (Walker, 1833)	*Halticoptera aenea*	USA	Oatman (1959); Palumbo *et al.* (1994)
Pteromalidae	Miscogastrinae	*Halticoptera arduine* (Walker, 1833)	*Halticoptera arduine*	Peru	Mujica & Kroschel (2011)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	Guatemala	Rodríguez-Castañeda *et al.* (2017)
Pteromalidae	Miscogastrinae	*Halticoptera circulus* (Walker, 1833)	*Halticoptera circulus*	USA	Stegmaier (1972), Oatman & Johnson (1981)
Pteromalidae	Miscogastrinae	*Halticoptera patellana* (Dalman, 1918)	*Halticoptera patellana*	Canada	McClanahan (1977)
Pteromalidae	Miscogastrinae	*Halticoptera patellana* (Dalman, 1918)	*Halticoptera patellana*	USA	Jensen & Koehler (1970), Stegmaier (1972)
Pteromalidae	Pteromalinae	*Cyrtogaster vulgaris* Walker, 1833	*Cyrtogaster vulgaris*	Iran	Lotfalizadeh & Gharali (2008)
Figitidae	Eucoilinae	*Banacuniculus utilis* (Beardsley, 1988)	*Ganaspidium hunteri*	USA	Johnson (1987)
Figitidae	Eucoilinae	*Ganaspidium pusillae* Weld, 1955	*Ganaspidium pusillae*	USA	Harding (1965)
Figitidae	Eucoilinae	*Gronotoma adachiae* Beardsley, 1988	*Gronotoma adachiae*	Vietnam	Abe & Konishi (2012)
Figitidae	Eucoilinae	*Gronotoma micromorpha* (Perkins, 1910)	*Gronotoma micromorpha*	Indonesia	Herlinadewi et al. (2013)
Figitidae	Eucoilinae	*Sinatra pacifica* (Yoshimoto, 1962)	*Cothonaspis pacifica*	USA	Johnson (1987)
Braconidae	Opiinae	*Desmiostoma parvulum* (Wesmael 1835)	*Desmiostoma parvulum*	USA	Marsh (1979
Braconidae	Opiinae	*Oenongastra microrhopalae* (Ashmead, 1896)	*Oenongastra microrhopalae*	Canada	McClanahan (1977)
Braconidae	Opiinae	*Opius aridis* Gahan 1913	*Opius aridis*	USA	Harding (1965)
Braconidae	Opiinae	*Opius bruneipes* Gahan, 1913	*Opius bruneipes*	USA	Harding (1965)
Braconidae	Opiinae	*Opius caricivorae* Fischer, 1964	*Opius caricivorae*	China	Chen *et al.* (2003)
Braconidae	Opiinae	*Opius caricivorae* Fischer, 1964	*Opius caricivorae*	Taiwan	Chien & Chang (2012)
Braconidae	Opiinae	*Opius chromatomyiae* Belokobylskij & Wharton, 2004	*Opius chromatomyiae*	Indonesia	Wayhuni *et al.* (2017)
Braconidae	Opiinae	*Opius chromatomyiae* Belokobylskij & Wharton, 2004	*Opius chromatomyiae*	Vietnam	Tran (2009)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	Canada	McClanahan (1975, 1977)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	China	Xu *et al.* (1999)
Braconidae	Opiinae	*Opius dimidiatus* (Ashmead, 1889)	*Opius dimidiatus*	USA	Harding (1965); Lema & Poe (1979)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	China	Xu *et al.* (1999)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	Guatemala	Rodríguez-Castañeda *et al.* (2017)
Braconidae	Opiinae	*Opius dissitus* Muesebeck, 1963	*Opius dissitus*	USA	Stegmaier (1972), Johnson (1987)
Braconidae	Opiinae	*Opius liriomyzae* Fischer, 1964	*Opius liriomyzae*	USA	Marsh (1979)
Braconidae	Opiinae	*Phaedrotoma biroi* (Fischer, 1960)	*Phaedrotoma biroi*	China	Xing *et al.* (2017)
Braconidae	Opiinae	*Phaedrotoma scabriventris* (Nixon, 1955)	*Opius (Gastrosema) scabriventris*	Brazil	Costa-Lima *et al.* (2014)
Braconidae	Opiinae	*Phaedrotoma suturalis* (Gahan, 1913)	*Opius suturalis*	USA	Oatman (1959); Harding (1965)

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Appendix 7.4 Overlap of parasitoid species recorded from the 3 *Liriomyza* spp.

(a) parasitoid species recorded from all 3 *Liriomyza* spp.

Family	Sub-Family	Species	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Braconidae	Opiinae	*Opius dimidiatus*	1	1	1
Braconidae	Opiinae	*Opius dissitus*	1	1	1
Eulophidae	Endedoninae	*Asecodes delucchii*	1	1	1
Eulophidae	Endedoninae	*Chrysocharis ainsliei*	1	1	1
Eulophidae	Endedoninae	*Chrysocharis caribea*	1	1	1
Eulophidae	Endedoninae	*Chrysocharis oscinidis*	1	1	1
Eulophidae	Endedoninae	*Chrysocharis pentheus*	1	1	1
Eulophidae	Endedoninae	*Closterocerus cinctipennis*	1	1	1
Eulophidae	Endedoninae	*Neochrysocharis diastatae*	1	1	1
Eulophidae	Endedoninae	*Neochrysocharis formosa*	1	1	1
Eulophidae	Endedoninae	*Neochrysocharis okazakii*	1	1	1
Eulophidae	Eulophinae	*Burkseus vittatus*	1	1	1
Eulophidae	Eulophinae	*Cirrospilus brevicorpus*	1	1	1
Eulophidae	Eulophinae	*Diaulinopsis callichroma*	1	1	1
Eulophidae	Eulophinae	*Diglyphus albiscapus*	1	1	1
Eulophidae	Eulophinae	*Diglyphus begini*	1	1	1
Eulophidae	Eulophinae	*Diglyphus crassinervis*	1	1	1
Eulophidae	Eulophinae	*Diglyphus intermedius*	1	1	1
Eulophidae	Eulophinae	*Diglyphus isaea*	1	1	1
Eulophidae	Eulophinae	*Diglyphus minoeus*	1	1	1
Eulophidae	Eulophinae	*Diglyphus pulchripes*	1	1	1
Eulophidae	Eulophinae	*Hemiptarsenus varicornis*	1	1	1
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi*	1	1	1
Figitidae	Eucoilinae	*Gronotoma micromorpha*	1	1	1
Figitidae	Eucoilinae	*Sinatra pacifica*	1	1	1
Pteromalidae	Miscogastrinae	*Halticoptera circulus*	1	1	1
Pteromalidae	Miscogastrinae	*Halticoptera patellana*	1	1	1
∑			**27**	**27**	**27**

(b) parasitoid species recorded from *L. huidobrensis* and *L. trifolii*, but not *L. sativae*

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Braconidae	Alysiinae	*Dacnusa sasakawai*	1	1	
Braconidae	Opiinae	*Opius pallipes*	1	1	
Eulophidae	Endedoninae	*Chrysocharis orbicularis*	1	1	
Eulophidae	Endedoninae	*Chrysocharis pubicornis*	1	1	
Eulophidae	Endedoninae	*Pediobius metallicus*	1	1	
Eulophidae	Eulophinae	*Diglyphus pachyneurus*	1	1	
Eulophidae	Eulophinae	*Hemiptarsenus ornatus*	1	1	
Eulophidae	Eulophinae	*Pnigalio incompletus*	1	1	
Eulophidae	Eulophinae	*Pnigalio katonis*	1	1	
Eulophidae	Eulophinae	*Pnigalio soemius*	1	1	
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae*	1	1	
∑			**11**	**11**	

(c) parasitoid species recorded from *L. huidobrensis* and *L. sativae*, but not *L. trifolii*

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Braconidae	Opiinae	*Opius caricivorae*	1		1
Braconidae	Opiinae	*Opius chromatomyiae*	1		1
Braconidae	Opiinae	*Phaedrotoma scabriventris*	1		1
Eulophidae	Endedoninae	*Chrysocharis flacilla*	1		1
Eulophidae	Endedoninae	*Chrysocharis vonones*	1		1
Eulophidae	Endedoninae	*Neochrysocharis beasleyi*	1		1
Eulophidae	Endedoninae	*Proacrias thysanoides*	1		1
Eulophidae	Eulophinae	*Diglyphus wani*	1		1
Eulophidae	Eulophinae	*Diglyphus websteri*	1		1
Eulophidae	Eulophinae	*Zagrammosoma multilineatum*	1		1
Figitidae	Eucoilinae	*Gronotoma adachiae*	1		1
Pteromalidae	Miscogastrinae	*Halticoptera arduine*	1		1
∑			**12**		**12**

(d) parasitoid species recorded from *L. trifolii* and *L. sativae*, but not *L. huidobrensis*

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***
Braconidae	Opiinae	*Opius bruneipes*		1	1
Braconidae	Opiinae	*Phaedrotoma biroi*		1	1
Eulophidae	Endedoninae	*Asecodes erxias*		1	1
Eulophidae	Endedoninae	*Closterocerus purpureus*		1	1
Eulophidae	Endedoninae	*Closterocerus utahensis*		1	1
Eulophidae	Endedoninae	*Neochrysocharis agromyzae*		1	1
Eulophidae	Eulophinae	*Diaulinopsis arenaria*		1	1
Eulophidae	Eulophinae	*Diglyphus chabrias*		1	1
Eulophidae	Eulophinae	*Diglyphus pusztensis*		1	1
Eulophidae	Eulophinae	*Meruacesa liriomyzae*		1	1
Eulophidae	Eulophinae	*Zagrammosoma americanum*		1	1
Eulophidae	Eulophinae	*Zagrammosoma variegatum*		1	1
Figitidae	Eucoilinae	*Banacuniculus utilis*		1	1
Figitidae	Eucoilinae	*Ganaspidium pusillae*		1	1
Pteromalidae	Miscogastrinae	*Halticoptera aenea*		1	1
Pteromalidae	Pteromalinae	*Cyrtogaster vulgaris*		1	1
∑				**16**	**16**

(e) parasitoid species recorded only from *L. huidobrensis*

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***
Braconidae	Alysiinae	*Dacnusa sibirica*	1		
Braconidae	Braconinae	*Bracon intercessor*	1		
Braconidae	Opiinae	*Opius mandibularis*	1		
Braconidae	Opiinae	*Opius meracus*	1		
Braconidae	Opiinae	*Phaedrotoma luteoclypealis*	1		
Braconidae	Opiinae	*Phaedrotoma mesoclypealis*	1		
Eulophidae	Endedoninae	*Chrysocharis bedius*	1		
Eulophidae	Endedoninae	*Chrysocharis ignota*	1		
Eulophidae	Endedoninae	*Chrysocharis tristis*	1		

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Eulophidae	Endedoninae	*Closterocerus pulcher*	1		
Eulophidae	Endedoninae	*Proacrias xenodice*	1		
Eulophidae	Eulophinae	*Diaulinopsis sp.*	1		
Eulophidae	Eulophinae	*Diglyphus bimaculatus*	1		
Eulophidae	Eulophinae	*Diglyphus pedicellus*	1		
Eulophidae	Eulophinae	*Diglyphus poppoea*	1		
Eulophidae	Eulophinae	*Hemiptarsenus fulvicollis*	1		
Eulophidae	Eulophinae	*Hemiptarsenus unguicellus*	1		
Eulophidae	Eulophinae	*Zagrammosoma latilineatum*	1		
Figitidae	Eucoilinae	*Zaeucoila clavatus*	1		
Pteromalidae	Miscogastrinae	*Halticoptera helioponi*	1		
Pteromalidae	Miscogastrinae	*Halticoptera peruviana*	1		
Pteromalidae	Miscogastrinae	*Thinodytes cyzicus*	1		
Pteromalidae	Pteromalinae	*Notoglyptus tzeltales*	1		
Tetracampidae	Platynocheilinae	*Platynocheilus cuprifrons*	1		
∑			**24**		

(f) parasitoid species recorded only from *L. trifolii*

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Braconidae	Alysiinae	*Dacnusa nipponica*		1	
Braconidae	Alysiinae	*Pseudopezomachus masii*		1	
Braconidae	Opiinae	*Opius ambiguus*		1	
Braconidae	Opiinae	*Opius basalis*		1	
Braconidae	Opiinae	*Opius exigua*		1	
Braconidae	Opiinae	*Opius gafsaensis*		1	
Braconidae	Opiinae	*Opius osogovoensis*		1	
Eulophidae	Endedoninae	*Apleurotropis kumatai*		1	
Eulophidae	Endedoninae	*Chrysocharis liriomyzae*		1	
Eulophidae	Endedoninae	*Chrysonotomyia rexia*		1	
Eulophidae	Endedoninae	*Chrysonotomyia ricini*		1	
Eulophidae	Endedoninae	*Chrysonotomyia smaragdulus*		1	

Family	Sub-Family	*Species*	*Liriomyza huidobrensis*	*Liriomyza trifolii*	*Liriomyza sativae*
Eulophidae	Endedoninae	*Closterocerus agromyzae*		1	
Eulophidae	Endedoninae	*Neochrysocharis ambitiosus*		1	
Eulophidae	Endedoninae	*Neochrysocharis arvensis*		1	
Eulophidae	Endedoninae	*Neochrysocharis chlorogaster*		1	
Eulophidae	Endedoninae	*Neochrysocharis indicus*		1	
Eulophidae	Endedoninae	*Neochrysocharis pictipes*		1	
Eulophidae	Endedoninae	*Neochrysocharis sericea*		1	
Eulophidae	Eulophinae	*Diglyphus horticola*		1	
Eulophidae	Eulophinae	*Pnigalio cristatus*		1	
Eulophidae	Eulophinae	*Sympiesis acalle*		1	
Eulophidae	Eulophinae	*Sympiesis gordius*		1	
Eulophidae	Eulophinae	*Zagrammosoma lineaticeps*		1	
Eulophidae	Eulophinae	*Zagrammosoma talitzkii*		1	
Eulophidae	Tetrastichinae	*Baryscapus impeditus*		1	
Eulophidae	Tetrastichinae	*Neotrichoporoides szelenyii*		1	
Eulophidae	Tetrastichinae	*Quadrastichus plaquoi*		1	
Figitidae	Eucoilinae	*Banacuniculus nigrimanus*		1	
Figitidae	Eucoilinae	*Gronotoma fetura*		1	
Figitidae	Eucoilinae	*Gronotoma guamensis*		1	
Figitidae	Eucoilinae	*Nordlanderia plowa*		1	
Figitidae	Eucoilinae	*Zaeucoila robusta*		1	
Pteromalidae	Miscogastrinae	*Halticoptera longipetiolus*		1	
Pteromalidae	Pteromalinae	*Sphegigaster brevicornis*		1	
Pteromalidae	Pteromalinae	*Sphegigaster hamugurivora*		1	
Pteromalidae	Pteromalinae	*Trichomalopsis oryzae*		1	
Tetracampidae	Tetracampinae	*Epiclerus nomocerus*		1	
∑				**38**	

(g) parasitoid species recorded only from *L. sativae*

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***
Braconidae	Opiinae	*Desmiostoma parvulum*			1
Braconidae	Opiinae	*Oenongastra microrhopalae*			1
Braconidae	Opiinae	*Opius aridis*			1
Braconidae	Opiinae	*Opius liriomyzae*			1
Braconidae	Opiinae	*Phaedrotoma suturalis*			1
Eulophidae	Endedoninae	*Achrysocharoides zwoelferi*			1
Eulophidae	Endedoninae	*Chrysocharis crassiscapus*			1
Eulophidae	Endedoninae	*Chrysocharis giraulti*			1
Eulophidae	Endedoninae	*Chrysonotomyia reticulata*			1
Eulophidae	Endedoninae	*Closterocerus cincinnatus*			1
Eulophidae	Endedoninae	*Closterocerus trifasciatus*			1
Eulophidae	Endedoninae	*Neochrysocharis arizonensis*			1
Eulophidae	Eulophinae	*Burkseus flavoviridis*			1
Eulophidae	Eulophinae	*Diglyphus sensilis*			1
Eulophidae	Eulophinae	*Hemiptarsenus waileseIIae*			1
Eulophidae	Eulophinae	*Pnigalio minio*			1
Eulophidae	Eulophinae	*Zagrammosoma mirum*			1
∑					**17**

Appendix 7.5 Parasitoid species by country records in descending order (pooling all records for a particular country for each *Liriomyza* sp.)

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Eulophinae	*Diglyphus isaea*	15	12	5	32
Eulophidae	Endedoninae	*Neochrysocharis formosa*	7	16	7	30
Eulophidae	Eulophinae	*Hemiptarsenus varicornis*	6	11	9	26
Braconidae	Opiinae	*Opius* sp.	13	6	1	20
Eulophidae	Endedoninae	*Chrysocharis pentheus*	5	5	5	15
Eulophidae	Eulophinae	*Diglyphus crassinervis*	5	6	2	13
Braconidae	Opiinae	*Opius dissitus*	4	5	3	12
Pteromalidae	Miscogastrinae	*Halticoptera circulus*	5	5	2	12
Eulophidae	Endedoninae	*Neochrysocharis okazakii*	3	5	3	11
Eulophidae	Endedoninae	*Chrysocharis* sp.	6	4		10
Braconidae	Opiinae	*Opius dimidiatus*	2	4	3	9
Eulophidae	Eulophinae	*Hemiptarsenus zilahisebessi*	3	5	1	9
Eulophidae	Eulophinae	*Diglyphus begini*	4	2	3	9
Eulophidae	Endedoninae	*Chrysocharis caribea*	2	2	4	8
Eulophidae	Endedoninae	*Chrysocharis oscinidis*	1	4	3	8
Eulophidae	Eulophinae	*Diglyphus* sp.	6	2		8
Pteromalidae	Miscogastrinae	*Halticoptera* sp.	7	1		8
Eulophidae	Endedoninae	*Asecodes delucchii*	2	2	3	7
Eulophidae	Eulophinae	*Diglyphus intermedius*	4	2	1	7
Eulophidae	Eulophinae	*Burkseus vittatus*	2	4	1	7
Braconidae	Alysiinae	*Dacnusa sibirica*	6			6
Eulophidae	Eulophinae	*Diglyphus minoeus*	3	2	1	6
Eulophidae	Eulophinae	*Diglyphus websteri*	3		3	6
Eulophidae	Endedoninae	*Neochrysocharis diastatae*	1	2	2	5
Eulophidae	Endedoninae	*Closterocerus cinctipennis*	1	3	1	5

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Endedoninae	*Neochrysocharis* sp.	1	4		5
Eulophidae	Eulophinae	*Diaulinopsis callichroma*	1	2	2	5
Figitidae	Eucoilinae	*Banacuniculus utilis*		4	1	5
Figitidae	Eucoilinae	*Sinatra pacifica*	1	3	1	5
Braconidae	Opiinae	*Phaedrotoma scabriventris*	3		1	4
Braconidae	Opiinae	*Opius pallipes*	2	2		4
Eulophidae	Endedoninae	*Pediobius metallicus*	3	1		4
Eulophidae	Endedoninae	*Chrysocharis pubicornis*	3	1		4
Eulophidae	Endedoninae	*Chrysonotomyia* sp.	2	2		4
Eulophidae	Endedoninae	*Chrysocharis vonones*	2		2	4
Eulophidae	Endedoninae	*Chrysocharis flacilla*	3		1	4
Eulophidae	Eulophinae	*Pnigalio* sp.	2	2		4
Eulophidae	Eulophinae	*Pnigalio katonis*	2	2		4
Eulophidae	Eulophinae	*Diglyphus pulchripes*	1	2	1	4
Eulophidae	Eulophinae	*Zagrammosoma* sp.	2	2		4
Eulophidae	Eulophinae	*Cirrospilus brevicorpus*	1	1	2	4
Eulophidae	Eulophinae	*Diglyphus albiscapus*	1	2	1	4
Eulophidae	Tetrastichinae	*Quadrastichus liriomyzae*	1	3		4
Eulophidae	Tetrastichinae	*Quadrastichus* sp.	3	1		4
Figitidae	Eucoilinae	*Gronotoma micromorpha*	1	2	1	4
Pteromalidae	Miscogastrinae	*Halticoptera patellana*	1	1	2	4
Braconidae	Opiinae	*Opius caricivorae*	1		2	3
Braconidae	Opiinae	*Opius chromatomyiae*	1		2	3
Eulophidae	Endedoninae	*Asecodes erxias*		2	1	3
Eulophidae	Endedoninae	*Chrysocharis bedius*	3			3
Eulophidae	Endedoninae	*Closterocerus* sp.	3			3
Eulophidae	Endedoninae	*Chrysocharis ainsliei*	1	1	1	3

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Endedoninae	*Proacrias thysanoides*	2		1	3
Eulophidae	Endedoninae	*Neochrysocharis beasleyi*	2		1	3
Eulophidae	Eulophinae	*Hemiptarsenus* sp.	2	1		3
Eulophidae	Eulophinae	*Zagrammosoma americanum*		1	2	3
Eulophidae	Eulophinae	*Meruacesa liriomyzae*		1	2	3
Eulophidae	Eulophinae	*Diglyphus chabrias*		2	1	3
Eulophidae	Eulophinae	*Hemiptarsenus ornatus*	1	2		3
Eulophidae	Eulophinae	*Diaulinopsis arenaria*		2	1	3
Figitidae	Eucoilinae	*Gronotoma* sp.	3			3
Pteromalidae	Miscogastrinae	*Halticoptera arduine*	2		1	3
Pteromalidae	unidentified	Pteromalidae species	3			3
Braconidae	Alysiinae	*Dacnusa* sp.	2			2
Braconidae	Alysiinae	*Dacnusa sasakawai*	1	1		2
Braconidae	Opiinae	*Phaedrotoma biroi*		1	1	2
Braconidae	Opiinae	*Opius bruneipes*		1	1	2
Braconidae	Opiinae	*Opius basalis*		2		2
Braconidae	Opiinae	*Opius exigua*		2		2
Eulophidae	Endedoninae	*Closterocerus utahensis*		1	1	2
Eulophidae	Endedoninae	*Neochrysocharis agromyzae*		1	1	2
Eulophidae	Endedoninae	*Proacrias xenodice*	2			2
Eulophidae	Endedoninae	*Asecodes* sp.	1	1		2
Eulophidae	Endedoninae	*Chrysocharis orbicularis*	1	1		2
Eulophidae	Endedoninae	*Closterocerus purpureus*		1	1	2
Eulophidae	Eulophinae	*Pnigalio soemius*	1	1		2
Eulophidae	Eulophinae	*Zagrammosoma multilineatum*	1		1	2
Eulophidae	Eulophinae	*Diaulinopsis* sp.	2			2
Eulophidae	Eulophinae	*Diglyphus pachyneurus*	1	1		2

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Eulophinae	*Pnigalio incompletus*	1	1		2
Eulophidae	Eulophinae	*Diglyphus wani*	1		1	2
Eulophidae	Eulophinae	*Diglyphus pusztensis*		1	1	2
Eulophidae	Eulophinae	*Zagrammosoma variegatum*		1	1	2
Eulophidae	Eulophinae	*Cirrospilus* sp. near *cinctiventris*		2		2
Figitidae	Eucoilinae	*Banacuniculus nigrimanus*		2		2
Figitidae	Eucoilinae	*Ganaspidium pusillae*		1	1	2
Figitidae	Eucoilinae	*Gronotoma adachiae*	1		1	2
Figitidae	Eucoilinae	*Ganaspidium* sp.	1	1		2
Pteromalidae	Miscogastrinae	*Halticoptera aenea*		1	1	2
Pteromalidae	Pteromalinae	*Sphegigaster* sp.	1	1		2
Pteromalidae	Pteromalinae	*Cyrtogaster vulgaris*		1	1	2
Braconidae	Alysiinae	*Dacnusa nipponica*		1		1
Braconidae	Alysiinae	*Pseudopezomachus masii*		1		1
Braconidae	Alysiinae	*Oenonogastra* sp.	1			1
Braconidae	Braconinae	*Bracon intercessor*	1			1
Braconidae	Opiinae	*Oenongastra microrhopalae*			1	1
Braconidae	Opiinae	*Phaedrotoma luteoclypealis*	1			1
Braconidae	Opiinae	*Opius* sp. 2		1		1
Braconidae	Opiinae	*Opius ambiguus*		1		1
Braconidae	Opiinae	*Opius aridis*			1	1
Braconidae	Opiinae	*Phaedrotoma suturalis*			1	1
Braconidae	Opiinae	*Opius* sp. 1		1		1
Braconidae	Opiinae	*Opius meracus*	1			1
Braconidae	Opiinae	*Opius thoracosema* sp. *3*		1		1
Braconidae	Opiinae	*Opius* near *brownsvillensis*		1		1
Braconidae	Opiinae	*Opius osogovoensis*		1		1

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Braconidae	Opiinae	*Phaedrotoma mesoclypealis*	1			1
Braconidae	Opiinae	*Opius gafsaensis*		1		1
Braconidae	Opiinae	*Phaedrotoma* sp.	1			1
Braconidae	Opiinae	*Opius liriomyzae*			1	1
Braconidae	Opiinae	*Desmiostoma parvulum*			1	1
Braconidae	Opiinae	*Opius mandibularis*	1			1
Braconidae	Tersilochinae	*Allophrys* sp.		1		1
Diapriidae	Diapriinae	*Trichopria* sp.	1			1
Eulophidae	Endedoninae	*Neochrysocharis indicus*		1		1
Eulophidae	Endedoninae	*Chrysonotomyia smaragdulus*		1		1
Eulophidae	Endedoninae	*Closterocerus agromyzae*		1		1
Eulophidae	Endedoninae	*Chrysocharis tristis*	1			1
Eulophidae	Endedoninae	*Pediobius* sp.		1		1
Eulophidae	Endedoninae	*Chrysocharis* near *aluta*	1			1
Eulophidae	Endedoninae	*Apleurotropis kumatai*		1		1
Eulophidae	Endedoninae	*Closterocerus pulcher*	1			1
Eulophidae	Endedoninae	*Neochrysocharis pictipes*		1		1
Eulophidae	Endedoninae	*Chrysocharis liriomyzae*		1		1
Eulophidae	Endedoninae	*Chrysonotomyia* sp.*F*		1		1
Eulophidae	Endedoninae	*Chrysonotomyia rexia*		1		1
Eulophidae	Endedoninae	*Neochrysocharis chlorogaster*		1		1
Eulophidae	Endedoninae	*Closterocerus trifasciatus*			1	1
Eulophidae	Endedoninae	*Chrysocharis crassiscapus*			1	1
Eulophidae	Endedoninae	*Chrysonotomyia ricini*		1		1
Eulophidae	Endedoninae	*Chrysonotomyia reticulata*			1	1
Eulophidae	Endedoninae	*Neochrysocharis sericea*		1		1
Eulophidae	Endedoninae	*Closterocerus cincinnatus*			1	1

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Endedoninae	*Chrysocharis giraulti*			1	1
Eulophidae	Endedoninae	*Chrysocharis ignota*	1			1
Eulophidae	Endedoninae	*Neochrysocharis ambitiosus*		1		1
Eulophidae	Endedoninae	*Proacrias* sp.	1			1
Eulophidae	Endedoninae	*Neochrysocharis arizonensis*			1	1
Eulophidae	Endedoninae	*Neochrysocharis arvensis*		1		1
Eulophidae	Endedoninae	*Achrysocharoides zwoelferi*			1	1
Eulophidae	Endedoninae	*Chrysonotomyia* sp. near *leptocera*		1		1
Eulophidae	Eulophinae	*Sympiesis* sp.		1		1
Eulophidae	Eulophinae	*Diglyphus horticola*		1		1
Eulophidae	Eulophinae	*Zagrammosoma mirum*			1	1
Eulophidae	Eulophinae	*Burkseus flavoviridis*			1	1
Eulophidae	Eulophinae	*Sympiesis acalle*		1		1
Eulophidae	Eulophinae	*Hemiptarsenus waileselIae*			1	1
Eulophidae	Eulophinae	*Zagrammosoma latilineatum*	1			1
Eulophidae	Eulophinae	*Zagrammosoma talitzkii*		1		1
Eulophidae	Eulophinae	*Diglyphus pedicellus*	1			1
Eulophidae	Eulophinae	*Diglyphus bimaculatus*	1			1
Eulophidae	Eulophinae	*Pnigalio* sp near *pectinicornis*		1		1
Eulophidae	Eulophinae	*Elachertus* sp. L		1		1
Eulophidae	Eulophinae	*Sympiesis gordius*		1		1
Eulophidae	Eulophinae	*Diglyphus sensilis*			1	1
Eulophidae	Eulophinae	*Diglyphus poppoea*	1			1
Eulophidae	Eulophinae	*Hemiptarsenus fulvicollis*	1			1
Eulophidae	Eulophinae	*Zagrammosoma lineaticeps*		1		1
Eulophidae	Eulophinae	*Cirrospilus* sp.		1		1
Eulophidae	Eulophinae	*Diglyphus* sp. *(near intermedius)*	1			1

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Eulophidae	Eulophinae	*Pnigalio minio*			1	1
Eulophidae	Eulophinae	*Meruacesa* sp.	1			1
Eulophidae	Eulophinae	*Pnigalio cristatus*		1		1
Eulophidae	Eulophinae	*Cirrospilus variegatus group*		1		1
Eulophidae	Eulophinae	*Hemiptarsenus unguicellus*	1			1
Eulophidae	Tetrastichinae	*Quadrastichus plaquoi*		1		1
Eulophidae	Tetrastichinae	*Baryscapus impeditus*		1		1
Eulophidae	Tetrastichinae	*Aprostocetus* sp.		1		1
Eulophidae	Tetrastichinae	*Neotrichoporoides szelenyii*		1		1
Eulophidae	Tetrastichinae	*Oomyzus* sp.		1		1
Figitidae	Eucoilinae	*Alloxysta* sp.	1			1
Figitidae	Eucoilinae	*Gronotoma guamensis*		1		1
Figitidae	Eucoilinae	*Nordlanderia plowa*		1		1
Figitidae	Eucoilinae	*Zaeucoila robusta*		1		1
Figitidae	Eucoilinae	*Tribliographa* sp.	1			1
Figitidae	Eucoilinae	*Gronotoma fetura*		1		1
Figitidae	Eucoilinae	*Moneucoela* sp.	1			1
Figitidae	Eucoilinae	*Zaeucolia* sp.	1			1
Figitidae	Eucoilinae	*Kleidotoma* sp.		1		1
Figitidae	Eucoilinae	*Zaeucoila clavatus*	1			1
Pteromalidae	Herbertiinae	*Herbertia indica*		1		1
Pteromalidae	Miscogastrinae	*Halticoptera* sp. *1*		1		1
Pteromalidae	Miscogastrinae	*Thinodytes cyzicus*	1			1
Pteromalidae	Miscogastrinae	*Halticoptera* sp. *2*		1		1
Pteromalidae	Miscogastrinae	*Thinodytes* sp.	1			1
Pteromalidae	Miscogastrinae	*Halticoptera nr. circulus*		1		1
Pteromalidae	Miscogastrinae	*Halticoptera longipetiolus*		1		1

Family	**Sub-Family**	***Species***	***Liriomyza huidobrensis***	***Liriomyza trifolii***	***Liriomyza sativae***	**∑**
Pteromalidae	Miscogastrinae	*Halticoptera peruviana*	1			1
Pteromalidae	Miscogastrinae	*Halticoptera helioponi*	1			1
Pteromalidae	Pteromalinae	*Heteroschema* sp.	1			1
Pteromalidae	Pteromalinae	*Notoglyptus tzeltales*	1			1
Pteromalidae	Pteromalinae	*Trichomalopsis oryzae*		1		1
Pteromalidae	Pteromalinae	*Trichomalopsis* sp.	1			1
Pteromalidae	Pteromalinae	*Sphegigaster hamugurivora*		1		1
Pteromalidae	Pteromalinae	*Sphegigaster brevicornis*		1		1
Tetracampidae	Platynocheilinae	*Platynocheilus cuprifrons*	1			1
Tetracampidae	Tetracampinae	*Epiclerus* sp.	1			1
Tetracampidae	Tetracampinae	*Epiclerus nomocerus*		1		1
		Grand Total	228	240	123	591

Appendix 7.6 Grenouillet C, Martinez M & Rasplus JY. 1993. Liste des parasitoïdes et des prédateurs des Liriomyza d'importance economique dans le monde (Diptera: Agromyzidae). Colloque sur les mouches mineuses des plantes cultivees, 24-26 Mar 1993, Montpellier, France, 143-156. [original and translation]

"LIRIOMYZA" - COLLOQUE SUR LES MOUCHES MINEUSES DES PLANTES CULTIVEES MONTPELLIER - 24, 25, 26 MARS 1993

LISTE DES PARASITOIDES ET DES PREDATEURS DES *LIRIOMYZA* D'IMPORTANCE ECONOMIQUE DANS LE MONDE (DIPTERA : AGROMYZIDAE)

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RESUME

Une liste mondiale de toutes les espèces de parastoïdes et de prédateurs des 26 espèces de *Liriomyza* d'importance économique est donnée. Les noms d'espèces des *Liriomyza*, des prédateurs et des parasitoïdes ont été corrigés en fonction des publications récentes concernant la systématique de ces groupes. La liste est présentée sous forme d'un tableau donnant pour chaque parasitoïde et prédateur, l'espèce de *Liriomyza* attaquée et la région biogéographique dans laquelle la donnée a été colligée. Un nombre total de 137 espèces de parasitoïdes (exclusivement Hymenoptera) et 5 espèces de prédateurs (4 Diptera et 1 Hemiptera) ont été recensées dans la littérature.

MOTS-CLES : *Liriomyza*, ravageurs, parasitoïdes, prédateurs, monde

SUMMARY

A world list of all known parasites and predators of the 26 economically important species of *Liriomyza* is given. Species names of *Liriomyza*, predators and parasitoids have been corrected in light of recent papers on the systematics of these groups. The list is presentated in a table-form, giving for each parasitoids and predators, the species of *Liriomyza* attacked and the biogeographical area where the record have been done. A total number of 137 species of parasitoids (exclusively Hymenoptera) and 5 species of predators (4 Diptera and 1 Hemiptera) have been recorded from the literature.

KEY-WORDS : *Liriomyza*, pests, parasitoids, predators, world

INTRODUCTION

Parmi les 339 espèces appartenant au genre *Liriomyza*, 26 espèces sont considérées comme d'importance économique en raison des dégâts qu'elles peuvent occasionner aux plantes cultivées (cf. publication de M. Martinez dans les actes de ce Colloque). Ces Agromyzides, comme la plupart des espèces d'insectes, sont les hôtes d'un certain nombre de parasitoïdes, esssentiellement des Hyménoptères qui se développent en consommant les tissus de la larve ou de la pupe. A coté de ces Hyménoptères parasites, quelques espèces de prédateurs sont connues pour attaquer, au moins occasionnellement, des *Liriomyza* et à ce titre peuvent être considérées comme de potentiels agents régulateurs des populations de ces déprédateurs.

Au cours des 15 dernières années, la lutte chimique contre certains de ces ravageurs des cultures a induit l'apparition de populations résistantes aux pesticides et de ce fait favorisé le développement des recherches sur les ennemis naturels de ces *Liriomyza*. De nombreuses publications mentionnant des parasitoïdes de ces Agromyzides ont été rédigées. Nous avons voulu faire une synthèse des ces informations et ainsi donner une liste aussi exhaustive que possible des espèces de parasitoïdes et de prédateurs connues pour attaquer dans le monde une ou plusieurs de ces 26 espèces de *Liriomyza* d'importance agronomique. Pour cela, nous avons recherché, dans la bibliographie, tous les articles citant des parasitoïdes et des prédateurs de ces Diptères. Plus de 50 ans de données bibliographiques ont ainsi été analysées. Nous avons revu l'ensemble des publications traitant de parasitoïdes d'Agromyzidae, ainsi que la plus grande partie des publications de taxonomie relatives aux espèces connues ou susceptibles d'être parasites de *Liriomyza*. Nous présentons le résultat de notre recherche bibliographique dans les sept tableaux ci-dessous. Les 6 premiers tableaux traitent des 13 espèces de *Liriomyza* les plus couramment citées et par conséquent les plus étudiées; le dernier tableau donnent les parasites pour 9 autres espèces beaucoup moins étudiées. Aux données publiées, nous avons rajoutées quelques données originales en particulier en ce qui concernent *L. huidobrensis*, qui en France est parasitée par *Dacnusa sibirica*, *Diglyphus isaea* et *Hemiptarsenus dropion* (C. Trouvé comm. pers.).

Plusieurs problèmes se sont posés à nous lors de l'élaboration de cette liste. En effet, la systématique des *Liriomyza* est particulièrement difficile et la plupart des espèces ont parfois été confondues et/ou souvent mal identifiées (cf. Martinez opp. cit.). Par exemple, les citations de parasites de *L. trifolii* en Europe avant 1975 concernent en réalité *L. congesta*, de même les citations de *L.strigata* dans la région afrotropicale, de *L. bryoniae* et de *L. flaveola* en région néotropicale sont erronées et correspondent en fait à d'autres espèces. Enfin certains auteurs ont mal nommé les espèces qu'ils ont étudié, ainsi les travaux d'Oatman sur *L. pictella* (Thomson) concernent en réalité *L. sativae*.

Il en est de même pour les parasitoïdes d'Agromyzidae qui appartiennnent à des genres de Braconidae ou de Chalcidoidea encore imparfaitement connus et particulièrement difficiles. En raison de cette méconnaissance taxinomique des *Liriomyza* et de leurs parasites, la littérature est encombrée d'erreurs d'identifications parfois difficiles à repérer et souvent transmises dans différents écrits et catalogues.

L'étude systématique de nombreux groupes de parasitoïdes se poursuivant toujours, la nomenclature des espèces citées comme parasites de *Liriomyza* a été profondément modifiée au cours des dernières années. Il en résulte qu'un certain nombre d'espèces couramment citées dans la littérature ne se retrouvent pas dans les tableaux. Dans nos tableaux, ces espèces sont citées sous le nom qui doit dorénavant être utilisé. Afin d'informer le lecteur de ces changements nomenclaturaux, nous avons annoté les tableaux et expliqué ces changements dans le paragraphe "Notes sur les citations".

Il ne nous a pas été possible d'indiquer l'ensemble de la bibliographie consultée (plusieurs centaines de titres), et par conséquent nous n'avons indiqué que les références les plus importantes ou celles apportant des données originales.

Au total, c'est 137 espèces de parasitoïdes, représentant 9 familles d'Hyménoptères qui sont citées dans la littérature et seulement 5 espèces de prédateurs (Diptères et Hémiptères). Deux familles de micro-hyménoptères dominent la catégorie des parasitoïdes : les Eulophidae (Chalcidoidea) et les Braconidae (Ichneumonoidea) avec respectivement 51 et 50 espèces citées (Tableau I). Viennent ensuite, les Pteromalidae

(Chalcidoidea) avec 19 espèces recensées et les Eucoilidae (Cynipoidea) avec 12 espèces. Parmi les familles restantes, seule celle des Tetracampidae comprend une espèce parasitant effectivement des *Liriomyza*; les quatres autres familles : Eupelmidae (1 sp.), Mymaridae (1 sp.), Scelionidae (1sp.) et Trichogrammatidae (1sp.) sont à notre avis, citées par erreur comme parasites de *Liriomyza*. (cf. notes). Pour seulement 4 espèces de *Liriomyza* d'importance agronomique aucun parasitoïdes n'est connus.

Il faut signaler que les régions Néarctique (Amérique du Nord) et Ouest-Paléarctique (Europe) sont les mieux étudiées et les cortèges parasitaires des *Liriomyza* vivant dans ces régions sont de loin les plus complets. De nombreuses espèces de parasites restent probablement à découvrir en Afrique, en Amérique du sud et en Asie où peu d'études ont été réalisées. Par ailleurs, il est certain que même en Europe de nouveaux parasites de ces mineuses restent à découvrir.

	FAMILLES HYMENOPTERES				
LISTE DES ESPECES DE *LIRIOMYZA*	Eulo	Brac	Pter	Euco	Tetr
Liriomyza betae (Coquillett, 1900) (A)	2	1	-	-	-
Liriomyza brassicae (Riley, 1885) (P, NE, AF, NO, O, A)	12	2	2	1	-
Liriomyza braziliensis (Frost, 1939) (NO)	-	-	1	-	-
Liriomyza bryoniae (Kaltenbach, 1858) (P)	7	10	3	-	-
Liriomyza caulophaga (Kleinschmidt, 1960) (O, A)	-	-	-	-	-
Liriomyza cepae (Hering, 1927) (P)	-	-	1	1	-
Liriomyza chinensis (Kato, 1949) (P, O)	-	-	-	-	-
Liriomyza cicerina (Rondani, 1856) (P, O)	2	2	-	-	-
Liriomyza clianthi (Watt, 1923) (A)	-	-	-	-	-
Liriomyza congesta (Becker, 1903) (P, O)	8	14	6	-	-
Liriomyza dianthicola (Venturi, 1949) (P, NO)	3	-	1	-	-
Liriomyza flaveola (Fallén, 1823) (P, O)	1	5	-	-	-
Liriomyza huidobrensis (Blanchard, 1926) (P, NE, AF, NO, A)	11	1	6	1	-
Liriomyza marginalis (Malloch, 1913) (NE, NO)	3	-	1	-	-
Liriomyza nietzkei Spencer, 1973 (P)	-	-	1	1	-
Liriomyza orbona (Meigen, 1830) (P)	-	-	-	-	-
Liriomyza pisivora Hering, 1957 (P)	-	2	-	-	-
Liriomyza pusilla (Meigen, 1830) (P, O)	3	6	2	-	-
Liriomyza quadrata (Malloch, 1934) (NO)	-	2	-	-	-
Liriomyza sativae Blanchard, 1938 (P, NE, NO, A)	20	9	2	9	
Liriomyza schmidti (Aldrich, 1929) (NE, NO)	1	-	-	-	-
Liriomyza strigata (Meigen, 1830) (P, O)	9	10	3	-	-
Liriomyza trifoliearum Spencer, 1973 (NE)	18	2	3	-	-
Liriomyza trifolii (Burgess, 1880) (P, NE, AF, NO, O, A)	27	10	3	5	1
Liriomyza urophorina Mik, 1894 (P, O)	-	-	1	-	-
Liriomyza xanthocera (Czerny, 1909) (P)	2	-	2	-	-
NOMBRE TOTAL D'ESPECES DE PARASITES :	51	50	19	12	1

Tableau I. Liste des espèces de *Liriomyza* traitées et nombre de parasites cités pour chacune d'entre-elles. P = région paléarctique ; NE = région néarctique ; AF = région afrotropicale (ou éthiopienne) ; NO = région néotropicale ; O = région orientale ; A = région australienne/océannienne (ces abbréviations sont aussi utilisées dans les autres tableaux). **Eulo** = Eulophidae; **Brac** = Braconidae; **Pter** = Pteromalidae; **Euco** = Eucoilidae; **Tetr** = Tetracampidae.

BRACONIDAE	Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
Aphidius ervi Haliday, 1834 (1)	P		•											
Aphidius matricariae Haliday, 1834 (1)	P		•											
Chorebus aphantus (Marshall, 1895)	P						•							
Chorebus cambricus Griffiths, 1968	P								•					
Chorebus daimenes (Nixon, 1945) (2)	P		•		•				•			•		
Chorebus misellus (Marschall, 1891) (3)	P		•		•							•		•
Chorebus pseudomisellus Griffiths, 1968	P				•									
Chorebus testaceipes Griffiths, 1968	P								•					
Colastes braconius Haliday, 1833	P								•					
Dacnusa adducta Griffiths, 1968	P						•							
Dacnusa areolaris (Nees, 1811) (4)	P											•		
Dacnusa discolor (Foester, 1862)	P											•		
Dacnusa dryas (Nixon, 1948) (5)	Ne												•	
Dacnusa flavicoxa Thomson, 1895	P				•									
Dacnusa hospita (Foester, 1862)	P		•											
Dacnusa maculipes Thomson, 1895 (6)	P		•		•				•			•		•
Dacnusa sibirica Telenga, 1934	P		•											•
Dapsilarthra balteata (Thomson, 1895)	P				•		•							
Dapsilarthra testacea Griffiths, 1968	P								•					
Diaeretiella rapae (M'Intosh, 1855) (1)	P	•?												
Euopius analis (Fischer, 1965)	Ne									•				•
Oenonogastra microrhopalae (Ashmead, 1896)	Ne									•				•
Opius abscissus Thomson, 1895	P													
Opius aemulus Haliday, 1837	P				•									

BRACONIDAE (suite)	Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
Opius aridis Gahan, 1913	Ne									•				
Opius bruneipes Gahan, 1913	Ne									•				•
Opius cinerariae Fischer, 1963	Ne A	•								•				
Opius dimidiatus (Ashmead, 1889)	Ne P		•							•			•	•
Opius dissitus Muesebeck, 1963	Ne P									•				•
Opius exiguus Wesmael, 1835	P											•		
Opius ilicis Nixon, 1939	P				•									
Opius levis Wesmael, 1835	P				•		•					•		
Opius lugens Haliday, 1837	P				•									
Opius mandibularis Gahan, 1915	Ne									•				•
Opius minor Fischer, 1957	P				•									
Opius monilicornis Fischer, 1962	P			•										
Opius pallipes Wesmael, 1835	P		•									•		•
Opius pendulus Haliday, 1837	P				•									
Opius phaseoli Fischer, 1963	O		•											
Opius polyzonius Wesmael, 1835 (7)	P											•		
Opius propodealis Fischer, 1958	P											•		
Opius pygmaeotor (Nees, 1834)	P				•									
Opius pygmaeus Fischer, 1962	P			•										
Opius rex Fischer, 1958	P						•							
Opius scabriventris Nixon, 1955	No							•						
Opius suturalis Gahan, 1913	Ne									•				
Pseudopezomachus cursitans (Ferrière, 1930)	P				•									

EULOPHIIDAE		Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
Chrysocharis ainsliei (Crawford, 1912)		Ne P							•		•			•	•
Chrysocharis caribea Boucek, 1977		No							•		•				
Chrysocharis chilo (Walker, 1839)		P	•										•		
Chrysocharis clarkae Yoshimoto, 1973		H												•	
Chrysocharis crassicapus (Thomson, 1878)	(8)	H	•								•				
Chrysocharis entedonoides (Walker, 1872)	(9)	H											•		
Chrysocharis flacilla (Walker, 1842)	(10)	No							•						
Chrysocharis giraulti Yoshimoto, 1973	(11)	Ne	•											•	
Chrysocharis liriomyzae Delucchi, 1954		P				•				•	•		•	•	•
Chrysocharis orbicularis (Nees, 1834)		P				•	•						•		•
Chrysocharis oscinidis Ashmead, 1888	(12)	Ne		•					•		•			•	•
Chrysocharis pentheus (Walker, 1839)		P							•						•
Chrysocharis pubicornis (Zetterstedt, 1838)		H O A		•		•		•					•	•	•
Chrysocharis tristis Hansson, 1987		P										•			
Chrysocharis viridis (Nees, 1834)	(13)	H				•				•	•		•		
Chrysonotomyia formosa (Westwood, 1833)	(14)	H		•		•	•				•		•	•	•
Chrysonotomyia pictipes (Crawford, 1912)		Ne	•												
Chrysonotomyia punctiventris (Crawford,1912)	(15)	Ne							•		•			•	•
Cirrospilus vittatus Walker, 1838		P													•
Closterocerus cinctipennis Ashmead, 1888	(16)	Ne									•			•	•
Closterocerus tricinctus (Ashmead, 1888)		Ne												•	
Closterocerus trifasciatus Westwood, 1833		Ne									•				
Closterocerus utahensis Crawford, 1912		Ne									•				
Diaulinopsis arenaria (Erdös, 1951)		P			•										•
Diaulinopsis callichroma Crawford, 1912		Ne	•								•			•	•

EULOPHIIDAE (suite)		Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
Diglyphus begini (Ashmead, 1904)	(17)	Ne	•						•		•			•	•
Diglyphus chabrias (Walker, 1838)		P											•		
Diglyphus crassinervis Erdös, 1958		P				•									•
Diglyphus intermedius (Girault, 1916)		Ne	•						•		•			•	•
Diglyphus indicus	(18)	O	•												
Diglyphus isaea (Walker, 1838)		H		•	•	•	•		•	•					•
Diglyphus minoeus Walker, 1838		P	•	•											•
Diglyphus pulchripes (Crawford, 1912)		Ne									•			•	•
Diglyphus websteri (Crawford, 1912)		Ne No							•		•			•	•
Hemiptarsenus dropion (Walker, 1839)		P							•						•
Hemiptarsenus fulvicollis Westwood, 1833		P													•
Hemiptarsenus varicornis (Girault, 1913)	(19)	Af O A									•				•
Hemiptarsenus zilahisebessi Erdös, 1951		P		•		•									
Hemiptarsenus sp.		Ne	•												
Meruana liriomyzae Boucek, 1988		Af	•										•		
Pediobius acantha (Walker, 1839)		P		•											•
Pediobius bucculatricis (Gahan, 1927)	(20)	Ne												•	
Pnigalio flavipes (Ashmead, 1886)		Ne									•				
Pnigalio minio (Wlaker, 1847)		Ne												•	
Pnigalio soemius (Walker, 1839)		P													•
Ratzeburgiola incompleta Boucek, 1971		P													•
Tetrastichus sp.		P	•												
Zagrammosoma americanum Girault, 1916		Ne													•
Zagrammosoma lineaticeps (Girault, 1915)	(21)	Ne													•
Zagrammosoma mirum Girault, 1916		Ne									•				
Zagrammosoma multilineatum (Ashmead, 1888)		Ne									•			•	

AUTRES FAMILLES	Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
EUCOILIDAE (22)														
Agrostocynips clavatus Diaz, 1976	No							•						
Cothonaspis sp.	Nc									•				•
Disorygma pacifica (Yoshimoto, 1962) (23)	Nc									•				
Eucoilidea sp.	Nc									•				•
Ganaspidium hunteri (Crawford, 1913) (24)	Nc									•				•
Ganaspidium pusillae Weld, 1955	Nc									•				•
Ganaspidium utilis Beardsley, 1988 (25)	Nc													•
Ganaspidium sp.	Nc	•												
Gronotoma micromorpha (Perkins, 1910)	Nc									•				
Hexacola sp.	Nc									•				
Pseudocoila sp.	Nc									•				
Zaeucoila unicaritana Ashmead, 1903	No									•				
PTEROMALIDAE														
Bubekia fallax Gahan, 1934	Nc												•	
Cyrtogaster vulgaris Walker, 1833	H		•											
Halticoptera aenea (Walker, 1833)	P				•			•		•				•
Halticoptera circulus (Walker, 1833) (26)	H	•	•		•	•		•		•		•	•	•
Halticoptera crius (Walker, 1839)	P		•											
Halticoptera flavicornis (Spinola, 1808)	Nc											•		
Halticoptera laevigata Thomson, 1876	Nc												•	
Halticoptera stella Girault, 1917	No							•	•					
Heteroschema sp.	No							•						
Lamprotatus tubero Walker, 1843	No							•						
Miscogaster sp.	P								•					
Miscogaster hortensis Walker, 1833	P				•									
Miscogaster maculata Walker, 1833	P				•									

AUTRES FAMILLES (suite)	Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
PTEROMALIDAE (suite)														
Solenotomorpha pampeana Blanchard, 1940	No							•						•
Sphegigaster orobanchiae Kurdjumov, 1912	P				•							•		
Stictomischus sp.	P													
Thinodytes cyzicus (Walker, 1839)	P				•									
Trigonogastrella parasitica Girault, 1915	Au	•												
EUPELMIDAE														
Anastatus sp. (27)		•												
MYMARIDAE														
Polynema sp (27)										•				
SCELIONIDAE														
Telenomus sp. (27)										•				
TETRACAMPIDAE														
Epiclerus nomocerus (Masi, 1934)	P													•
TRICHOGRAMMATIDAE														
Trichogramma minutum Riley, 1871 (27)	Nc									•				

PREDATEURS	Distribution	*L. brassicae*	*L. bryoniae*	*L. cicerina*	*L. congesta*	*L. dianthicola*	*L. flaveola*	*L. huidobrensis*	*L. pusilla*	*L. sativae*	*L. schmidti*	*L. strigata*	*L. trifoliearum*	*L. trifolii*
Aphidoletes aphidimyza (Rondani, 1847) Cecidomyiidae	P													•
Coenosia attenuata Stein, 1902 Muscidae	P		•											
Crossopalpus subaenescens Collin, 1960 Empididae (28)	P													•
Dicyphus tamaninii Wagner, 1951 Miridae	P													•
Platypalpus annulatus (Fallén, 1815) Empididae (29)	P													•

	Distribution	*L. betae*	*L. braziliensis*	*L. cepae*	*L. marginalis*	*L. nietzkei*	*L. pisivora*	*L. quadrata*	*L. urophorina*	*L. xanthocera*
BRACONIDAE										
Dacnusa maculipes Thomson, 1895	P						•			
Opius chilensis De Santis, 1966	No							•		
Opius cinerariae Fischer, 1963	P	•								
Opius eurytomoides De Santis, 1966	No							•		
Opius tersus (Foerster, 1862)	P						•			
EUCOILIDAE										
Microstilba bidentata Förster, 1869	P			•?		•				
EULOPHIDAE										
Chrysocharis sp.	P	•								
Chrysonotomyia punctiventris (Crawford, 1912)	Ne				•					
Cirrospilus vittatus Walker, 1838	P									•
Diaulinopsis callichroma Crawford, 1912	Ne				•					
Diglyphus isaea (Walker, 1838)	P									•
Hemiptarsenus sp.	P	•								
Zaommomyia vonones (Walker, 1839) (30)	Ne				•					
PTEROMALIDAE										
Halticoptera circulus (Walker, 1833)	P			•?		•				•
Halticoptera sp.	No		•							
Heteroschema sp.	Ne				•					
Miscogaster elegans Walker, 1833	P									•
Seladerma sp.	P								•	

NOTES SUR LES CITATIONS

(1) Ces espèces appartiennent à la sous-famille des Aphidiinae et par conséquent sont considérées comme étant des parasites stricts de pucerons. Ces citations sont douteuses.
(2) Cité aussi sous le nom de *Dacnusa daimenes* et *Dacnusa* sp. vic. *daimenes* Nixon. *Dacnusa incerta* Gourreau cité par Fulmek, 1962 comme parasite de *Liriomyza pusilla* est en fait un parasite de *Liriomyza pascuum* (Griffiths, 1968).
(3) Cité aussi sous le nom invalide de *Dacnusa misella*.
(4) Cité sous le nom de genre *Rhizarcha*, un synonyme de *Dacnusa*
(5) Cette espèce est en fait un parasite d'*Agromyza frontella* (Rondani) introduit en Amérique du nord et attaquant occasionnellement des *Liriomyza*.
(6) Cité sous le nom de genre *Rhizarcha*, un synonyme de *Dacnusa*
(7) Cité aussi sous les noms génériques invalides de *Nosopoeus* et *Nosopoea*.
(8) *Chrysocharis mallochi* Gahan, 1917 est synonyme de cette espèce (Hansson, 1987).
(9) *Chrysocharis albicans* Delucchi, 1954 est synonyme de cette espèce (Hansson, 1985).
(10) *Paracrias phytomyzae* Bréthes, 1923 cité aussi sous le nom de genre *Chrysocharis* est en fait un synonyme de cette espèce (Hansson, 1987).
(11) *Chrysocharis petiolota* Girault, 1918 est un nom préoccupé correspondant à ce taxon.
(12) *Chrysocharis parksi* Crawford, 1912, espèce fréquemment citée, est synonyme de cette espèce (Hansson, 1985).
(13) *Chrysocharis melaenis* Walker, 1839 est synonyme de cette espèce (Hansson, 1985).
(14) *Chrysonotomyia fullawayi* (Crawford) et *C. variipes* (Crawford) parfois cité sous le nom de genre *Derostenus* sont des synonymes de *C. formosa*. Les noms de genre : *Achrysocharella*, *Achrysocharis*, *Enaysma*, *Neochrysocharis* parfois utilisés pour cette espèce sont des synonymes de *Chrysonotomyia* (Boucek, 1988) même si cela est parfois contesté.
(15) *Chrysonotomyia arizonensis* (Crawford) et *C. agromyzae* (Crawford) sont synonymes de *C. punctiventris* (Yoshimoto, 1978), cette dernière espèce pourrait être en fait un synonyme de *C. pictipes* (Lasalle et Parella, 1991).
(16) Les espèces appartenant à ce genre méritent une révision et les citations pourraient être erronées.
(17) Dans les publications anciennes, les espèces de *Diglyphus* sont parfois citées sous le nom impropre de *Solenotus*.
(18) Espèce citée par Ipe (1987) et dont nous n'avons pu retrouver la description, probablement un *nomen nudum*.
(19) *Hemiptarsenus semialbiclava* Girault, 1916, parfois cité sous l'émendation injustifiée de *semialbiclavus*, est en fait un synonyme de cette espèce (Boucek, 1988).
(20) Cette espèce citée comme parasite de *Liriomyza* est aussi connue comme parasite de *Bucculatrix* (Lépidoptère), cette citation mérite confirmation.
(21) *Mirzagrammosoma* Girault qui est le nom de genre le plus couramment utilisé pour cette espèce est un synonyme de *Zagrammosoma* (Lasalle, 1989).
(22) La nomenclature générique des Eucoilidae mérite une totale révision et de nombreuses citations sont en fait dues à de fausses identifications.
(23) Espèce décrite et citée sous le nom générique de *Cothonaspis*.
(24) Parfois cité sous les noms de genre *Pseudeucoila* ou *Eucoila*.
(25) Cette espèce a été confondue avec *Ganaspidium hunteri* (Crawford, 1913) à Hawaii.
(26) Les citations sous le nom de *H. patellana* (Dalman, 1818) sont dues à des erreurs d'identification et doivent en fait correspondre à cette espèce.
(27) Au vue de la biologie des espèces appartenant à ce genre, cette citation est très probablement erronée.
(28) Citée sous le nom générique de *Drapetis*.
(29) Citée sous le nom générique de *Tachydromia*.
(30) *Chrysocharis majoriani* (Girault) est un synonyme de cette espèce

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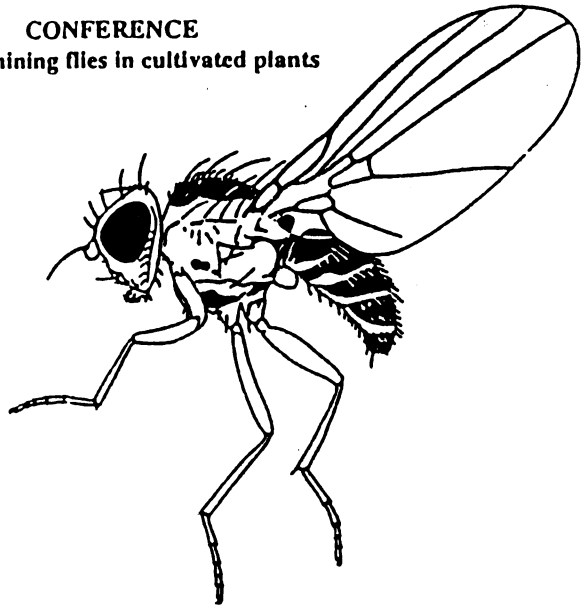
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"LIRIOMYZA"

COLLOQUE
sur les mouches mineuses des plantes cultivées

CONFERENCE
on leaf-mining flies in cultivated plants



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SOMMAIRE

SESSIONS PLENIERES

Pages

IIEME 1 : "ASPECTS SYSTEMATIQUES, INTRODUCTIONS, REGLEMENTATION" :
"SYSTEMATIC, INTRODUCTIONS, REGULATION" :

- Liste des espèces de Liriomyza d'importance agronomique, leurs synonymes et leurs régions biogéographiques,
  MARTINEZ M. Station de Zoologie *INRA* (Versailles) 1
- Identification of Liriomyza species using enzyme electrophoresis,
  OUDMAN L., Plant Protection Service (Wageningen) 7
- Identification of Liriomyza spp. using PCR fingerprinting with random arbitrary primers,
  CARTER M.J. Queens University (Belfast) 9
- Techniques d'élevage de masse permanent de Liriomyza trifolii et de Liriomyza huidobrensis,
  DALLE M., BORDAT D., *CIRAD-CA* (Montpellier) 17
- Plant Quarantine experience of Liriomyza spp. in England and Wales,
  BARTLETT P.W., Central Science laboratory *MAFF* (Harpenden) 23
- Liriomyza huidobrensis : Réseau de surveillance mis en place en France depuis 1990,
  PETTER F., *SRPV* Ile de France (Rungis)
  et LABORDE V. *DGAL-SDPV* (Paris) 31

IEME 2 : "BIOLOGIE, COMPORTEMENT DE LIRIOMYZA SPP." :
"BIOLOGY, COMPORTMENT OF LIRIOMYZA SPP." :

- Influence de la température sur le comportement de ponte des femelles de Liriomyza trifolii et de Liriomyza huidobrensis, mineuses serpentines américaines,
  OLIVERA C., BORDAT D., LETOURMY P., *CIRAD-CA* (Montpellier) 37
- Situation en France de Liriomyza huidobrensis,
  TROUVE C., P.V.(Loos en Gohelle),
  PHALIP M., P.V. (Biard),
  MARTINEZ M., *INRA* (Versailles) 49

• Les mouches mineuses du genre Liriomyza à l'Ile Maurice,
RAJABALEE, Institut de Recherches de l'Industrie Sucrière (Ile Maurice) 59

• Relation entre Liriomyza huidobrensis et Liriomyza trifolii à l'Ile de la Réunion,
VERCAMBRE B., *CIRAD-CA* (Saint-Denis),
DE CROZALS A., *ISTOM* (Cergy Pontoise) 71

• Statutory action against Liriomyza huidobrensis in the United Kingdom,
CHEEK S., MACDONALD O.C., BARTLETT P.W.
Central Science laboratory, *MAAF* (Harpenden) 79

• Incidence des dégâts de Liriomyza trifolii sur le rendement des cultures de haricot et de tomate dans les conditions du Sénégal,
COLY E.V., *ISRA-CDH* (Dakar),
BORDAT D. et LETOURMY P., *CIRAD-CA* (Montpellier) 87

ME 3 : "LUTTE CHIMIQUE, RESISTANCE VARIETALE"
"CHEMICAL CONTROL VARIETY RESISTANCE" :

• Observations comparées d'épidémiologie et de méthodes de lutte contre les mouches mineuses L. trifolii et L. huidobrensis sur cultures horticoles en Roussillon,
MARTIN C., SCHOEN L., *AGRIPHYTO* Méditerranée (Perpignan) 93

• Leafminer control and Chrysanthemum productivity with abamectin in strategies of resistance prevention/management,
MORENO - DUARTE R., Agroindustrias del Rio Frio Ltd. (Bogota, Columbia),
STARNER V. MALDONADO M. and SING V., *MERCK & Co.* (Rahway, USA) ... 103

• Liriomyza trifolii résistance in chrysanthemum,
VAN DIJK M.J. Institute for Evolutionary and Ecological Sciences (Leiden, the Netherlands),
DE JONG J., *CPRO-DLO* (Wageningen),
VAN DER MEIJDEN E., *I.E.E.S.* (Leiden) 111

• Susceptibility of Chrysanthemum to Liriomyza huidobrensis and Liriomyza trifolii, a comparison,
ED VAN DER MEIJDEN, R. BUTOT and M. J.VAN DIJK,
Institute for Evolutionary and Ecological Sciences (Leiden, The Netherlands) 119

• Recherche de résistances chez le melon envers Liriomyza trifolii,
PITRAT M., Station INRA d'amélioration des plantes maraîchères (Avignon),
BORDAT D. et DALLE M., *CIRAD-CA* (Montpellier) 127

• Sélection de lignées de tomates résistantes aux Liriomyza spp. à partir de l'espèce sauvage Lycopersicon cheesmanii,
LATERROT H., *INRA* (Avignon),
BORDAT D., *CIRAD-CA* (Montpellier)
MORETTI A., *INRA* (Avignon), DALLE M. et
RENAND M., *CIRAD-CA* (Montpellier) 135

THEME 4 : "LUTTE BIOLOGIQUE" / "*BIOLOGICAL CONTROL*"

- Liste des parasitoïdes et des prédateurs des Liriomyza d'importance économique dans le monde (Diptera, Agromyzidae),
  GRENOUILLET C. *ENSH* (Versailles),
  MARTINEZ M. et RASPLUS J.Y. *INRA* (Versailles) 143

- Biological control of leafminers in glasshouse lettuce,
  VAN DER LINDEN A., Glasshouse Crops Research Station (Naaldwijk) 157

- Comparaison des potentialités de Diglyphus isaea et de Hemiptarsenus varicornis, parasitoïdes de Liriomyza trifolii,
  FRANCO E. & BAUDRY G.
  Laboratoire *INRA* de biologie des invertébrés (Antibes) 163

- Influence de l'avermectine sur deux espèces de parasitoïdes de Liriomyza trifolii,
  NIELSEN-LE ROUX C., Institut Pasteur (Paris),
  et BORDAT D., *CIRAD-CA* (Montpellier) 171

- Sensibilité d'Opius dissitus et d'Hemiptarsenus varicornis à deux souches de Paecilomyces fumosoroseus,
  LAURIDSEN L., BORDAT D., *CIRAD-CA*
  et ROBERT P., *INRA* (Guyancourt) 183

POSTERS

- The overwintering potential of Liriomyza huidobrensis in the U.K.
  MACDONALD O.C. and WALTERS K.F.A., C.S.L. - MAFF (Harpenden) 193

Translation [nomenclature used by the authors has been retained, although some changes have been made in recent years].

"*Liriomyza*" - Symposium on Leafminer Flies in Cultivated Plants
List of parasitoids and predators of *Liriomyza* of economic importance in the world (Diptera: Agromyzidae)
C Grenouillet, M. Martinez & J.Y. Rasplus

Summary
A world list of all known parasites and predators of the 26 economically important species of *Liriomyza* is given. Species names of *Liriomyza* predators and parasitoids have been corrected in light of recent papers on the systematics of these groups. The list is presented in a table-form, giving for each parasitoids and predators, the species of *Liriomyza* attacked and the biogeographical area where the record have been done. A total number of 137 species of parasitoids (exclusively Hymenoptera) and 5 species of predators (4 Diptera and 1 Hemiptera) have been recorded from the literature.

Key-words: *Liriomyza*, pests, parasitoids, predators, world
Among the 339 species belonging to the genus *Liriomyza*, 26 species are considered to be of economic importance because of the damage they can cause to cultivated plants (cf. publication by M. Martinez in the proceedings of this Symposium). These agromyzids, like most insect species, are hosts for a number of parasitoids, primarily Hymenoptera which develop by consuming the tissues of the larva or pupa. Alongside these parasitic Hymenoptera, some species of predators are known to attack, at least occasionally, *Liriomyza* and as such can be considered as potential regulatory agents of the populations of these predators.

During the last 15 years, chemical control against some of these crop pests has led to the emergence of populations resistant to pesticides and thus favoured the development of research on the natural enemies of these *Liriomyza*. Many publications mentioning parasitoids of these Agromyzids have been written. We wanted to summarize this information and thus give as exhaustive a list as possible of the species of parasitoids and predators known to attack one or more of these 26 *Liriomyza* species of agronomic importance in the world. For this, we searched, in the bibliography, all the articles citing parasitoids and predators of these Diptera. More than 50 years of bibliographic data have been analyzed in this way. We have reviewed all the publications dealing with Agromyzidae parasitoids, as well as most of the taxonomic publications relating to species known or likely to be parasites of *Liriomyza*. We present the results of our literature search in the seven tables below. The first 6 tables deal with the 13 most commonly cited and therefore the most studied *Liriomyza* species; the last table gives the parasites for 9 other much less studied species. To the published data, we added some original data in particular concerning *L. huidobrensis*, which in France is parasitized by *Dacnusa sibirica*, *Diglyphus isaea* and *Hemiptarsenus dropion* (C. Trouve pers. comm.).

Several problems arose in the preparation of this list. In fact, the systematization of *Liriomyza* is particularly difficult and most species have sometimes been confused and/or often poorly identified (cf. Martinez *op. cit*.). For example, the citations of parasites of *L. trifolii* in Europe before 1975 actually relate to *L. congesta*, likewise the citations *of L. strigata* in the Afrotropical region, of *L. bryoniae* and of *L. flaveoIa* in the Neotropical region are erroneous, and in fact correspond to other species. Finally, some authors have misnamed the species they have studied, so Oatman's work on *L. pictella* (Thomson) actually concerns *L. sativae*.

It is the same for the parasitoids of Agromyzidae which belong to genera of Braconidae or Chalcidoidea which are still imperfectly known and particularly difficult. Due to this taxonomic ignorance of *Liriomyza* and their parasites, the literature is cluttered with identification errors that are sometimes difficult to locate and often transmitted in various writings and catalogs.

The systematic study of many groups of parasitoids still continuing, the nomenclature of the species cited as parasites of *Liriomyza* has been profoundly modified in recent years. The result is that a certain number

of species commonly cited in the literature are not found in the tables. In our tables, these species are mentioned under the name which should henceforth be used. In order to inform the reader of these nomenclatural changes, we have annotated the tables and explained these changes in the paragraph "Notes on citations".

It was not possible for us to indicate the whole of the bibliography consulted (several hundred titles), and consequently we only indicated the most important references or those providing original data.

In total, 137 species of parasitoids, representing 9 families of Hymenoptera are cited in the literature and only 5 species of predators (Diptera and Hemiptera). Two families of micro-hymenoptera dominate the category of parasitoids: the Eulophidae (Chalcidoidea) and the Braconidae (Ichneumonoidea) with respectively 51 and 50 species cited (Table 1). Next come the Pteromalidae (Chalcidoidea) with 19 recorded species and the Eucoilidae (Cynipoidea) with 12 species. Among the remaining families, only that of the Tetracampidae includes a species effectively parasitizing *Liriomyza*; the four other families: Eupelmidae (1 sp.), Mymaridae (1 sp.), Scelionidae (1 sp.) and Trichogrammatidae (1 sp.) are in our opinion mistakenly cited as parasitoids of *Liriomyza*. (cf. notes). For only 4 species of *Liriomyza* of agronomic importance are no parasitoids known.

It should be noted that the Nearctic (North America) and West-Palearctic (Europe) regions are the best studied and the parasitic processions of *Liriomyza* living in these regions are by far the most complete. Many species of parasites probably remain to be discovered in Africa, South America and Asia where few studies have been carried out. Moreover, it is certain that even in Europe new parasites of these miners remain to be discovered.

Appendix 7.6 Table 1. List of *Liriomyza* species treated and the number of parasites cited for each of them.

Liste des espèces de *Liriomyza*	**Eulo**	**Brac**	**Pter**	**Euco**	**Tetr**
Liriomyza betae (Coquillet, 1900) (A)[1]	2	1			
Liriomyza brassicae (Riley, 1885) (P, NE, AF, NO, O, A)	12	2	2	1	
Liriomyza braziliensis (Frost, 1939) (NO)			1		
Liriomyza bryoniae (Kaltenbach, 1858) (P)	7	10	3		
Liriomyza caulophaga (Kleinschmidt, 1960) (O, A)					
Liriomyza cepae (Hering, 1927) (P)			1	1	
Liriomyza chinensis (Kato, 1949) (P, O)					
Liriomyza cicerina (Rondani, 1856) (P, O)	2	2			
Liriomyza clianthi (Watt, 1923) (A)					
Liriomyza congesta (Becker, 1903) (P, O)	8	14	6		
Liriomyza dianthicola (Venturi, 1949) (P, NO)	3		1		
Liriomyza flaveola (Fallen, 1823) (P, O)	1	5			
***Liriomyza huidobrensis* (Blanchard 1926) (P, NE, AF, NO, A)**	**11**	**1**	**6**	**1**	
Liriomyza marginalis (Malloch, 1913) (NE, NO)	3		1		
Liriomyza nietzkei Spencer, 1973 (P)			1	1	
Liriomyza orbona (Meigen, 1830) (P)					
Liriomyza pisivora Hering, 1957 (P)		2			
Liriomyza pusilla (Meigen, 1830) (P,O)	3	6	2		
Liriomyza quadrata (Malloch, 1934) (NO)		2			
***Liriomyza sativae* Blanchard, 1938 (P, NE, NO, A)**	**20**	**9**	**2**	**9**	
*Liriomyza schmidti (*Aldrich, 1921) (NE, NO)	1			-	
Liriomyza strigata (Meigen, 1830) (P 0)	9	10	3		
Liriomyza trifoliearum Spencer, 1973 (NE)	18	2	3		
***Liriomyza trifolii* (Burgess, 1880) (P, NE, AF, N, O A)**	**27**	**10**	**3**	**5**	**1**

Liste des espèces de *Liriomyza*	**Eulo**	**Brac**	**Pter**	**Euco**	**Tetr**
Liriomyza urophorina Mik, 1894 (P, O)			1		
Liriomyza xanthocera (Czerny, 1909) (P)	2		2		
Nombre total d'espèces de parasites:	**51**	**50**	**19**	**12**	**1**

P = Palaearctic region: NE = Nearctic region: AF = Afrotropical (or Ethiopian) region: NO = Neotropical region: O = Oriental region: A = Australian / Oceanian region (these abbreviations are also used in other tables). Eulo = Eulophidae: Brac = Braconidae: Pter = Pteromalidae:
Euco = Eucoilidae: Tetr = Tetracampidae.

[1]*Liriomyza chenopodii* (synonym *Liriomyza betae*) has only been recorded in Australia and New Zealand so its region has been altered to A from P

Appendix 7.6 Table 2.

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
Braconidae	*Chorebus daimenes* (Nixon, 1945)	2	P		1		1				1			1		
	Chorebus misellus (Marshall, 1891)	3	P		1		1							1		1
	Dacnusa areolaris (Nees, 1811)	4	P											1		
	Dacnusa dryas (Nixon, 1948)	5	NE												1	
	Dacnusa maculipes Thomson, 1895	6	P		1		1				1			1		1
Eulophidae	*Chrysocharis crassicapus* (Thomson, 1878)	8	H	1								1				
	Chrysocharis entedonoides (Walker, 1872	9	H											1		
	Chrysocharis flacilla (Walker, 1842)	10	NO							1						
	*Chrysocharis girault*i Yoshimoto, 1973	11	NE												1	
	Chrysocharis oscinidis Ashmead, 1888	12	NE		1					1		1				1
	Chrysocharis viridis (Nees, 1834)	13	H				1				1	1		1		
	Chrysonotomyia formosa (Westwood, 1833)	14	H		1		1	1				1		1		1
	Chrysonotomyia punctiventris (Crawford, 1912)	15	NE							1		1				1
	Closterocerus cinctipennis Ashmead, 1888	16	NE									1				1
	Diglyphus begini (Ashmead, 1904)	17	NC							1		1			1	1
	Diglyphus indicus	18	O	1												
	Hemiptarsenus varicornis (Girault, 1913)	19	AF, O, A									1				1
	Pediobius bucculatricis (Gahan, 1927)	20	NE												1	
	Zagrammosoma lineaticeps (Girault, 1915)	21	NE													1
Eucoilidae		22														
Eucoilidae	*Disorygma pacifica* (Yoshimoto, 1962)	23	NE									1				
Eucoilidae	*Ganaspidium hunteri* (Crawford, 1913	24	NE									1				1
Eucoilidae	*Ganaspidium utilis* Beardsley, 1988	25	NE													1
Pteromalidae	*Halticoptera circulus* (Walker, 1833)	26	H	1	1		1	1		1		1		1	1	1
Braconidae	*Euopius analis* (Fischer, 1965)		NE									1				1

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
	Oenonogastra microrhopalae (Ashmead, 1896)		NE									1				1
Braconidae	*Opius bruneipes* Gahan, 1913		NC									1				1
	Opius dimidiatus Ashmead, 1889		NE, P		1							1			1	1
	Opius dissitus Muesebeck, 1963		NE, P									1				1
	Opius mandibularis Gahan, 1915		NE									1				1
	Opius aridis Gahan, 1913		NE									1				
	Opius cinerariae Fischer, 1963		NE, A	1								1				
	Opius suturalis Gahan, 1913		NE									1				
Eucoilidae	*Cothonaspis* sp.		NE									1				1
	Eucoilidea sp.		NE									1				1
	Ganaspidium pusillae Weld, 1955		NE									1				1
	Gronotoma micromorpha (Perkins, 1910)		NE									1				
	Hexacola sp.		NE									1				
	Pseudocoila sp.		NE									1				
	Zaeucoila unicaritana Ashmead, 1903		NO									1				
Eulophidae	*Chrysocharis ainsliei* (Crawford, 1912)		NE, P							1		1			1	1
	Diglyphus intermedius (Girault, 1916)		NE	1						1		1			1	1
	Diglyphus websteri Crawford, 1912)		NE, NO							1		1			1	1
	Chrysocharis liriomyzae Delucchi, 1954		P				1				1	1		1	1	1
	Diaulinopsis callichroma Crawford, 1912		NE	1								1				1
	Diglyphus pulchripes (Crawford, 1912)		NE									1			1	1
	Chrysocharis caribea Bouček, 1977		NO							1		1				
	Closterocerus trifasciatus Westwood, 1833		NE	1								1				
	Closterocerus utahensis Crawford, 1912		NE									1				
	Pnigalio minio (Walker, 1847)		NE									1			1	
	Zagrammosoma mirum Girault, 1916		NE									1				
	Zagrammosoma multilineatum (Ashmead, 1888)		NE									1			1	

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
Pteromalidae	*Halticoptera aenea* (Walker, 1833)		P				1			1		1				1
Braconidae	*Dacnusa sibirica* Telenga, 1934		P		1											1
Braconidae	*Opius pallipes* Wesmael, 1835		P		1											1
	Chorebus aphantus (Marshall, 1895)		P						1							
	Chorebus cambricus Griffiths, 1968		P								1					
	Chorebus pseudomisellus Griffiths, 1968		P				1									
	Chorebus testaceipes Griffiths, 1968		P								1					
	Colastes braconius Haliday, 18.33		P								1					
	Dacnusa adducta Griffiths, 1968		P						1							
	Dacnusa discolor (Foester, 1862)		P											1		
	Dacnusa flavicoxa Thomson. 1895		P				1									
	Dacnusa hospita (Foester, 1862)		p		1											
	Dapsilarthra balteata (Thomson, 1895)		P				1		1							
	Dapsilarthra testacea Griffiths, 1968		P								1					
	Opius abscissus Thomson, 1895		P													
	Opius aemulus Haliday. 1837		P													
	Opius exiguus Wesmael, 1835		P													
	Opius ilicis Nixon, 1939		P				1									
	Opius levis Wesmael, 1835		P				1		1							
	Opius lugens Haliday, 1837		P				1									
	Opius minor Fischer, 1957		p				1									
	Opius monilicornis Fischer, 1962		P			1										
	Opius pendulus Haliday, 1837		P				1									
	Opius phaseoli Fischer, 1963		O		1											
	Opius polyzonius Wesmael, 1835		P													
	Opius propodealis Fischer, 1958		P													
	Opius pygmaeotor (Nees, 1834)		P													

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
	Opius pygmaeus Fischer, 1962		P			1	1									
	Opius rex Fischer, 1958		P						1							
	Opius scabriventris Nixon, 1955		NO							1						
Braconidae	*Pseudopezomachus cursitans* Ferrière, 1930)		P				1									
Eucoilidae	*Agrostocynips clavatus* Diaz, 1976		NO							1						
Eucoilidae	*Ganaspidium* sp.		NE	1												
Eulophidae	*Chrysocharis pentheus* (Walker, 1839)		P							1						1
	Diglyphus isaea (Walker, 1838)		H		1	1	1	1		1	1					1
	Hemiptarsenus dropion (Walker, 1839)		P							1						1
	Chrysocharis orbicularis (Nees. 1834)		P				1	1						1		1
	Chrysocharis pubicornis (Zetterstedt, 1838)		H, O, A		1		1		1					1		1
	Cirrospilus vittatus Walker, 1838		P													1
	Diaulinopsis arenaria (Erdös, 1951)		P													1
	Diglyphus crassinervis Erdös, 1958		P				1									1
	Diglyphus minoeus Walker, 1838		P	1	1											1
	Hemiptarsenus fulvicollis Westwood, 1833		P													1
	Pediobius acantha (Walker, 1839)		P		1											1
	Pnigalio soemius (Walker, 1839)		P													1
	Ratzeburgiola incompleta Bouček, 1971		P													1
	Zagrammosoma americanum Girault, 1916		NE													1
	Chrysocharis chilo (Walker, 1839)		P	1										1		
	Chrysocharis clarkae Yoshimoto, 1973		H												1	
	Chrysocharis tristis Hansson, 1987		P										1			
	Chrysonotomyia pictipes (Crawford, 1912)		NE	1												
	Closterocerus tricinctus (Ashmead, 1888)		NE													
	Diglyphus chabrias (Walker, 1838)		P											1		
	Hemiptarsenus zilahisebessi Erdös, 1951		P		1		1									

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
	Hemiptarsenus sp.		NE	1												
	Meruana liriomyzae Bouček, 1988		AF	1										1		
	Pnigalio flavipes (Ashmead, 1886)		NE													
	Tetrastichus sp.		P	1												
Pteromalidae	*Solenotomorpha pampeana* Blanchard, 1940		NO							1						1
	Bubekia fallax Gahan, 1934		NO												1	
	Cyrtogaster vulgaris Walker, 1833		H		1											
	Halticoptera crius (Walker, 1839)		P		1											
	Halticoptera flavicornis (Spinola, 1808)		NE											1		
	Halticoptera laevigata Thomson, 1876		NE												1	
	Halticoptera stella Girault, 1917		NO							1	1					
	Heteroschema sp.		NO							1						
	Lamprotalus tubero Walker, 1843		NO							1						
	Miscogaster sp.		P								1					
	Miscogaster hortensis Walker, 1833		P				1									
	Miscogaster maculata Walker, 1833		P				1									
	Sphegigaster orobanchiae Kurdjumov, 1912		P				1									
	Stictomischus sp.		P											1		
	Thinodytes cyzicus (Walker, 1839)		P				1									
	Trigonogastrella parasitica Girault, 1915		AU	1												
Tetracampidae	*Epiclerus nomocerus* (Masi, 1934)		P													1
Questionable																
Mymaridae	*Polynema* sp.	27										1				
Scelionidae	*Telenomus* sp.	27										1				
Trichogrammatidae	*Trichogramma minutum* Riley, 1871	27	NE									1				
Eupelmidae	*Anastatus* sp.	27		1												

Family	Species	Footnote	Distr.	*L. bra*	*L. bry*	*L. cice*	*L. con*	*L. dia*	*L. flav*	*L. hui*	*L. pus*	*L. sat*	*L. sch*	*L. str*	*L. trifolea*	*L. trifoli*
Braconidae	*Aphidius ervi* Haliday, 1834	1	P		1											
Braconidae	*Aphidius matricariae* Haliday, 1834	1	P													
Braconidae	*Diaeretiella rapae* (M'Intosh, 1855)	1	P	?												
Predators																
Cecidomyiidae	*Aphidoletes aphidimyza* (Rondani, 1847)		P													1
Muscidae	*Coenosia attenuata* Stein, 1902		P		1											
Empididae	*Crossopalpus subaenescens* Collin, 1960	28	P													1
Miridae	*Dicyphus tamaninii* Wagner, 1951		P													1
Empididae	*Platypalpus annulatus* (Fallén, 1815)	29	P													1

L. bra=Liriomyza brassicae; L. bry=Liriomyza bryoniae; L. cice=Liriomyza cicerina; L. con=Liriomyza congesta; L. dia=Liriomyza dianthicola; L. flav=Liriomyza flavella; L. hui=Liriomyza huidobrensis; L. pus=Liriomyza pusilla; L. sat=Liriomyza sativae; L. sch=Liriomyza schmidti; L. str=Liriomyza strigata; L. trifolea=Liriomyza trifolearum; L. trifoli=Liriomyza trifolii

Appendix 7.6 Table 2 continued

Family	Species	Distrib.	L. betae[1]	L. braz	L. cep	L. marg	L. niet	L. pisiv	L. quad	L. uroph	L. xan
Braconidae	*Dacnusa maculipes* Thompson, 1895	P						1			
	Opius chilensis De Santis, 1966	No							1		
	Opius cinerariae Fischer, 1963	A[1]	1								
	Opius eurytonoides De Santis, 1966	No							1		
	Opius tersis (Foerster, 1862)	P						1			
Eucoilidae	*Microstilba bidentata* (Förster, 1869)	P			1?		1				
Eulophidae	*Chrysocharis* sp.	A[1]	1								
	Chrysonotomyia punctiventris (Crawford, 1912)	Ne				1					
	Cirrospilus vittatus Walker, 1838	P									1
	Diaulinopsis callichroma (Crawford, 1912)	Ne				1					
	Diglyphus isaea (Walker, 1838)	P									1

Family	Species	Distrib.	L. betae[1]	L. braz	L. cep	L. marg	L. niet	L. pisiv	L. quad	L. uroph	L. xan
	Hemiptarsenus sp.	A[1]	1								
	Zoommomyia vonones (Walker, 1839) (30)	Ne				1					
Pteromalidae	*Halticoptera circulus* (Walker, 1833)	P			1?		1				1
	Halticoptera sp.	No		1							
	Heteroschema sp.	Ne				1					
	Miscogaster elegans Walker, 1833	P									1
	Seladerma sp.	P								1	

L. betae=Liriomyza betae; L. braz=Liriomyza braziliensis; L. cep=Liriomyza cepae; L. marg=Liriomyza marginalis; L. niet=Liriomyza nietzkei ; L. pisiv=Liriomyza pisivora; L. quad=Liriomyza quadrata; L. uroph=Liriomyza urophorina
L. xan=Liriomyza xanthocera

[1]*Liriomyza chenopodii* (synonym *Liriomyza betae*) has only been recorded in Australia and New Zealand so the region for the three recorded parasitoids has been altered to A from P

Notes on citations

(1) These species belong to the Aphidiinae subfamily and therefore are considered to be strict aphid pests. These quotes are questionable.
(2) Also cited under the name *Dacnusa daimenes* and *Dacnusa* sp. vic. *daimenes* Nixon. *Dacnusa uncerta* Goureau cited by Fulmek, 1962 as a parasite of *Liriomyza pusilla* makes it a parasite of *Liriomyza pascuum* (Griffiths 1968).
(3) Also cited under the invalid name of *Dacnusa misella*.
(4) Cited under the genus name *Rhizarcha*, a synonym of *Dacnusa*
(5) This species is in fact a parasite of *Agromyza frontella* (Rondani) introduced in North America and occasionally attacking *Liriomyza*.
(6) Cited under the genus name *Rhizarcha*, a synonym of *Dacnusa*
(7) Also cited under the invalid generic names of *Nosopoeus* and *Nosopoea*.
(8) *Chrysocharis mallochi* Gahan, 1917 is synonymous with this species (Hansson 1987)
(9) *Chrysocharis albicans* Delucchi, 1954 is synonymous with this species (Hansson 1985)
(10) *Paracrias phytomyzae* Bréthes, 1923 also cited under the name of the genus *Chrysocharis* is in fact a synonym of this species (Hansson 1987)
(11) *Chrysocharis petiolota* Girault, 1918 is a concerned name corresponding to this taxon
(12) *Chrysocharis parksi* Crawford, 1912, a frequently cited species, is synonymous with this species (Hansson 1985).
(13) *Chrysocharis melaenis* Walker, 1839 is synonymous with this species (Hansson 1985).
(14) *Chrysonotomyia fullawayi* (Crawford) and *C. variipes* (Crawford) sometimes referred to as the genus *Derostenus* are synonyms of *C. formosa*. The names of the genus: *Achrysocharella*, *Achrysocharis*, *Enaysma*, *Neochrysocharis* sometimes used for this species are synonyms of *Chrysonotomyia* (Boucek, 1988) although this is sometimes disputed.
(15) *Chrysonotomyia arizonensis* (Crawford) *and C. agromyzae* (Crawford) are synonyms of *C. punctiventris* (Yoshimoto 1978) the latter species could in fact be a synonym of *C. pictipes* (LaSalle and Parrella, 1991).
(16) Species belonging to this genus deserve revision and citations could be wrong.
(17) In older publications, *Diglyphus* species are sometimes cited under the improper name of *Solenotus*.
(18) Species cited by Ipe (1987) and whose description we have not been able to find, probably a *nomen nudum*.
(19) *Hemiptarsenus semialbiclava* Girault 1916, sometimes cited under the unjustified emendation of *semialbiclavus*, is in fact a synonym of this species (Bouček 1988).
(20) This species cited as a parasite of *Liriomyza* is also known as a parasite of *Bucculatrix* (Lepidoptera), this citation deserves confirmation.
(21) *Mirzagrammosoma* Girault which is the most commonly used genus name for this species is a synonym of *Zagrammosoma* (LaSalle 1989).
(22) The generic nomenclature of Eucoilidae deserves a total revision and many citations are in fact due to false identifications.
(23) Species described and cited under the generic name of *Cothonaspis*.
(24) Sometimes cited under the genus *Pseudeucoiia* or *Eucoila*.
(25) This species has been confused with *Ganaspidium hunteri* (Crawford, 1913) in Hawaii.
(26) The citations under the name of *H. patellana* (Dalman, 1818) are due to identification errors and must in fact correspond to this species.
(27) In view of the biology of the species belonging to this genus, this quote is very probably erroneous.
(28) Cited under the generic name of *Drapetis*.
(29) Cited under the generic name of *Tachydromia*.
(30) *Chrysocharis majoriani* (Girault) is a synonym of this species

References

As in the original.

Appendix 7.7 Bolton M. 1986. Die Amerikaanse blaarmyner: 'n belangrike plaag by tamaties. *Roodeplaat Bulletin* 11, 8-10.

Translation

The American leaf miner: an important pest in tomatoes
Magda Bolton

The American leaf miner (*Liriomyza trifolii*) was first described in 1880 in Maryland, USA, but it was not until 1976 that pest control began to take on chrysanthemums in California. In 1977 identical problems occurred in Britain and since then the insect has spread worldwide mainly due to the international trade in chrysanthemum cuttings: Typical leaf miner damage is caused by the larvae of *L. trifolii* tunnelling into the leaf tissue. The foliage quality of cut flowers is consequently weak and in tomatoes damage can be so severe that leaf functions become benign and leaves wither and die off. The harvest period is therefore shortened and fruit quality reduced.

The two main reasons for the pest status of the insect are the wide range of plants that serve as hosts, as well as the short life cycle. This leaf miner is a pest in various vegetable and ornamental plant species and was first found in South Africa during February 1983 on chrysanthemums and Gypsophila in glass houses near Krugersdorp and Kempton Park. Later it was also found on chrysanthemums and vines under outdoor cultivation in the Malelane district. Infestation at the respective growers was particularly high and large financial losses were suffered due to the unmarketability of the cut flowers. Within one year after the first reporting of the insect in the RSA, serious damage occurred to tomatoes in the Tzaneen (Letsitele and Mooketsie) area. At that time no remedy was registered against the pest and the normal spray program for the control of the complex other pests did not have the necessary effect. Since then, the plague has also been found on tomatoes in the districts of Barberton (Louws Creek, Malelane), Leydsdorp, Gravelotte, Pretoria (Skeerpoort), Brits and Rustenburg.

Especially in the Letsitele and Mooketsi areas, several weed and wild plant species were found that served as hosts for *L. trifolii* and the following were identified:

Asteraceae:
Acanthospermum hispidum (upright cluster)
Bidens pilosa (handsome boyfriend)
Epaltes gariepina
Flaveria bidentis (smelterbossie)
Senecio inauquidens (hunger bush type)
Tagetes minuta (Kakiebos)
Xanthium strumarium (cancerous rose)

Chenopodiaceae:
Chenopodium carinatum (green dog bush)
Convolvulaceae:
Ipomoea plebeia africana (wild potato)

Cucurbitaceae:
Cucumis metuliferus (wild cucumber)

Lamiaceae:
Leonotis nepetifolia (wildedagga)

Solanaceae:
Datura stramonium (stink leaf)

Regular surveys by farmers showed that all of the above hosts were not equivalent in susceptibility to *L. trifolii*. The heaviest infestation occurred on *F. bidentis* and *S. inauquidens*. However, all the host plants served as a source for re-infestation of new tomato plantations and were therefore important contributing factors to the pest status of *L. trifolii*.

The insect has four growth stages, viz. egg, larva, pupae and adult flies. The flies are about 1 mm long with a characteristic yellow and black body. Using her ovipositor, the female makes small holes or punctures in the leaves. These punctures are visible as fine, light speckles on the leaves and the plant juice that flows from them serves as a food source for both the male and female flies. The female lays her eggs in 8 to 15% of these punctures.

At a temperature of 26-28 ° C the eggs hatch and the larvae start to tunnel in the leaves to cause the typical leaf miner damage - as shown in this photo.



At a temperature of 27 °C (± 1°C) the eggs hatch within 2 days and the larvae begin to tunnel into the leaves. After 7 to 8 days, the larvae mature, move out of the leaves and fall to the ground to pupate. After 8 days, the adult flies appear and according to the literature, the female is able to lay up to 250 eggs. The life cycle is thus completed within a short period of time; consequently, a population can build up rapidly at high temperatures.

The only pesticide currently registered for the control of the leaf miner in tomatoes is an insect growth regulator that interrupts or prevents the normal processes of egg hatching, larval moulting and also pupation. In this way the insect is prevented from reaching maturity and the succession of generations is broken. The product has cyromazine as active ingredient and is commercially available as Patron

(wettable powder). The recommended dose is 20 g/100 L water up to a maximum of 500 L spray mixture /ha. When applying 600 L spray mixture / ha, only 110 g should be used and for 700 L, 120 g and so on. The maximum spray mixture per ha must not exceed 1,000 L and 150 g Patron/ha/spray. Weekly sprays are recommended and the safety period (last application until harvest) of 3 days must be strictly adhered to. According to the manufacturer, it is important to mix the toxin correctly, by first mixing the powder with a small amount of water to a liquid cream. The "cream" is then added to half the required volume of water in the tank and then the water is replenished to the desired volume while stirring the mixture. It is also desirable to pour the "cream" through a 50-mesh sieve.

Observations among farmers have highlighted the following important aspects for the control of L. trifolii in tomatoes:

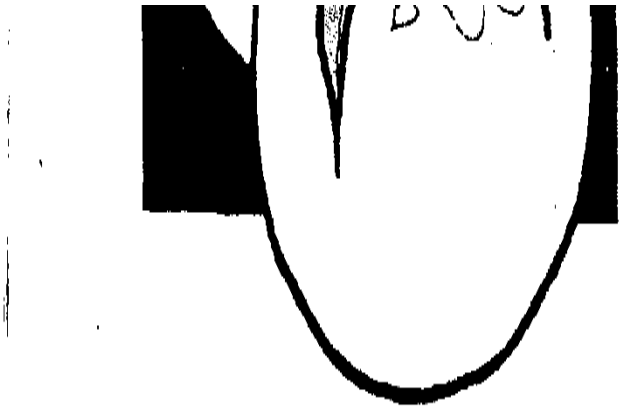
• Seedling growers must apply strict control measures to prevent the pest from spreading through infested seedlings.

• Producers should only plant seedlings that are free of leaf miner infestation (If seedlings are purchased, it is important to insist on clean planting material).

Producers must apply effective control measures during the 10 to 12 weeks from transplanting to the first harvest, as this is crucial for the duration of the harvest period as well as the quality of the fruit.

• Old tomato plants must be destroyed. and weed control should be applied in order to limit the build-up of leaf miner populations and spread to new plantings.

Original



Tipiese blaarmyniersskade word deur die larwes van *L. trifolii* veroorsaak, deurdat hulle in die blaarweefsel tonnel. Die loofkwaliteit van snyblomme is gevolglik swak en by tamaties kan beskadiging so ernstig wees dat blaarfunksies benadeel word en blare verdroog en afsterf. Die oesperiode word dus verkort en vrugkwaliteit verlaag.

Die twee hoofredes vir die plaagstatus van die

insek is die wye reeks plante wat as gashere dien, asook die kort lewensiklus.

Hierdie blaarmyner is 'n plaag by verskeie groente- en sierplantsoorte en is die eerste keer in Suid-Afrika gedurende Februarie 1983 op krisante en gipskruid in glashuise naby Krugersdorp en Kempton Park aangetref. Later is dit ook op krisante en rankgewasse onder buiteverbouing in die Malelane-distrik aangetref. Infestasie by die onderskeie kwekers was besonder hoog en groot finansiële verliese is gely vanweë die onbemarkbaarheid van die snyblomme.

Binne een jaar na die eerste aanmelding van die insek in die RSA, het ernstige skade by tamaties in die Tzaneen- (Letsitele- en Mooketsie-) area voorgekom. Indertyd was geen middel geregistreer teen die plaag nie en die normale spuitprogram vir die beheer van die kompleks ander plae het nie die nodige uitwerking gehad nie. Sedertdien is die plaag ook in die distrikte Barberton (Louws Creek, Malelane), Leydsdorp, Gravelotte, Pretoria (Skeerpoort), Brits en Rustenburg op tamaties aangetref.

Veral in die Letsitele- en Mooketsi-areas is verskeie onkruid- en wildeplantspesies aangetref wat as gashere vir *L. trifolii* gedien het en die volgende is geïdentifiseer:

Asteraceae: *Acanthospermum hispidum* (regopsterklits)
Bidens pilosa (knapsekêrel)
Epaltes gariepina
Flaveria bidentis (smelterbossie)
Senecio inauquidens (hongerbossie-tipe)
Tagetes minuta (Kakiebos)
Xanthium strumarium (kankerroos)

Chenopodiaceae : *Chenopodium carinatum* (groenhondebossie)

Convolvulaceae : *Ipomoea plebeia africana* (wildepatat)

Cucurbitaceae : *Cucumis metuliferus* (wildekomkommer)

Lamiaceae : *Leonotis nepetifolia* (wildedagga)

Solanaceae : *Datura stramonium* (stinkblaar)

Gereelde opnames by boere het getoon dat al bogenoemde gashere nie gelykwaardig ten opsigte van vatbaarheid vir *L. trifolii* was nie. Die swaarste infestasie het op *F. bidentis* en *S. inauquidens* voorgekom. Al die gasheerplante het egter as bron vir herinfestasie van nuwe tamatie-aanplantings gedien en was dus belangrike bydraende faktore tot die plaagstatus van *L. trifolii*.

Die insek het vier groeistadia, nl. eier, larwe, papie en volwasse vlieg. Die vlieë is ongeveer 1 mm lank met 'n kenmerkende geel en swart liggaam. Met behulp van haar eierboor maak die wyfie klein gaatjies of punkture in die blare. Hierdie punkture is as fyn, ligte spikkels op die blare sigbaar en die plantsap wat daaruit vloei dien as voedselbron vir beide die mannetjie- en wyfievlieë. Die wyfie lê haar eiers in 8 tot 15% van dié punkture.



By 'n temperatuur van 26-28°C broei die eiers uit en die larwes begin in die blare tonnel om die tipiese blaarmynerskade - soos op dié foto uitgebeeld - te veroorsaak

By 'n temperatuur van 27 °C (± 1 °C) broei die eiers binne 2 dae uit en die larwes begin in die blare tonnel. Na 7 tot 8 dae is die larwes volgroei, beweeg uit die blare en val grond toe om te pupeer. Na 8 dae verskyn die volwasse vlieë en volgens die literatuur is die wyfie in staat om tot 250 eiers te lê. Die lewensiklus word dus binne 'n kort tydperk voltooi; gevolglik kan 'n populasie vinnig by hoë temperature opbou.

Die enigste plaagdoder wat tans vir die beheer van die blaarmyner by tamaties geregistreer is, is 'n insekgroeireguleerder wat die normale prosesse van eieruitbroeiing, larvale vervelling en ook pupering onderbreek of voorkom. Sodoende word die insek verhoed om volwassenheid te bereik en word die opeenvolging van generasies verbreek. Die middel het siromasien as aktiewe bestanddeel en is in die handel as Patron (benatbare poeier) beskikbaar. Die aanbevole dosis is 20 g/100 ℓ water tot 'n maksimum van 500 ℓ

spuitmengsel/ha. Wanneer 600 ℓ spuitmengsel/ha toegedien word, moet slegs 110 g gebruik word en vir

tamaties na vore gebring:

- Saailingkwekers moet streng beheermaatreëls

Appendix 7.8 Visser D & Weintraub P. 2001. Outbreak of the potato leafminer, comparisons between Israel and South Africa. *Chips* 15(2), 38–39; Potato leafminer - lessons Israel. *Chips* 16(4), 39–40.

Outbreak of the Potato Leafminer
Comparisons between Israel and South Africa

(Diedrich Visser, ARC-Roodeplaat and Dr Phyllis Weintraub, Gilat Research Station, Israel)

Presence of the potato leafminer, *(Liriomyza huidobrensis)* was first reported in South Africa during the summer of 1999/2000. During that time it was responsible for losses of millions of rands to the potato industry. Yields dropped from above 40 tons/ha to below 15 tons/ha. Some farmers nearly went out of business because of the impact of this pest on their annual yields.

However, during the following summer (2000/2001), the damage caused by the fly decreased to some extent, enabling most farmers to obtain reasonable yields. What caused the sudden appearance of this pest? And why were the extreme losses in the first year not repeated during the second? These questions will most probably never be answered satisfactorily, but to broaden our views relating to this pest, we have examined the situation in Israel and tried to compare it to the situation in South Africa.

Introduction of the potato leafminer into Israel

Events surrounding the outbreak of the leafminer in Israel closely resemble that in South Africa. As in South Africa, the American leafminer *(Liriomyza trifolii)*, also occurs in Israel. However, just as in South Africa, they suddenly found themselves with a new devastating leafminer that could not be controlled chemically. The first outbreak occurred in 1992 in the Jordan Valley on chrysanthemums and was first reported on potatoes in 1993. Researchers suspected that the leafminer could have entered Israel a year or two before the first outbreak. This is also possible for the Sandveld, because some farmers confirmed that they noticed this leafminer years before the devastating outbreak in 1999. It is thus possible that a resistant, or partially resistant, population found its way to the Sandveld; probably became totally resistant over a short period following excessive application of various insecticides. Researchers in Israel also suspect that an already resistant population entered their country from Europe. The actual route of introduction, however, may never be known for either country.



Very characteristic leaf damage of the potato leafminer (Photo: Dr. Weintraub)

Change in pest status

After its introduction to potatoes in Israel in 1993, the potato leafminer caused heavy yield losses of around 30%. Population densities in potato fields increased yearly up to 1998, after which a drastic decline occurred.

Currently the potato leafminer is not considered a serious pest on potatoes in Israel. A similar situation is emerging in the Sandveld region. In the first year of introduction (1999), the leafminer caused extensive yield losses to almost all potato plantings in that region. During the following summer, damage levels lowered dramatically with only the odd farmer still reporting losses. What has caused this sudden decline in pest status?

There are three main reasons why pest numbers reach the economic (damage) threshold for a given crop: Firstly, the environmental conditions (weather); secondly, the absence of natural enemies in that ecosystem; and thirdly, the inability of direct control measures (spraying programs) to lower that pest's numbers.

Weather conditions

The influence of weather conditions on the survival of the potato leafminer is not always as simple as it seems. As with all insects, temperature affects their development: extreme heat and cold kill, cool temperatures slow development, and moderate to warm temperatures provide optimum developmental rates. The leafminer evolved in the relatively cool foothills of the Andes Mountains of South America and is sensitive to hot and dry conditions. However, other factors also change with the seasons, i.e. humidity and the presence of parasitoids. It is mentioned in the literature from Peru that parasitoids reproduce much slower during the colder months. The apparent increase in leafminer numbers during the cooler months could therefore be attributed both to the fact that this is the temperature of their "native" habitat and to the absence of parasitoids effective at cooler temperatures. To use temperature as a prediction of what will be expected is therefore overly simplistic – it can, however, be used as an indication of what can be expected. It is also noteworthy that the drastic decline in pest status of the leafminer in Israel followed an abnormally hot summer. Scientists also differ in opinion on the influence of humidity on the survival of the potato leafminer. Humidity can become an important factor when potatoes are under irrigation.

Researchers in Morocco and Chili men-

tioned that the potato leafminer prefers high humidities. However, more leafminers were found under relatively low ambient humidities in celery fields in Israel. It is possible that relative humidity could play a role, but then only as one of a complex of factors.

Natural enemies

When the pest appeared for the first time in both countries or rather, when heavy damage levels were first noticed, farmers tried to control the pest with their regular insecticides. When this did not have the desired effect, they started experimenting with all possible products. Neither chemical companies nor researchers had effective solutions. During this time the farmers were used (and sometimes misused) as guinea pigs by agents trying to prove that they had the wonder chemical that would solve the problem. Needless to say, most of these insecticides were ineffective. By the time the farmers realized this, they had unknowingly destroyed most of the natural enemy complex that normally keeps the leafminer in check. Thus, the destruction of this complex could have added to the abnormally fast increase in leafminer numbers. One of the most important parasitoids of the potato leafminer, *(Diglyphus* sp.), was found in both South Africa and Israel at the time of introduction of the leafminer. We therefore know that this natural enemy had been present, and that it was most probably eliminated by the wide spectrum of insecticides used against the leafminer.

Insecticides

When farmers realized that most insecticides were ineffective against adult leafminers, attempts were made to identify those chemicals used in other parts of the world with proven efficacy. Only two insecticides were commonly used on a worldwide scale against Liriomyza, abamectin and cyromazine. When farmers started using these two insecticides as recommended by Novartis (Syngenta), the situation seemed to improve. It was, however, only during the following year in the Sandveld that good control was reported with these two insecticides. Abamectin and cyromazine are translaminar insecticides; that is, unlike conventional insecticides, they penetrate through the leaf and affect the leafminer larvae. Therefore, they must be used differently than conventional insecticides; they need to be applied early when the leafminers are still in the larval stages in the leaf. Abamectin, cyromazine, and more recently spinosad, are the insecticides of choice in Israel for control of the leafminer.

Next issue

In the next issue of Chips we will discuss the use of insecticides, parasitoids and other control strategies against the potato leafminer in both South Africa and Israel. We will try to report on what works and what does not, and will also discuss research work conducted by Dr Weintraub at the Gilat Research Station in Israel. This will include studies on application timing of abamectin and cyromazine (Agrimec and Patron) and their effects on parasitoids. We will also look at the potential of neem as an insecticide against the potato leafminer. Farmers are invited to contact Diedrich Visser at (012) 841 9700 if there are still aspects relating to the leafminer that need clarifying. We shall then try to incorporate these questions in our next article in the next issue of *Chips*.

POTATO LEAFMINER – LESSONS FROM ISRAEL

(Diedrich Visser, ARC-Roodeplaat, Dr Phyllis Weintraub, Gilat Experiment Station, Israel)

The potato leafminer, *Liriomyza huidrobrensis*, is a serious pest on potato and various vegetables in South Africa. It first appeared in the Sandveld region during the summer of 1999/2000 and subsequently spread to the rest of South Africa in the short period of only one year. The same leafminer first appeared in Israel in 1992.

Since then, the Gilat Experimental Station in Israel undertook several research projects in an effort to gain more information regarding this pest and its control. Here we look at results of a few of these projects. Neem, Azadirachta indica is a tall evergreen tree native to India, grown for its bark and resin. Oil extracts from its seeds (also known as neem) are used as an insecticide against several insect pests. Neem has a low contact toxicity and needs to be ingested by insects in order to be effective. Neem is also considered an organic insecticide, and is therefore more acceptable to environmentalists, and one of the few insecticides available to organic growers for control of leafminers. The neem-based insecticide, Neemix-45 (4.5% azadirachtin), was evaluated against the potato leafminer on bean plants. A surfactant (Agral 90; 0.01%) was added after which plant leaves were dipped.

A soil drench was also performed on the soil surrounding the plants.Leaf dipping: At 15 ppm,, neem prevented any adults from emerging leaves treated at least one day prior to infestation, and 97% of adults when leaves were treated and when larvae were in their first instar. However, the number of pupae that emerged did not differ from the untreated control. It is therefore important to note that damage may still occur. Although the cycle of the fly is broken. Neem will thus not be able to give satisfactory control, if a source of infestation is just outside or near a field.

Soil drench: Pupae emerged from plants in treated soils with all concentrations up to 10 ppm, but no adults succeeded in emerging from these pupae, even at the lowest dosage of 1 ppm when treated at least one day prior to infestation. When soils were treated after infestation, a few adults succeeded in occlusion; but when the dose was increased to 10 ppm, no adults emerged, even if soils were treated when larvae were in their first instar. It is important to note that damage to plants may still occur, but the cycle is broken at the pupal stage (no adults emerge). Because neem seems to break down quickly when exposed to ultraviolet light, it might have potential in drip irrigation systems. The latter, however, still needs to be investigated. Another drawback to neem-based insecticides is their cost; however, commercial companies are aggressively working on reducing costs and making this natural insecticide more widely available.



Timing application of insecticides
Currently there are four insecticides registered against the potato leafminer in South Africa namely: Agrimec (abamectin), Patron (cyromazine), Suntap (cartab hydrocvhloride) and Tracer (spinosad). They all penetrate leaf surfaces to kill the larvae (translaminar and/or systemic). Suntap also has good contact activity against the flies, while Tracer also suppresses adult flies to some extent by acting as a stomach poison. The question arises as to which insecticide should be applied at what time, and with how many repetitions. Syngenta has indicated that Agrimec is more effective on very young plants, and therefore they recommended applying it first. Alternations of the four registered insecticides within a spraying program are essential to prevent the development of resistance against them. A good rule-of-thumb to prevent development of resistance is one kind (mode of action, e.g. stomach poison, contact poison, etc.) of insecticide per pest generation. Exactly how such a program must look is still under discussion.

However, the above-mentioned recommendations from Syngenta and data from Israel regarding the longevity of abamectin and cyromazine within plants, can help farmers make these decisions. In Israel, abamectin (Vertimec, 18g/) and cyromazine (Trigard, 50g/) were tested at manufacturer's recommended dosages of 10.8g ai/ha and 75g ai/ha, respectively for their longevity in potato plants. One percent Ultrafine oil was added to Vertimec. These two products were compared statistically in potato fields. Based on extensive monitoring of the leafminer, it was known that adult populations start increasing dramatically at the beginning of spring (April in Israel). Both these insecticides were sprayed only once, a week before the majority of the larvae would have been pupating (a stage not vulnerable to insecticides). A single application of cyromazine effectively controlled the potato leafminer until harvest (42 days after application). Abamectin, however, was effective for only one week after application. Residue analysis confirmed that cyromazine remained active in the foliage, albeit at decreasing levels over time, for at least three weeks after application. Similar trials were performed in celery fields. It was found early initial treatments, when leafminer populations were still very low, would break the cycle and prevent build-up of large field populations.



Leaf growth dies as a result of leafminer damage.



Characteristic punctures (feeding marks) in which eggs are also laid.

Monitoring

Nowadays most farmers do not monitor for the potato leafminer. Their presence is unmistakable and they normally arrive in such large numbers and in such a short period of time that monitoring is unnecessary.

However, there are some cases where monitoring could be useful, e.g. in an area where they have not been introduced or where their numbers remain low for some reason. But which monitoring techniques are best and what can one learn from monitoring the adults in a field? Monitoring methods vary from counting live larvae, mines, and pupae, to rearing adults from foliage, catching adults with a vacuum sampler and trapping adults in either yellow water buckets or on yellow sticky boards. Experiments with sticky yellow traps (13 x 16 cm) showed that most flies get caught at plant height (30 to 50 cm above ground). The least number of flies were caught at 10 cm above ground. This is an indication that the flies do not hide like the potato tuber moth, but prefers the open canopy. Overall there were no differences between the number of males and females that were caught in the sticky traps. No significant differences occurred in fly numbers on traps placed vertically and traps placed horizontally (sticky side up). In time-related catches (diel activity), it was found that almost all flies (82%) were caught between 6 and 9 o'clock in the morning, with the most active hour between 6 and 7 am. It seems as if sunrise might be the primary stimulant for activity, while the flies become less active when temperature rises. The fact that fly activity is at its peak during the hour following sunrise, can be used to time the spraying of adulticides. Insecticides like Suntap and Tracer, which also kill the adults, will therefore presumably come into contact with more adults during this time and therefore be more effective. Additionally abamectin-based insecticides are broken-down by ultraviolet light and should only be applied in the early morning hours or late afternoon.

Parasitoids

From various sources it is clear that the presence of parasitoids in a field is the only way to keep the potato leafminer under natural control. Several parasitoids species are known to attack the potato leafminer, of which *Diglyphus isaea* is mentioned to be the most effective. But what is the effect of insecticides used against the leafminer on these parasitoids? The answer is not simple. A lot of studies on both abamectin and cyromazine have contradictory results. One must remember that the insecticides are developed to "poison" the leafminer larvae inside leaves, and that the parasitoids larvae again must feed on these poisoned and dying larvae. The parasitoids larva therefore may not be able to complete its life cycle under these circumstances. It is thus understandable that these chemicals may indirectly affect parasitoids. The contact action of different insecticides against *Diglyphus isaea* is currently being investigated by ARC-Roodeplaat.

For more information regarding the potato leafminer or for the possible inclusion of insecticides in our tests, contract Diedrich Visser or Anelia Steyn at (012) 841 9611.

Appendix 5: Prospects for mass-rearing parasitoids to enable augmentative biological control of polyphagous agromyzid leafminers (*Liriomyza* spp.) in Australia

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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Contents

Executive Summary 3
1. Introduction 6
2. History of augmentative biological control of *Liriomyza* spp. 6
3. Life history of *Liriomyza* spp. 8
4. Life history of parasitoids with mass rearing potential 9
Idiobiont eulophid parasitoids 9
Koinobiont parasitoids 10
Braconid parasitoids *Dacnusa sibirica*, *Opius dissitus*, *O. pallipes* and *O. scabriventris* 10
Eucoiline parasitoids 10
5. Mass-rearing protocols for parasitoids of *Liriomyza* spp. 11
Plant production 11
Leafminer production 12
Parasitoid production 12
6. Constraints to commercial mass rearing 14
High production osts 14
Storage 15
Biased sex ratio 15
Management issues 16
7. Potential solutions to current constraints 16
Use of thelytokous strains 16
Quality control of mass-reared natural enemies 17
8. Australian situation 17
Diglyphus isaea 18
Neochrysocharis formosa 18
Opius spp. 18
Other parasitoid species 18
Conclusions 19
Appendices 21
References 39

Executive Summary

Relevant research has been reviewed to assess the current status of mass-rearing protocols required to produce parasitoid wasps for augmentative biological control of the polyphagous *Liriomyza* leafmining flies (*Liriomyza bryoniae*, *L. huidobrensis*, *L. sativae* and *L. trifolii*) in protected cropping.

Overseas, problems with the polyphagous *Liriomyza* pest species are universally associated with destruction of their parasitoids by excessive use of non-selective insecticides. It has been demonstrated repeatedly that to utilise parasitoids to control agromyzid leafminers, such as *L. sativae*, as part of an IPM program, it is necessary to minimise the use of broad-spectrum insecticides and to develop economic thresholds to reduce the number of spray applications.

In Europe and North America, only two species, *Diglyphus isaea* and *Dacnusa sibirica*, are currently reared commercially for augmentative biological control of *Liriomyza* leafminers. Relatively high costs of production have restricted usage of these natural enemies to mainly glasshouse producers. The more important mass-reared parasitoid, *Diglyphus isaea*, is a eulophid ectoparasitoid – the female wasp paralyses the agromyzid larva, then lays its egg in the mined leaf close to the agromyzid larva. After hatching, the parasitoid larva then feeds on the host larva and finally pupates in the leaf. The female wasp also kills many host larvae through host-feeding (to obtain sufficient reserves to allow oviposition) which is an important component of mortality of hosts. This leads to a dilemma: host feeding is important for the development of parasitoid eggs but adversely impacts the economics of mass-rearing as a large proportion of available hosts are killed outright rather than being a host for parasitoid larvae. The braconid wasp, *D. sibirica,* performs better than the eulophid, *D. isaea*, at lower temperatures but the situation is reversed at higher temperatures. In addition to *D. isaea* and *D. sibirica*, systems for mass-rearing of several other parasitoid species that attack *Liriomyza* spp. have been developed and, in some cases, applied on a small commercial scale.

These existing systems will prove useful in guiding any future efforts to commercially mass-rear wasps in Australia against leafminer pests. This review details further research and development that is required to facilitate successful mass-rearing of parasitoids of exotic agromyzid leafminers in Australia, including *L. sativae*, *L. trifolii*, *L. bryoniae* and *L. huidobrensis*, with a focus on *L. sativae*, due to this pest having already established within the far northern quarantine zones of Queensland. Mass-rearing requires strict protocols to ensure synchrony of the leafminer host populations to enable predictable and optimal production of parasitoids. To assist local natural enemy producers to design suitable systems, our recommendations are as follows:

1. The decision to mass-rear parasitoids for release against polyphagous agromyzid leafminer in Australia should be market-driven and should not be considered until the distribution and damage caused by the pests become apparent in Australia.
2. Augmentative control is not expected to be a viable option for outdoor crops. Costs of repeated releases of natural enemies will be high and so their use in outdoor settings would be prohibitively expensive. Instead, growers of outdoor crops should focus on management practices that enhance control via naturally present levels of parasitoid populations. However, because a natural presence of parasitoids cannot be relied upon within closed cropping, here augmentative control may become an important component of pest management. For glasshouse crops where growers are already using augmentative biological control of pests such as thrips, whitefly or mites, releases for biological control of leafminers will need to be integrated into these other systems.

3. The prime target parasitoid for mass-rearing for release against *L. sativae* should be *D. isaea*. Fortuitously, *D. isaea* is already present in Australia. It was first found in Sydney, New South Wales in 2001 and has since been found in Victoria, Tasmania and ACT. A major mass-production issue with *D. isaea* has been its tendency for a biased male sex ratio when being mass-reared.
4. With further research, it may be ultimately possible to manipulate endosymbionts in wasps to develop parthenogenetic (female-only) lines of wasps, such as *D. isaea*, which could dramatically reduce the costs associated with a mass rearing system. The female-only line of *Neochrysocharis formosa* found recently in Victoria should also be investigated further to assess its suitability for control of agromyzids.
5. Additional candidates for mass-rearing within Australia are unclear. *Dacnusa sibirica* is not present in Australia. It is highly unlikely that it could be imported into Australia, given its wide host range, and it would not be well suited to Australian conditions. Instead, it will be valuable to monitor the parasitoid complex attacking *L. sativae* if and when it spreads into different parts of Australia on a range of crops; this would provide a clearer picture of which parasitoids should be investigated further to assess their suitability for use in augmentative biological control. Assessing suitability should include tests of fecundity, host feeding, thermal biology, endosymbiont status, dispersal activity and searching efficiency at low host density. Potential candidates for more detailed study could include *Zagrammosoma latilineatum*, *Cirrospilus brevicorpus, Closterocerus mirabilis, Trigonogastrella parasitica*, and *Opius cinerariae*. It will be important to conduct these tests using Australian populations of *L. sativae* rather than a surrogate host such as *L. brassicae*. The same assessments would need to be made to identify candidates for control of *L. trifolii, L. bryoniae* and *L. huidobrensis* should either of these pests establish.
6. In order to reduce production costs, cold storage of mass-reared *D. isaea* and other potential parasitoids needs to be investigated carefully in order to increase storage life without unduly affecting the performance of the parasitoids. In the longer-term, studies to determine whether the Australian parasitoid species enter diapause or quiescence are needed. The ability to induce and break diapause in these parasitoids would give more flexibility for natural enemy producers.

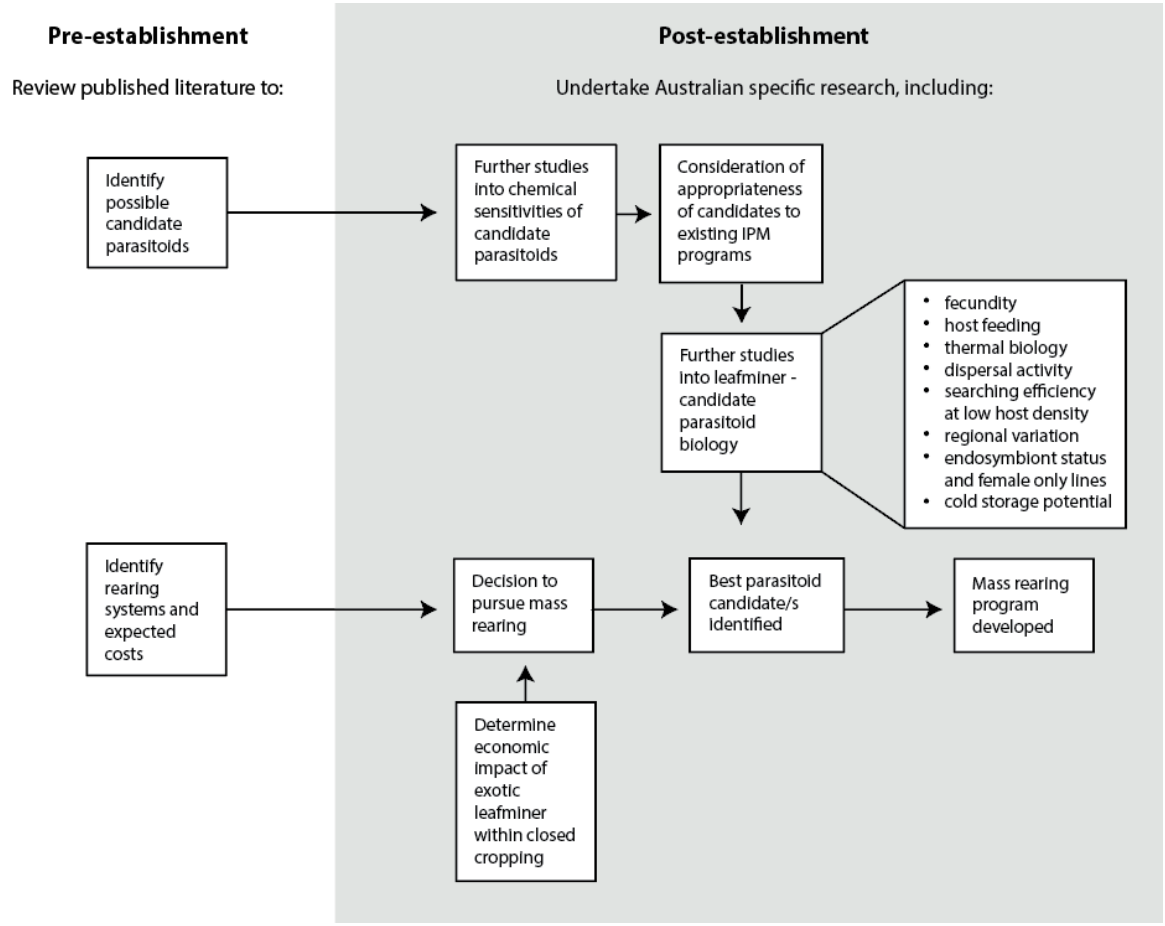


Figure 1. The progression of research that is necessary before a mass rearing program for polyphagous agromyzid leafminers can be developed.

1. Introduction

The vegetable leafminer, *Liriomyza sativae* Blanchard, poses a risk to vegetable and nursery production in mainland Australia since it established in Cape York in 2015 (Ridland *et al.* 2020). There is also a risk of other polyphagous leafminer pests entering and establishing in Australia, particularly *Liriomyza bryoniae* (Kaltenbach), *Liriomyza huidobrensis* (Blanchard) and *Liriomyza trifolii* (Burgess). Problems with polyphagous *Liriomyza* pest species typically result from the destruction of their parasitoid wasps by excessive use of non-selective insecticides. Effective control overseas depends on maximising the impact of natural enemies. Overseas, invading *Liriomyza* populations have frequently been exploited by endemic parasitoids (often found on non-pest agromyzid species), and non-crop hosts have played a role as reservoirs of these parasitoids (Ridland *et al.* 2020).

In protected cropping in Europe, Japan and North America, seasonal inoculative biological control (SIBC) is now used extensively (van Lenteren 2007; van Lenteren *et al.* 2018) for control of the polyphagous agromyzid (Diptera: Agromyzidae) leafminers. Regular releases are needed to ensure that parasitoids are present in the greenhouse as soon as leafminer damage is observed (Onillon 1999; Chow & Heinz 2004; Jacobson 2004; van der Linden 2004). Vegetable crops such as tomatoes, capsicums and cucumbers can withstand relatively high levels of mining damage to leaves before yield is affected. In contrast, mined foliage is unacceptable in potted flowering plants or bedding plants, so it is only in cut flowers or flowering plant crops sold for seed that SIBC has been deployed successfully in ornamental crops (Jones *et al.* 1986; Chow & Heinz 2004). The biological control of *Liriomyza* leafminers was reviewed by Liu *et al.* (2009, 2011) and Ridland *et al.* (2020).

In this review, we focus on mass-rearing techniques used overseas for parasitoids of polyphagous *Liriomyza* spp. to see how these findings could be applied to the augmentative biological control of polyphagous agromyzid leafminers, with a focus on *L. sativae*, in Australia.

2. History of augmentative biological control of *Liriomyza* spp.

Liriomyza bryoniae became a significant pest of tomatoes in north-western European glasshouses in the mid-1970s when growers began using rockwool or nutrient film for growing plants (Wardlow 1984; Minkenberg & van Lenteren 1986; Parrella *et al.* 1999). Previously, the disinfestation of soil during winter killed overwintering puparia, so any leafminer infestations of the crop came in spring or summer from invading flies when the crop was well-established. When this management practice ceased, *L. bryoniae* emerging from the overwintering puparia could infest crops immediately. The arrival of *L. trifolii* into European glasshouses in the late 1970s led to increased problems as growers were unable to control them with conventional insecticides due to insecticide resistance. In many crops, insecticide use was curtailed to allow biological control of greenhouse whitefly, *Trialeurodes vaporarium* (Westwood) by *Encarsia formosa* Gahan (van Lenteren *et al.* 1979). The lack of insecticide pressure enabled two endemic species of braconid parasitoids, *Dacnusa sibirica* Telenga (Hymenoptera: Braconidae: Alysiinae) and *Opius pallipes* Wesmael (Hymenoptera: Braconidae: Opiinae), to be able to overwinter in the glasshouses along with the *L. bryoniae* puparia and thereby provided some early season control of leafminers (Parrella *et al.* 1999). Later in the season, the endemic eulophid ectoparasitoid, *Diglyphus isaea* (Walker), moved into the crop from outside and often controlled leafminer infestations when the use of broad-spectrum insecticides was curtailed (Minkenberg & van Lenteren 1990).

In the 1980s, numerous trials were conducted to improve the level of biological control achieved in tomato glasshouses by observing the patterns of natural colonisation of crops by *L. bryoniae* and its parasitoids (Hendrikse & Zucchi 1979; Woets & van der Linden 1982; Wardlow 1984). The observed variability in results led to trials to test the value of experimental releases of mass-reared parasitoids early in the crop development. There was no systematic selection process for the most appropriate species (or strains) in Europe, rather the endemic species already invading the glasshouses were mass reared. The imported eulophid endoparasitoid, *Chrysocharis oscinidis* Ashmead (then referred to as *C. parksi*), was evaluated in a number of trials. *Chrysocharis oscinidis* was an abundant parasitoid of *L. sativae* in field tomatoes in California (Johnson *et al.* 1980) but provided inconsistent control in trials (Woets & van der Linden 1985; Frijters *et al.* 1986; van der Linden 1986, 1990) and commercial rearing companies then focused on producing *D. isaea* sourced from local populations.

At present, the principal mass-reared parasitoid for release against *Liromyza* spp. is *D. isaea* with additional control provided by *D. sibirica* early in the season in northern Europe where *Liriomyza* spp. overwinter in glasshouses (van Lenteren & Tommasini 1999) (Table 1). Both these species are sold in more than 20 countries and are included in the top nine most important invertebrate biological control agents used in the world in augmentative biological control (van Lenteren 2012). Other parasitoids mass-reared for control of leafminers are *Neochrysocharis formosa* (Westwood) which has been available in Asia since 1990, *Diglyphus begini* (Ashmead) which is reared in Latin America and *Opius pallipes* which is still reared in small numbers in Europe (Cock *et al.* 2010; van Lenteren 2012; van Lenteren *et al.* 2018).

Table 1. Commercial availability of leafminer parasitoids worldwide for augmentative biological control in protected cropping, with region of use, year of first use and market value as per September 2016 (van Lenteren *et al.* 2018). Target crops include tomato, capsicum, cucurbits, chrysanthemum and gerbera.

Reared leafminer parasitoid	**Market Value[a]**	**Region where used**
Diglyphus isaea	L	Europe, Africa North & South, North & Latin America, South America, Asia
Dacnusa sibirica	L	Europe, Africa North, North & Latin America, Asia
Diglyphus begini	M	Latin America
Neochrysocharis formosa	M	Asia
Opius pallipes	S	Europe

[a]Market Value:

L = large (hundred thousand to millions of individuals sold per week)

M = medium (ten thousand to a hundred thousand individuals sold per week)

S = small (hundreds to a few thousand individuals sold per week)

Africa North = North of Sahara; Africa South = South of Sahara; North America = Canada + USA; Latin America = Caribbean, Central and South America.

For successful mass-rearing of parasitoids, tightly synchronised cultures of host leafminer and parasitoids are required. Temperature-development studies for some significant parasitoids of agromyzids, together with their agromyzid hosts (Appendix 1), will provide guidance for timing for host-parasitoid systems.

3. Life history of *Liriomyza* spp.

It is essential to understand the lifecycle of *Liriomyza* spp. when considering attempts to utilise biological control agents in protected cropping. The female fly uses her ovipositor to make fan-shaped feeding punctures in the epidermis from which both female and male flies feed on the leaf exudate. The feeding punctures lead to a characteristic stippling of the leaf, which indicates leafminer presence. In contrast, when the female inserts an egg under the epidermis of the leaf, the oviposition puncture is tubular (Bethke & Parrella 1984; Choi *et al.* 2003).

After hatching, the first instar larva begins to tunnel under the epidermis forming the characteristic mine of the particular species. The third (and final) instar of *L. sativae* exits the mine and falls to the ground where pupariation begins. The puparium of the higher Diptera consists of the sclerotized integument of the third instar larva enclosing the pupa.

After pupation, the adult fly emerges from the puparium early in the photophase. Early morning is the most crucial time in the management of *Liriomyza* spp. because of (i) mating and oviposition, (ii) exiting of the third instar larva from the mine, and (iii) eclosion of the adult flies from the puparia in the soil, all beginning around sunrise (Leibee 1984; Parrella 1987; Petitt 1991). The larval stage only occupies about 31% of the total larval development time (Table 2) and the first, second, and third instars of *L. sativae* occupies an average of 38, 27 and 35% respectively of the total larval development time (Petitt *et al.* 1991). Parasitoids attacking early third instars have only a limited window in the lifecycle of the host.

Table 2. Duration (days) and per cent of total immature development for each life stage of *Liriomyza sativae* at different temperatures (Source: Petitt & Wietlisbach 1994).

Stage	**15°C**		**20°C**		**25°C**	
	days	**%**	**days**	**%**	**days**	**%**
Egg	7.8	19%	6.4	24%	4.4	21%
Larva	12.5	31%	8.7	33%	5.8	28%
Pupa	19.7	49%	11.5	43%	10.2	50%
Total	40.4		26.6		20.6	

4. Life history of parasitoids with mass rearing potential

Idiobiont eulophid parasitoids

Most eulophid wasps (Hymenoptera: Eulophidae) parasitising agromyzid leafminers are idiobionts, meaning they paralyze their hosts before they oviposit. They cause mortality in their agromyzid larval hosts in three ways: (i) host parasitism, (ii) host feeding and (iii) host stinging (where the larva is paralysed but the female wasp does not oviposit or host feed) (Table 3) (Zhang *et al.* 2014; Cheng *et al.* 2017).

Host parasitism. Because the agromyzid larva is paralysed, the food resource for the parasitoid is fixed at the time of oviposition, so eggs are generally laid in or beside the third instar (the oldest stage) to maximise the size of the food resource available for the wasp larva. Ectoparasitoids such *D. isaea*, *Closterocerus mirabilis* Edwards & La Salle and *Zagrammosoma latilineatum* Ubaidillah lay their egg beside the paralyzed larva. After hatching, the parasitoid larva feeds on the paralysed host larva and then pupates beside the dead host larva before emerging from the mine. On the other hand, idiobiont endoparasitoids, such as *Neochrysocharis okazakii* Kamijo or *N. formosa* , also paralyze the host larva but the egg is laid inside the larva, and the parasitoid larva feeds and pupates inside the host larva and emerges from the mine.

Host feeding. The idiobiont parasitoid females emerge with very few eggs, so they need to feed on fluids of hosts to develop their eggs before oviposition. Female wasps locate and puncture smaller larvae in the leaf. After they feed on the punctured larva, it dies. Host feeding is strongly related to wasp fecundity and longevity (Chien *et al.* 2004; Zhang *et al.* 2014; Cheng *et al.* 2017).

Host stinging. Sometimes a female wasp paralyses the agromyzid larva but does not oviposit into or beside the paralysed larva, nor feed on the host's body fluids. However, the stinging of the host will still lead to the death of the larva. In a study with *D. isaea* and *N. formosa*, Xuan *et al.* (2018) hypothesised that killing potential hosts by host stinging was a mechanism to protect their own progeny to avoid eggs being laid by another parasitoid.

Table 3. Lifetime host mortality caused by host feeding, parasitism and host stinging (reported as number of larvae killed) by two eulophid parasitoids of *Liriomyza* spp. (reared at 27 °C).

Parasitoid	**Host**	**Oviposition (∑larvae killed)**	**Host Feeding (∑larvae killed)**	**Host Stinging (∑larvae killed)**	**∑ Mortality (∑larvae killed)**	**Adult longevity (days)**	**Reference**
Neochrysocharis formosa	*Liriomyza sativae*	63	44	20	127	15	Zhang *et al.* (2014)
Diglyphus isaea	*Liriomyza sativae*	126	57	136	319	25	Zhang *et al.* (2014)
Hemiptarsenus varicornis	*Liriomyza trifolii*	134	304	84	522	15	Cheng *et al.* (2017)

For all idiobiont eulophids, the adult wasps emerge from the mine so, inconveniently, natural enemy producers have to sell the wasps as adults which have a very short shelf life, rather than as pupae. Typically, the producers sell bottles of 250 or 500 wasps.

Systems for mass-rearing of several idiobiont eulophid species have been documented [*D. begini* (Parrella *et al.* 1989); *D. isaea* (Maharjan *et al.* 2017); *Hemiptarsenus varicornis* (Girault) (Ozawa *et al.* 2004; Ho & Ueno 2008); *N. formosa* (Hondo *et al.* 2006a; Saleh *et al.* 2010); *N. okazakii* (Tran *et al.* 2007)], but as yet no other species apart from *D. isaea* and *D. sibirica* have been commercialized on a large scale. The large commercial natural enemy producers in Europe, Israel and North America such as Koppert Biological Systems, BioBest Group, BioBee Integrated Crop Systems and Bioline AgroSciences, have reared leafminer parasitoids for many years (Appendix 2) and, understandably, their precise rearing protocols are closely guarded intellectual property (Bolckmans 1999). Protocols for rearing parasitoids of agromyzids are discussed in detail in Section 5.

Koinobiont parasitoids

Braconid parasitoids *Dacnusa sibirica*, *Opius dissitus*, *O. pallipes* and *O. scabriventris*

Four braconid wasps parasitising *Liriomyza* spp., namely *D. sibirica*, *O. dissitus*, *O. pallipes* and *O. scabriventris*, are common koinobiont larval-pupal endoparasitoids overseas. These wasps do not paralyse the host larva at oviposition; the agromyzid larva continues to mine the leaf until pupation. The parasitoid does not begin to develop until the host larva pupates. The wasp will oviposit into any of the three instars of the host larva since the wasp's larva will not begin development until its host begins to pupate. In most cases, the parasitoid can immunosuppress the host and so prevent encapsulation of its egg by the host haemocytes (Strand & Pech 1995). However, there have been several reports where indigenous parasitoids are unable to attack a newly-arrived agromyzid successfully. In Europe, eggs of *O. pallipes*, but not *D. sibirica*, were encapsulated by *L. trifolii* so mass-rearing and release in Europe focused on *D. sibirica* (Minkenberg & van Lenteren 1986). In contrast, the inability of *D. sibirica* to develop in *L. sativae* was considered a significant reason for the dominance of *L. sativae* over *L. trifolii* observed in Japan (Abe *et al.* 2005; Abe 2017).

Techniques were developed for mass-rearing *D. sibirica* and *O. pallipes* (Hendrikse 1980) using *L. bryoniae* on tomato as the host. One-week old puparia could be stored at 5°C. In the USA, mass-rearing and release of *Opius dissitus* for control of *L. sativae* has been undertaken for many years at the EPCOT Center, Florida (Petitt 1993). *Liriomyza sativae* was reared on bush lima beans (*Phaseolus lunatus* L. cv. Henderson) (Petitt & Wietlisbach 1994) and the process of collecting and counting the pupae of *O. dissitus* was mechanised successfully (Petitt *et al.* 1996). In South America, *Opius scabriventris* Nixon is a frequently recorded solitary koinobiont larval–pupal endoparasitoid of *Liriomyza* spp. (including *L. sativae* and *L. huidobrensis*) (Salvo & Valladares 1995). *Opius scabriventris* has recently been imported to Kenya where it has become the second most abundant parasitoid in the mid- and low-elevation fields (Foba *et al.* 2016). The biology of *O. scabriventris* using *L. sativae* as host have been studied in detail in Brazil (Costa-Lima *et al.* 2014, 2019a). *Dacnusa sibirica* is only released in northern Europe in the winter when it is too cold for *D. isaea* to be effective (Helyer *et al.* 2014).

Eucoiline parasitoids

Banacuniculus (=*Ganaspidium*) *utilis* (Beardsley) (Figitidae, Eucoilinae) is a koinobiont larval-endoparasitoid. It was introduced into Hawaii from Texas and has become a dominant parasitoid of *Liriomyza* spp. in Hawaii (Johnson 1987; Petcharat & Johnson 1988). Subsequently, *B. utilis* was introduced into Guam and Tonga where it apparently has become the dominant parasitoid of *L.*

trifolii (Johnson 1993). Unfortunately, there are no confirmed records of eucoilines parasitising agromyzids in Australia (Ridland *et al.* 2020). If such species were found, they should be evaluated for impact on *L. sativae*.

5. Mass-rearing protocols for parasitoids of *Liriomyza* spp.

Mass-rearing protocols have been published for several parasitoids of *Liriomyza* spp. (Parrella *et al.* 1989; Rathman *et al.* 1991; Costa-Lima *et al.* 2017, 2019b; Maharjan *et al.* 2017). The protocols follow a similar process, likely to be applicable for mass-rearing parasitoids for *L. sativae* in Australia:

- maintaining supply of host plants
- exposing host plants to leafminers for a short time to achieve synchrony of leafminer population
- exposure of leafminer-infested plants to parasitoids for a short time to achieve synchrony of parasitoid population
- collection of leafminer puparia to maintain leafminer culture
- maintenance of parasitised leafminers (parasitoid species dependent)
- collection and packaging parasitoids from parasitised leafminers

Parrella *et al.* (1989) devised and costed an effective process for mass-rearing *Diglyphus begini* for seasonal inoculative release against *L. trifolii* on glasshouse chrysanthemum, *Chrysanthemum × morifolium*. *Diglyphus begini* is endemic to the USA and has very similar biological characteristics to *D. isaea*. Modelling of the rearing system to optimise production (Heinz *et al.* 1993; Heinz 1996) provided valuable insights into the process and should be followed in Australia. Maharjan *et al.* (2017) outlined a very similar process for mass-rearing *D. isaea* on *L. huidobrensis* reared on the faba bean, *Vicia fabae*. Rathman *et al.* (1991) described and costed a system for mass-rearing *Banacuniculus* (=*Ganaspidium*) *utilis* (Beardsley) (Figitidae, Eucoilinae) on *L. trifolii* reared on lima beans. Unlike *Diglyphus* spp., *B. utilis* pupates inside the *Liromyza* puparia. Costa-Lima *et al.* (2017, 2019b) (Appendix 3) described protocols for mass-rearing *O. scabriventris,* which included details of collection chambers for collecting large numbers of final stage larvae as they exit leaves before pupation.

The rearing system outlined by Parrella *et al.* (1989), and modified by others, serves as a good basis for potential mass-rearing operations in Australia. This system consisted of the three components, plant production, leafminer production and parasitoid production, which are each discussed in more detail in the following sections.

Plant production

Host plants used were commercially rooted chrysanthemum cuttings, planted weekly – four cuttings per pot (12.7 cm diameter). Plant buds were pinched out after 2 weeks and had sufficient leaf area for infestation after one month in the glasshouse (16:8 [L:D] photoperiod; 21–27°C). Chrysanthemums were chosen as the host plant rather than quicker growing beans in order to condition the parasitoids to the target crop (but without experimental evidence to support this decision, see (Hands 2013) for *L. bryoniae*/*D. isaea* system with French bean, *Phaseolus vulgaris*, and tomato, *Solanum lycopersicum*, as plant host).

Leafminer production

Adult *L. trifolii* were kept in cages (30.5 cm high, 66.0 cm wide, 78.7 cm long). About 250 flies (sex ratio 1:1) were released into the cage daily, with the populations maintained at between 1,000 and 2,000 flies.

Chrysanthemums (36 plants) were exposed to the flies for 2 h (usually in the morning) in each cage. The short exposure time is the key to ensuring that larval development was synchronous. After exposure, plants were taken back to a glasshouse.

When most larvae reached late second instar or early third instar (about 5 days under the glasshouse conditions), the plants were ready either for exposure to parasitoids or for producing more leafminers.

About 1/3 of the infested plants were used for maintenance of the *L. trifolii* culture. The foliage was cut off at the soil line and placed on a wire mesh screen over a sand-filled tray to collect the exiting third instar larvae. Puparia were sifted from the sand and the numbers estimated by weighing.

Parasitoid production

Adult *D. begini* were held in glass-topped sleeve cages (43 cm high, 95 cm wide, 49.5 cm long). Parasitoid numbers were kept between 200 and 300, with 50 wasps (sex ratio 1:1) being added each day. Infested cuttings were exposed to the parasitoids for 24 hours. After the exposure, the pots were watered then held on custom tip racks for 2 days with the plants extending over sand-filled trays to collect any unparasitized exiting larvae. In this system, the mean number of unparasitized larvae exiting the leaves was 47, while on average 99 parasitoids were produced per pot. This suggests that there is scope for increasing the efficiency of the parasitism process. The plants were then taken back to another glasshouse room for development. After 10 days (just before the emergence of parasitoids from the pupae in the leaves), the leaves were cut at the soil line and placed in a parasitoid cage for the emergence of the wasps. Wasps were collected in small vials with an aspirator for releases or colony maintenance.

The breakdown of time requirements for the three components to produce 5,000 *D. begini* parasitoids, 4,000 *D. isaea* and 5,000 *Banacuniculus (=Ganaspidium) utilis*, respectively are given in Tables 4, 5 & 6. With *Diglyphus* spp., the most time-consuming activity was the final aspiration and counting of the parasitoids. Potential time-savings here would be to use a photo-optic collection device (Rathman *et al.* 1991; Saleh *et al.* 2010) and a photo-electronic counting system. Rathman *et al.* (1991) noted that aspiration time required for *G. utilis* was considerably less than for *Diglyphu*s spp. since the sex ratio for *B. utilis* was very stable.

The production cycle for both *Diglyphus* species would be shortened if *L. trifolii* and *L. huidobrensis* were reared *on Phaseolus vulgaris,* as is used for rearing *D. isaea* on *L. bryoniae* (Hands 2013) and *N. formosa* (Saleh *et al.* 2010). Beans with two true leaves and with the apex pinched out were ready for infestation with *L. huidobrensis* after 12–15 days (Saleh *et al.* 2010). Hands (2013) observed that *P. vulgaris* retained their leaves for longer after infestation with *L. bryoniae* than did faba beans, *Vicia faba*. Hands (2013) also found that *D. isaea* reared on beans infested with *L. bryoniae* were as effective against *L. bryoniae* on tomato as were *D. isaea* reared on tomato infested with *L. bryoniae*.

Rathman *et al.* (1991) used bush lima bean, *Phaseolus lunatus*, to rear *L. trifolii*. Beans were grown in black plastic mulch bags (40 beans per bag; 90% germination rate) containing about 60 g of

vermiculite. No fertiliser was used, and the beans were ready for infestation with *L. trifolii* after 10–11 days. This was much faster than with chrysanthemum and greatly reduced costs.

Table 4. Hours of labour required per day in mass production of *Diglyphus begini* using chrysanthemum (*Chrysanthemum × morifolium*) as a host plant and *Liriomyza trifolii* as the host agromyzid (Parrella *et al.* 1989)

Procedure	Step duration (days)	Time required per 5,000 parasitoids[1] (h:min:s)
Potting and maintaining plants	1–30	0:44:13
Exposing plants to leafminers	30	0:27:29
Exposing plants to parasitoids	34	0:46:43
Plants placed on tip racks (watered first)	35	0:12:18
Plant and leafminer colony maintenance, while parasitoids develop	37	0:03:15
Cutting plants and placing in cage	48	0:17:04
Aspirating parasitoids from cage	50	3:42:48
Total		**6:13:43**

[1]50 pots, with 100 parasitoids per pot

Table 5. Hours of labour required per day in mass production of *D. isaea* using faba bean (*Vicia fabae*) as a host plant and *Liriomyza huidobrensis* as the host agromyzid (Maharjan *et al.* 2017)

Procedure	Step duration (days)	Time required per 4,000 parasitoids[1] (h:min:s)
Potting and maintaining plants	1–20	0:44:30
Exposing plants to leafminers	20	0:28:24
Exposing plants to parasitoids	25	0:46.18
Plants placed on tip racks (watered first)	26	0:17:12
Plant and leafminer colony maintenance, while parasitoids develop	28	0:03:30
Cutting plants and placing in cage	40	0:18:06
Aspirating parasitoids from cage	43	4:00:30
Total		**6:38:24**

[1]50 pots, with 80 parasitoids per pot

Table 6. Hours of labour required per day in mass production of *Ganaspidium utilis* using lima bean, *Phaseolus lunatus*, as a host plant and *Liriomyza trifolii* as the host agromyzid (Rathman *et al.* 1991)

Procedure	Step duration (days)	Time required per 5,000 parasitoids[1] (h:min:s)
Potting and maintaining bean plants (14 bags[a] per day	1–10	0:60:00
Exposing plants to leafminers (14 bags)[b]	11	0:15:00
Exposing plants to parasitoids (12 bags)[b]	15	0:25:00
Pinching leaves for leafminers (2 bags)	16	0:15:00
Removing plants/pinching leaves for parasitoids	17	0:60:00
Transferring dry leaves to cartons and collecting puparia (*Liriomyza*)	18–23	0:15:00
Transferring dry leaves to cartons and collecting puparia (parasitoids)	17-	0:40:00
Collecting parasitoids	39–48	0:30:00
Collecting *Liriomyza*	36–38[c]	0:10:00
Total		**4:30:00**

[1]50 pots, with 80 parasitoids per pot

[a]40 bean seeds per black plastic mulch bag containing 59 g vermiculite

[b]plants were sprayed with a 10:1 solution of water and molasses just before exposure to leafminers and parasitoids. The molasses provided a carbohydrate source for the adult leafminers and parasitoids.

[c]Leafminer emergence estimated using temperature-development equations listed by Parrella (1987)

6. Constraints to commercial mass rearing

High production costs

Unfortunately, *D. isaea* is very expensive to rear with the retail unit cost ranging from USD 0.32 to USD 0.55 per wasp (assuming 1:1 sex ratio, the unit cost ranges from USD 0.64 to USD 1.09 per ♀ wasp). Typically, 3–5 weekly releases are made in glasshouses per crop. With release rates of between 0.25–1 wasps per m^2 or 2,500–10,000 wasps per ha (USD 750 to USD 3,000 per hectare per release using USD 0.30 per wasp) (Table 7), it will not be economical to use commercially-reared *D. isaea* augmentatively in the open field situation.

Table 7 Indicative release rates of *Diglyphus isaea* [Miglyphus®] outlined by Koppert (2000)

Miglyphus®	**light infestation**	**heavy infestation**
Rate m²/unit (500 wasps)	2,000	500
Rate wasps/m²	0.25	1
Rate wasps/ha	2,500	10,000
Interval (days)	7	7
Frequency	min. 3x	min. 3x
∑wasps/ha	7,500	30,000
Remarks	if >1 larva/10 plants	if >1 larva/3 plants

Storage

Production costs remain high since the shelf life for the adult wasps is very short. Cold storage of insect parasitoids affects a wide range of fitness-related traits (Colinet & Boivin 2011), so detailed research is needed to ensure that cold storage protocols are optimised for any mass-reared beneficial arthropod. With mass-reared *D. isaea*, cold storage has been investigated in an attempt to increase storage life and so cater to unpredictable demand. Burgio & Nicoli (1994) observed that *D. isaea* wasps stored at 4°C for 120 days (and fed with honey and sucrose solution solidifed in agar) had higher survival and fecundity than wasps stored at 10°C. In contrast, Steinberg *et al.* (1991) stored *D. isaea* at 15°C, 20°C and 26°C for a month and found that optimum performance of the wasps was at 20°C. Interestingly, the sex ratio (% males) was reduced to 29% at 26 °C compared with 54% at 15°C and 44% at 20°C (Steinberg *et al.* 1991). Also, *D. isaea* kills many hosts for food, so the yield of parasitoids is diminished. For example, at 27°C, *D. isaea* ♀ had a mean lifetime fecundity of 126 eggs but caused the death of 319 host larvae (through oviposition, host feeding and host stinging) (Zhang *et al.* 2014).

For maximising fertility of *H. varicornis* for mass rearing, Chien *et al.* (2005a) found that suitable conditions for storing wasps were, in priority, (i) female wasps at 15℃ for 10 days, (ii) female wasps at 15℃ for 20-30 days or 25℃ for 10-15 days and (iii) 1-day-old pupae at 10℃ for 1-4 weeks. For field releases, where host-killing was more important than fertility, suitable conditions for storing wasps were, in priority, (i) female wasps at 15 ℃ for 10-30 days or 25℃ for 10-15 days, and (ii) female wasps at 15℃ for 40 days or 1-day-old pupae at 10℃ for 1-4 weeks.

Biased sex ratio

A further problem in mass-rearing *D. isaea* is the production of excess males. For example, Heimpel & Lundgren (2000) recorded 77% of males in samples obtained from commercial insectaries. Assuming a unit cost of USD 0.32 per wasp, this equates to a cost per ♀of > USD 1.30. While this can be partially overcome with the manipulation of host sizes offered to the wasps (Chow & Heinz 2005, 2006), this adds complexity to the rearing system.

Management issues

There can be foliage damage before the parasitoids control the leafminer population. In glasshouse chrysanthemum crops in the Netherlands, Zuijderwijk *et al.* (2008) observed that preventative introductions (five weekly releases) of *D. isaea* into the crop still required an initial treatment with cyromazine (an insect growth regulator) to reduce initial numbers of leafminer larvae. However, they noted that multiple applications of cyromazine prevented the establishment of *D. isaea*. They also observed difficulties in establishing *D. isaea* in winter when growth lights were used because the adult parasitoids were being attracted to the growth lights and killed. Jacobson (2008) noted that costs could be reduced if parasitoids produced early in the season could be re-distributed to other parts of the glasshouse later in the season, effectively creating a within-crop rearing system. This system has also been used successfully in Kenya (Lee 2004). Helyer *et al.* (2014) suggested that growers can harvest heavily mined and parasitised milk thistle, *Sonchus arvensis*, plants from outside their crop, as a source of *D. isaea* for their crops. This would require placing the leaf material in a small cage with a mesh size less than 625µ (Bethke & Paine 1991; Bethke *et al.* 1994) to retain the leafminer flies but allow the parasitoids to pass through the mesh and out into the glasshouse.

Management issues for other pests will also act as a driver for adopting leafminer biological control. For example, in Canada, use of biological control in ornamental crops has increased greatly in recent years, largely driven by the difficulties in controlling western flower thrips, *Frankliniella occidentalis* Pergande, because of resistance to spinosad (Murphy *et al.* 2011). They noted that Canadian chrysanthemum and gerbera growers were largely relying on release of *D. isaea* because chemicals registered for control of *L. trifolii* were ineffective due to increasing resistance issues.

> "The move to biocontrol of thrips resulted in a ripple effect in which other pests had to be controlled biologically as well, since there is limited availability of pesticides compatible with the predatory mites used for thrips control. Of particular importance is the leafminer *Liriomyza trifolii* in chrysanthemum and gerbera. Registered active ingredients in Canada include permethrin, abamectin and cyromazine, none of which is currently offering effective control. A number of chrysanthemum growers in particular are using *Diglyphus* for control of leafminer, however, it is very sensitive to pesticides, even when used cautiously, and some growers have had difficulty in maintaining long term control of this pest." Murphy *et al.* (2011)

7. Potential solutions to current constraints

Use of thelytokous strains

A potential solution to improve the economics of mass-rearing parasitoids for augmentative control is to rear a female-only strain (Ramirez-Romero *et al.* 2012) with fecundity and longevity similar to a bisexual population (Sivinski 2013). Thelytokous strains of *N. formosa* were associated with a *Rickettsia* sp. infection in Japan (Hagimori *et al.* 2006, 2008; Adachi-Hagimori & Miura 2008; Adachi-Hagimori *et al.* 2011) and China (Yang *et al.* 2017). However, the Japanese thelytokous strain does not appear to have been widely utilised, with growers relying on *D. isaea* and *D. sibirica* imported from Europe (Abe 2017).

Recently, a newly described thelytokous species, *Diglyphus wani* Liu, Zhu & Yefremova, was found to be common in Chinese horticultural regions (Ye *et al.* 2018), providing a new candidate

parasitoid with favourable life-history traits for mass rearing. The mechanism underlying thelytoky in this species has not yet been determined. The authors detected a *Rickettsia* sp. in a relatively high frequency, but females continued to produce only female offspring five generations after tetracycline treatment to remove this endosymbiont bacteria (Ye *et al.* 2018).

Quality control of mass-reared natural enemies

It is difficult to maintain high quality, mass-reared natural enemies. Unintended selection within populations for traits favouring high levels of production in a mass-rearing facility, such as reduced ability to disperse and high parasitism rates at high host density, can reduce optimal effectiveness in the field, where high dispersal activity and high parasitism levels at low host density are preferred (van Lenteren & Tommassini 1999; van Lenteren 2003). The rapid rate of laboratory adaptation in laboratory cultures of insects was highlighted by Hoffmann & Ross (2018) who stressed the need to rear field-release populations under conditions that minimize the reduction of field fitness or ensure that colonies for field releases are renewed regularly from field material. A pro-active approach to utilising local parasitoid species would be to exploit intraspecific variation in biocontrol traits by selective breeding, as outlined by Kruitwagen *et al.* (2018) who used biological control of *D. suzukii* as a case study. The four-step process they proposed was to (i) assess interspecific variation in biocontrol traits, (ii) assess intraspecific variation in biocontrol traits of chosen candidate species, (iii) determine the genetic an environmental factors explaining the variation in biocontrol traits, and (iv) exploit intraspecific variation by selective breeding.

8. Australian situation

Begum *et al.* (2017) reviewed the history of the Australian commercial production of invertebrate biological control agents and highlighted some of the difficulties facing the small industry, such as the high costs of developing new biological control agents and limited industry take up. With this background, a decision to mass-rear parasitoids for release against *L. sativae* (or *L. trifolii*, *L. bryoniae* or *L. huidobrensis*) should not be considered until the distribution and damage caused by the pests become apparent in Australia. Costs of repeated releases of natural enemies will be high and so their use in augmentative releases in outdoor crops would be prohibitively expensive.

If exotic leafminers do become a problem in glasshouse crops and control by insecticides becomes disruptive to augmentative biological for pests such as thrips, whitefly or mites, then biological control of leafminers would be needed to avoid disruption of the established program.

It will be important to monitor the parasitoid complex of *L. sativae* if and when it spreads into different parts of Australia on a range of crops, to get a clearer picture of which parasitoids should be investigated further for rearing. This would also be true for *L. trifolii, L. bryoniae* and *L. huidobrensis* should any of these pests establish in Australia. For *L. sativae*, it is likely that the Australian situation will be comparable to the southern European situation where this pest can overwinter more successfully in the open than in northern Europe. Until the Australian specific interactions between *L. sativae* and parasitoids become clear, the best recommendations for mass rearing targets are as follows:

Diglyphus isaea

Based on the long-term experiences in Europe and the USA, we recommend that the prime target parasitoid species for mass-rearing for release against *L. sativae* be *D. isaea.*

Fortuitously, *D. isaea* is already present in Australia. It was first found in Sydney, NSW, in 2001 (ALA 2020) and has since been found in Victoria (Bjorksten *et al.* 2005), Tasmania and ACT (Lambkin *et al.* 2008) frequently parasitising *Chromatomyia syngenesiae* infesting sow thistle, *Sonchus oleraceus*, and *Phytomyza plantaginis* infesting *Plantago lanceolata*. The species has already been cultured in the laboratory in Australia (Bjorksten & Robinson 2005) using *L. brassicae* reared on Chinese cabbage, *Brassica rapa* ssp. *pekinensis*, at 22°C. Under these conditions, egg-adult development times were 13 days for *D. isaea* ♀ and 12 d for *D. isaea* ♂.

As with other adventive biological agents in Australia such as the aphid parasitoid, *Diaeretiella rapae* (Baker *et al.* 2003), it is likely that the Australian founding population(s) of *D. isaea* will have reduced genetic diversity compared to populations worldwide. It will be important to test the biological characteristics of populations *of D. isaea* from different parts of Australia to ensure that the most suitable population is chosen for mass-rearing (Lommen *et al.* 2017).

We recommend that *Phaseolus vulgaris* or *P. lunatus* be used as a host plant for *L. sativae,* but testing of a range of cultivars should be undertaken to choose the most appropriate line for rearing *L. sativae* and *D. isaea.*

Neochrysocharis formosa

In preliminary studies at the University of Melbourne, it appears that a female-only strain of *N. formosa* has been found in the field (Marianne Coquilleau and Xuefen Xu, personal communication). Once it is cultured, its potential as a biological control agent needs to be assessed, as well as the mechanism for its thelytoky.

Opius spp.

Until *L. sativae* becomes widely established in Australia, its capacity to encapsulate eggs of the three endemic species of *Opius* known to attack agromyzids in Australia (Belokobylskij *et al.* 2004) remains unknown. However, three adventive agromyzid species in Australia (*Chromatomyia syngenesiae*, *Liriomyza brassicae* and *Phytomyza plantaginis*) are all parasitised by the endemic *Opius* spp., suggesting that encapsulation has not been an issue in these relatively new associations (Ridland *et al.* 2020). *Opius cinerariae* Fischer has been reared for laboratory experiments in Australia using *L. brassicae* as host agromyzid by Lardner (1991).

If an *Opius* sp. is found parasitising *L. sativae* in significant numbers, its biology should be investigated with a view to evaluating it as a complementary biological control agent to *D. isaea*.

Other parasitoid species

Endemic eulophid species such as *Zagrammosoma latilineatum*, *Closterocerus mirabilis* and *Cirrospilus brevicorpus* and the pteromalid, *Trigonogastrella parasitica,* are often recorded parasitising agromyzids in Australia. In fact, *Z. latilineatum* and *C. mirabilis* have also already been observed parasitizing *L. sativae* within the Torres Strait, as have parasitoids of the genus *Cirrospilus* (Elia Pirtle unpublished data). It would therefore be worth investigating the biology of

these species further to assess their potential use for mass rearing. There may be ways of improving mass-rearing through identification of thelytokous strains or manipulating parasitoids with endosymbionts to achieve novel thelytokous strains.

The suitability of these strains should be assessed through tests of fecundity, host feeding, thermal biology, endosymbiont status, and both searching efficiency and parasitism efficiency at low host density (Minkenberg & van Lenteren 1990). There is also the opportunity to employ modern genetic and genomic techniques to improve the efficacy of existing and new biocontrol agents (Leung *et al.* 2020).

Conclusions

- Overseas, *D. isaea* has been shown repeatedly to be a very effective augmentative biological control agent for *Liriomyza* spp. While local mass-rearing of *D. isaea* is technically feasible using protocols developed overseas, it is likely to be expensive to implement. It would almost certainly be uneconomic for control of *L. sativae* through augmentation in open field production. However, costs could be reduced if the sex ratio of this organism can be manipulated to more strongly biased towards females, such as through the use of endosymbionts that generate thelytoky.
- Until *L. sativae* is shown to be a major problem in Australian glasshouse crops that cannot be managed with the insecticides currently available to growers under permit, parasitoids are unlikely to be a viable option for most growers, unless they were undertaking augmentative biological control for other pests.
- It would be valuable for the benefits of augmentative biological control of leafminers to be assessed in glasshouses and this would require close co-operation between the biological control agent producers, growers and researchers.
- *Dacnusa sibirica* is not present in Australia. It is highly unlikely that it could be imported into Australia, given its wide host range, and it would not be well suited to Australian conditions.
- It will be valuable to monitor the parasitoid complex of *L. sativae* if and when it spreads into different parts of Australia on a range of crops, to get a clearer picture of which parasitoids should be investigated further to assess their potential suitability for use in augmentative biological control. Assessing suitability should include tests of fecundity, host feeding, thermal biology, endosymbiont status, dispersal activity and searching efficiency at low host density. Potential candidates for more detailed study could include *Zagrammosoma latilineatum*, *Cirrospilus brevicorpus, Closterocerus mirabilis, Trigonogastrella parasitica*, and *Opius cinerariae*. It will be important to conduct these tests using Australian populations of *L. sativae* rather than a surrogate host such as *L. brassicae*.
- In order to reduce production costs, cold storage of mass-reared *D. isaea* and other potential parasitoids needs to be investigated carefully in order to increase storage life without unduly affecting the performance of the parasitoids. In the longer-term, studies to determine whether the Australian parasitoid species enter diapause or quiescence are needed. The ability to induce and break diapause in these parasitoids would give more flexibility for the natural enemy producers.
- If candidate species such as *D. isaea* are mass-reared in Australia, it will be important to measure the variability in demographic performance and dispersal behaviour between regional populations in Australia. There are IOBC quality control guidelines for the production of natural enemies (van Lenteren *et al.* 2003), including *D. sibirica* and *D. isaea*

(Appendix 4) which should be adapted for Australian requirements and species selected for mass-rearing.

Appendices

Appendix 1 Temperature development data for *Hemiptarsenus varicornis*, *Diglyphus isaea*, *Neochrysocharis formosa*, *N. okazakii*, *Chrysocharis vonones*, *Opius cinerariae*, *O. caricivorae* and *O. scabriventris* on a range of agromyzid hosts (Ridland *et al.* 2020)

Parasitoid species	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (D°)	Thermal Optimum T_{opt} (°C)	Host Plant	Reference
Hemiptarsenus varicornis [Australia]		*Liriomyza brassicae*	9.4	192.3		rape (*Brassica napus*)	Lardner (1991)
		Liriomyza brassicae	8.4	307		rape (*Brassica napus*)	Lardner (1991)
Hemiptarsenus varicornis	♀	*Liriomyza bryoniae*	8.4	149.5		kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis	♂	*Liriomyza bryoniae*	8.2	146.2		kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis		*Liriomyza trifolii*	11.6 ±	156 ±		kidney bean (*Phaseolus vulgaris*)	Chien *et al.* (2004)
Hemiptarsenus varicornis	♀	*Liriomyza trifolii*	11.8	135.1		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Hemiptarsenus varicornis	♂	*Liriomyza trifolii*	11.7	126.6		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Hemiptarsenus varicornis	♀	*Liriomyza trifolii*	8.5	150.8		kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
Hemiptarsenus varicornis	♂	*Liriomyza trifolii*	8.9	137.6		kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1997)
		Liriomyza trifolii	9.5	256.4		kidney bean (*Phaseolus vulgaris*)	Saito *et al.* (1995)
Diglyphus isaea	♀	*Chromatomyia syngenesiae*	12.8 ± 0.12*	154.6 ± 5.92*		lettuce (*Lactuca sativa*)	Cheah (1987)
Diglyphus isaea	♂	*Chromatomyia syngenesiae*	12.9 ± 0.10*	152.4 ± 6.65*		lettuce (*Lactuca sativa*)	Cheah (1987)
	♀	*Chromatomyia syngenesiae*	12.0 ± 0.06*	207.0 ± 6.17*		lettuce (*Lactuca sativa*)	Cheah (1987)
	♂	*Chromatomyia syngenesiae*	11.6 ± 0.07*	211.0 ± 7.30*		lettuce (*Lactuca sativa*)	Cheah (1987)
Diglyphus isaea	♀	*Liriomyza huidobrensis*	9.3	165.0	32.6†	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)

Parasitoid species	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (D°)	Thermal Optimum T_{opt} (°C)	Host Plant	Reference
Diglyphus isaea	♂	*Liriomyza huidobrensis*	9.3	157.4	31.0†	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
		Liriomyza huidobrensis	8.1	270.3	25.0†	kidney bean (*Phaseolus vulgaris*)	Lanzoni *et al.* (2002)
Diglyphus isaea	♀	*Liriomyza sativae*	9.4	161.3	33.6†	cucumber (*Cucumis sativa*)	Haghani *et al.* (2007b)
Diglyphus isaea	♂	*Liriomyza sativae*	9.2	153.8	33.5†	cucumber (*Cucumis sativa*)	Haghani *et al.* (2007b)
		Liriomyza sativae	10.2	250.0	36.2†	cucumber (*Cucumis sativa*)	Haghani *et al.* (2007a)
Diglyphus isaea	♀	*Liriomyza trifolii*	9.2	161.8	33.3†	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
Diglyphus isaea	♂	*Liriomyza trifolii*	9.5	151.4	32.3†	kidney bean (*Phaseolus vulgaris*)	Bazzocchi *et al.* (2003)
		Liriomyza trifolii	10.5	232.6	29.6†	kidney bean (*Phaseolus vulgaris*)	Lanzoni *et al.* (2002)
Diglyphus isaea (native)	♀	*Liriomyza trifolii*	8.5	185.2		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Diglyphus isaea (native)	♂	*Liriomyza trifolii*	8.4	172.4		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Diglyphus isaea (introduced)	♀	*Liriomyza trifolii*	9.1	175.4		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Diglyphus isaea (introduced)	♂	*Liriomyza trifolii*	8.8	169.5		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Diglyphus isaea	♀	*Liriomyza bryoniae*	8.8	175.4		tomato (*Solanum lycopersicum*)	Minkenberg (1989)
Diglyphus isaea	♂	*Liriomyza bryoniae*	7.9	158.7		tomato (*Solanum lycopersicum*)	Minkenberg (1989)
		Liriomyza bryoniae	8.1	295.9		tomato (*Solanum lycopersicum*)	Minkenberg & Helderman (1990)
Diglyphus isaea	♀	*Liriomyza trifolii*	8.7	166.7		tomato (*Solanum lycopersicum*)	Minkenberg (1989)
Diglyphus isaea	♂	*Liriomyza trifolii*	9.0	166.7		tomato (*Solanum lycopersicum*)	Minkenberg (1989)
		Liriomyza trifolii	9.1	264.6		tomato (*Solanum lycopersicum*)	Minkenberg (1988)
Neochrysocharis formosa		*Liriomyza sativae*	12.1	197.3		kidney bean (*Phaseolus vulgaris*)	Song *et al.* (2005)

Parasitoid species	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (D°)	Thermal Optimum T_{opt} (°C)	Host Plant	Reference
Neochrysocharis formosa		*Liriomyza sativae*	14.3	154.2		kidney bean (*Phaseolus vulgaris*)	Wang *et al.* (2009)
Neochrysocharis formosa		*Liriomyza trifolii*	13.0 ± 0.5*	165 ± 12*		kidney bean (*Phaseolus vulgaris*)	Chien *et al.* (2005b)
		Liriomyza trifolii	8.7 ± 0.9*	294 ± 32*		kidney bean (*Phaseolus vulgaris*)	Chien & Ku 1996
Neochrysocharis formosa	♀	*Liriomyza trifolii*	9.7	227.3		kidney bean (*Phaseolus vulgaris*)	Hondo *et al.* (2006)
Neochrysocharis okazakii		*Liriomyza sativae*	8.1	188.7		bean	Chen & Ye (2002)
Neochrysocharis okazakii	♀	*Liriomyza chinensis*	11.3	172.4		onion (*Allium fistulosum*)	Tran *et al.* (2012)
Neochrysocharis okazakii	♂	*Liriomyza chinensis*	11.5	166.7		onion (*Allium fistulosum*)	Tran *et al.* (2012)
		Liriomyza chinensis	11.4	312.5		onion (*Allium fistulosum*	Tran *et al.* (2007)
Neochrysocharis okazakii	♀	*Liriomyza trifolii*	11.3	178.6		kidney bean (*Phaseolus vulgaris*)	Tran *et al.* (2012)
Neochrysocharis okazakii	♂	*Liriomyza trifolii*	11.6	166.7		kidney bean (*Phaseolus vulgaris*)	Tran *et al.* (2012)
Neochrysocharis okazakii		*Liriomyza trifolii*	11.3 ± 0.9*	192 ± 26*		kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2009)
Chrysocharis vonones		*Liriomyza sativae*	7.4	246.3		cowpea (*Vigna unguiculata*)	Costa-Lima *et al.* (2014)
		Liriomyza sativae	7.3	308.6		cowpea (*Vigna unguiculata*)	Costa-Lima *et al.* (2009)
Opius cinerariae [Australia]	♀	*Liriomyza brassicae* I instar	9.1	270.3		rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♂	*Liriomyza brassicae* I instar	9.1	250.0		rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♀	*Liriomyza brassicae* II instar	9.2	263.2		rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♂	*Liriomyza brassicae* II instar	9.4	238.1		rape (*Brassica napus)*	Lardner (1991)
Opius cinerariae [Australia]	♀	*Liriomyza brassicae* III instar	7.9	294.1		rape (*Brassica napus)*	Lardner (1991)

Parasitoid species	Sex	Host Agromyzid	Lower Threshold (°C)	Thermal Constant (D°)	Thermal Optimum T_{opt} (°C)	Host Plant	Reference
Opius cinerariae [Australia]	♂	*Liriomyza brassicae* III instar	7.9	270.3		rape (*Brassica napus)*	Lardner (1991)
Opius caricivorae		*Liriomyza huidobrensis*	7.03 ± 0.39*	286 ± 20*		kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2012)
		Liriomyza huidobrensis	7.6 ± 0.3*	278 ± 14*		kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2008)
Opius caricivorae		*Liriomyza sativae*	7.61 ± 0.46*	263 ± 17*		kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2012)
		Liriomyza sativae	11.1 ± 0.2*	236 ± 7*		kidney bean (*Phaseolus vulgaris*)	Chien & Chang (2007)
Opius scabriventris		*Liriomyza sativae*	7.3	257.1		cowpea (*Vigna unguiculata*)	Costa-Lima *et al.* (2014)

*Standard error of mean calculated using method of Campbell *et al.* (1974)

†Thermal optimum from Logan-6 equation used to describe the non-linear relationship between temperature and rate of development

Appendix 2 Links to Major Producers of *Diglyphus isaea* and *Dacnusa sibirica*

Koppert Biological Systems

https://www.koppert.com/miglyphus/

https://www.koppert.com/minusa/

https://www.koppert.com/challenges/leaf-miners/

Biobest Group

https://www.biobestgroup.com/en/biobest/products/biological-pest-control-4463/beneficial-insects-and-mites-4479/diglyphus-system-4630/

https://www.biobestgroup.com/en/biobest/products/biological-pest-control-4463/beneficial-insects-and-mites-4479/dacnusa-system-12961/

https://www.biobestgroup.com/en/news/biobest%E2%80%99s-dacnusa-diglyphus-combination%3A-a-grower-s-perspective

https://www.biobestgroup.com/en/news/biobest-offers-complete-leafminer-control-strategy

BioBee Integrated Crop Systems

https://www.biobee.com/biological-ipm/solutions/biodiglyphus/

https://www.biobee.com/biological-ipm/pests/leaf-miners/

Bioline AgroSciences Ltd., England

https://www.biolineagrosciences.com/products/digline/

https://www.biolineagrosciences.com/pests/leafminer/

https://www.biolineagrosciences.com/pests/leafminer/

Bugs for Growers

https://blog.bugsforgrowers.com/wasps/control-four-leafminers-with-mini-wasp-diglyphus-isaea/

Rincon-Vitova Insectaries

http://www.rinconvitova.com/leafminer%20etc.htm

Appendix 3 Mass-rearing system for *Opius scabriventris*

Costa-Lima TC, Geremias LD, Begiato AM et al. (2017) Sistema de criação de parasitoide de mosca-minadora. Embrapa Semiárido, Petrolina. Circular Téchnica 116. 8 pp.
https://www.infoteca.cnptia.embrapa.br/infoteca/bitstream/doc/1078329/1/CTE116.pdf

Leafminer parasitoid breeding system – Google translation 2020–08–12; edited by PM Ridland

Introduction
Leafmining flies of the genus *Liriomyza* (Diptera: Agromyzidae) are important pests of many horticultural crops, with three *Liriomyza* species with worldwide distribution: *L. trifolii* (Burgess), *L. sativae* Blanchard (Figure 1a) and *L. huidobrensis* (Blanchard) (Costa-Lima *et al.* 2015; Murphy & LaSalle 1999).

Because the selection of insecticide-resistant populations is common in leafminers, interest in the use of biological control is growing (Parrella *et al.* 1999). Since the 1980s, *Liriomyza* parasitoids [*Diglyphus isaea* (Walker), *Dacnusa sibirica* Telenga and *Opius pallipes* Wesmael] have been marketed, mainly in Europe and North America, directed mainly to protected crops (European and Mediterranean Plant Protection Organization 2016; White & Johnson 2016). Despite the publication of methods for the breeding of *Liriomyza* parasitoids (Hendrikse 1980; Parrella *et al.* 1989; Saleh *et al.* 2010), insect rearing companies do not provide information on the breeding techniques used (Lenteren & Woets 1988).

In Brazil, there are still no companies that sell biological control agents for the flies. However, in recent studies, the potential of *L. sativae* parasitoids collected in Brazil, such as *Opius* (= *Phaedrotoma*) *scabriventris* Nixon (Hymenoptera: Braconidae), has been explored (Figure 1b) (Costa-Lima 2011; Costa-Lima *et al.* 2014). Producers are already interested in this management option, mainly in crops such as melon (*Cucumis melo* L.), watermelon (*Citrullus lanatus* Thumb. Mansf.), potato (*Solanum tuberosum* L.), tomato (*Lycopersicum esculentum* Mill.), onion (*Allium cepa* L.) and ornamental plants.

This work presents the details of the system of breeding larval-pupal parasitoids of leafminer flies. As a model, the braconid, *O. scabriventris*, was reared on the leafminer fly, *L. sativae*. However, the same system can be applied to other species of mining flies and larval-pupal parasitoids. The possibility of mass breeding of this parasitoid allows its application in biological control and / or pest management programs.



Appendix 3 - Figure 1. a) Adult of *Liriomyza sativae* and b) Female of *Opius scabriventris* parasitizing *L. sativae* larva. Photo: Tiago C. da Costa-Lima

Host plants
Even with the advent of artificial diets for rearing of a great diversity of insects (Parra 2009), most of the natural enemies commercialized in the world are produced on the pest reared in their host plant (Lenteren 2000).

For rearing *L. sativae*, it is recommended to use cowpea plants [*Vigna unguiculata* (L.) Walp]. This species is a natural host of *L. sativae* in Northeastern Brazil and is easier to cultivate, when compared to other hosts such as melon. Another option that can be adopted is the pork bean [*Canavalia ensiformis* (L.) DC]. This species is not a preferred host for *L. sativae*, so there is a need for a greater number of generations to adapt the population to this plant.

The positive factors for the use of *C. ensiformis*, is its large leaf area of cotyledon leaves and the low incidence of pests and diseases in its cultivation. Depending on the region, ease and availability, other host plants can be used, considering the polyphagy of the main pest species of the genus *Liriomyza*.

Planting
For planting cowpea, the use of square pots allows better use of the internal space of the cages. In the evaluations carried out, three plants sown directly in 19 cm side and 19 cm high pots were used, containing substrate with a mixture of soil, sand and cattle manure (1: 1: 1).

The plants must be kept in a greenhouse with irrigation (Figure 2) and sent for breeding when they have at least three expanded trifoliate leaves. After cutting the leaves infested by the leafminer fly, the plants can return to the greenhouse for reuse. On average, it is possible to use each plant twice in the rearing. The use of trays for planting seedlings can also be adopted, which facilitates the transportation of plants.



Appendix 3 - Figure 2. Cowpea plants [*Vigna unguiculata* (L.) Walp] kept in a greenhouse with sprinkler irrigation for later use in the system of rearing the leafmining flies.
Photo: Tiago C. da Costa-Lima

Care in plant maintenance

The availability of healthy plants is the main point for maintaining the leafmining fly breeding system. For this, special care is necessary at this stage of production. In the greenhouse, it is recommended to adapt an antechamber with an air curtain, to hinder the entry of other pests and diseases. When planting cowpea in a greenhouse, powdery mildew and pest arthropods, such as aphids, mites and the leafmining fly, are

common. In the latter case, care must be taken not to place new plants in cages with larvae of *Liriomyza* spp. so as not to contaminate the rearing with an unknown species. The use of yellow adhesive traps in the greenhouse assists in the detection of adult leafmining flies, aphids and whiteflies. The plantings should be staggered, at least once a week, to always have plants at the age appropriate for use.

Adult leafminers

Obtaining the initial population

The easiest way to obtain the initial population of leafmining flies is by collecting larvae from the leaves of host plants. The greater the number of individuals obtained, the greater is the genetic variability.

On the way to the laboratory, the leaves must be stored in paper bags. The collected leaves must be positioned in specific collectors for the isolation of pupae (Figure 3). Since there is no specific structure for collecting pupae, the leaves can only be perforated with wire to be suspended and positioned above a plastic tray. With a fine-tipped brush, moisten and remove the pupae to store in a petri dish. With the emergence of adults, release is performed inside the cages.

Identification

Because of the difficulty in morphological identification of *Liriomyza* spp. in Brazil, the most practical method is the molecular characterization of the population. There are a large number of sequences deposited in GenBank relating to polyphagous species of the genus *Liriomyza* that can be compared. The mitochondrial gene of Cytochrome Oxidase I (COI) is the most used (Kox *et al.* 2005; Scheffer *et al.* 2006).

In Brazil, studies have also been carried out using this technique (Costa-Lima *et al.* 2009; Ferreira 2014; Parish *et al.* 2016).

Cages for adult leafmining flies

The cages for rearing the flies need to have their sides and top face with anti-aphid screen. If you do not have these areas for air exchange, there is condensation inside the cage due to the concentration of plants and, consequently, the death of adults can occur.

Different sizes of cages can be used. If they are accommodated on a metal shelf (1.90 m high, 90 cm wide and 30 cm deep) with two shelves, one should try to make the most of the space. Therefore, the recommended cages should have a 52 cm x 80 cm base and 63 cm high, with the side and top faces with anti-aphid screen and front door with a zipper opening.

Above the cages, two fluorescent lamps (20 W) or equivalent LEDs should be placed, connected to a timer with regulation for 12-hour photophase. To reduce the escape of insects during the exchange of plants, it is advisable to install lamps on the wall directed towards the rear face of the cage. At the time of exchange, the lamps above the creation must be extinguished and the rear lights turned on.

Each cage can support approximately 1,800 adults with up to eight potted plants. However, this proportion of fly / plant per cage can be determined visually as the person responsible for breeding gains experience in this management. A high infestation can result in premature fall of the leaves of the bean and consequent death of the larvae, or lead to pupae of less weight and, consequently, females of less fertility (Petitt & Wietlisbach 1994).

For food, 10% honey should be made available, which increases the longevity of adults. This can be supplied by capillarity, through cotton or filter paper placed in plastic containers. There can be no space that allows adults to enter the honey solution. The solution must be changed at most every 3 days to avoid the proliferation of fungi.

It is also feasible to keep the cage-breeding cages in a greenhouse. However, especially in very hot regions, it must have systems to reduce the ambient temperature. Thus, some generations must be necessary to adapt the breeding of flies in this condition. To change the cages, it is advisable to use a dark fabric on the top and the cage door to reduce the escape of adults.

Immature fly stages
Egg / larva development phase

After the plants have been exposed to the leafmining flies for 24 hours in the cages, they must be taken to the greenhouse, where the egg and larva stages will develop. When the larvae are of average size, in the second and beginning of the third instar (corresponds to 4 to 5 days at 25 °C), the leaves must be cut and taken to the laboratory. At this stage, 30% of the larvae must be directed to maintaining the leafmining fly population and 70% to the parasitoids.

Collection of the leafmining fly pupae
This is one of the main stages in the mass-rearing system. For this, collectors have been developed that reduce working time and do not require pupal manipulation. Two types were tested and demonstrated efficiency, which are:

1) Collector on shelf
It consists of a metal shelf 1.90 m high and 30 cm wide. Each rack has three collectors. The infested leaves are hung on wires, in a total of 11, arranged in the antero-posterior direction of the shelf, fixed under a shelf. Below this, two PVC gutters with 25 cm in diameter and with 25° of inclination are positioned, directed to a plastic container of 12 cm in diameter and 8 cm in height, which collects the pre-pupae from the leaves. This is darkened, as long as the larvae have a negative phototropic behavior (they seek the dark), causing them to reduce their movement and consequent energy expenditure.

In a fourth shelf of the bookshelf, the containers with pupae are placed, closed with a voile lid (Figure 3a). In regions with a drier climate, the shelves should be kept in a small room with a humidifier (RH above 70%) to slow the loss of water by the leaves. Regarding the collection capacity, each wire can fix up to 60 leaves, therefore, 660 per shelf and 1,980 per shelf.

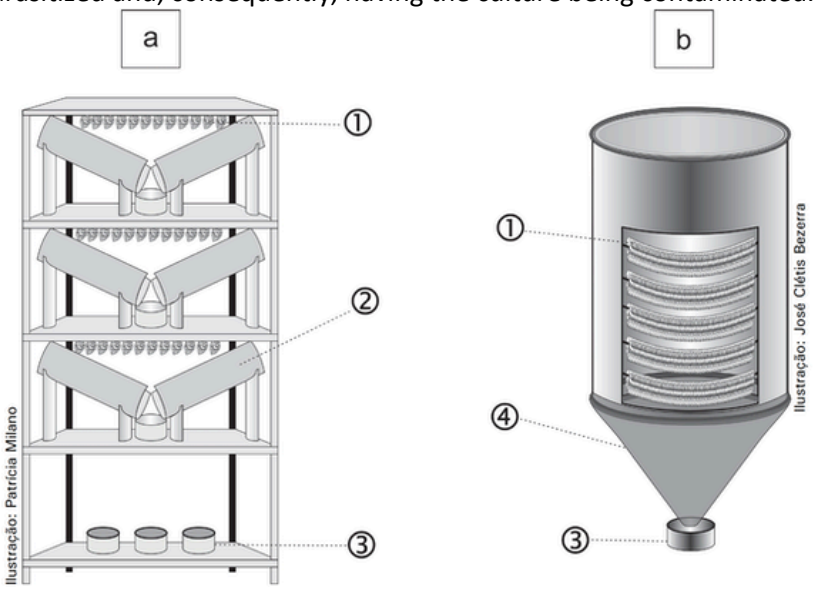
2) Cylindrical collector
It consists of a cylindrical metal base (85 cm high and 55 cm in diameter - 200 L capacity), with a door made of transparent plastic material (55 cm high and 40 cm wide) with a zipper opening (Figure 3b). Inside, there are five 20 cm iron bars on each side (opposite) spaced 20 cm from each, in the upper-lower direction. These serve as a support to accommodate the bean-infested leaves with miner fly larvae. For this, sets of 100 infested leaves are perforated by wires 1 mm in diameter and 40 cm in length. This set is placed on the internal supports mentioned. At each level (total of five), it is possible to accommodate up to three sets of sheets. Thus, the chamber supports a total of 15 sets of 100 sheets each, totalling 1,500 at a time.

The upper circle of the chamber is closed with sheer fabric. Below the cylinder, the structure is similar to that used by Haji *et al.* (2002) for the collection of eggs from *Sitotroga cerealella*. A funnel made of transparent plastic material is attached, with a top opening of 55 cm in diameter and a bottom opening of 10 cm, with 50 cm in length. In the lower opening, a 1,000 ml cylindrical plastic container (12.5 cm high, 10 cm in diameter) is screwed. The larvae that come out of the leaves fall into the funnel and are collected in the darkened container (Figure 3b).

For both collectors, the container must be changed daily and closed with a voile lid. As soon as the first adult flies emerge, they are inserted into the breeding cages. The collection capacity of each system can be estimated considering that the leaves remain fixed for 3 days and establishing an average of 30 larvae /

leaf. Thus, the support capacity every 3 days for the collector on a shelf would be 69,300 and for the cylindrical, 45,000 pupae.

Both are efficient, however, for regions with lower RH, the cylindrical collector may be more suitable, considering that it does not require the use of a humidifier. As well, because it is closed, it reduces the risk of the larvae being parasitized and, consequently, having the culture being contaminated.



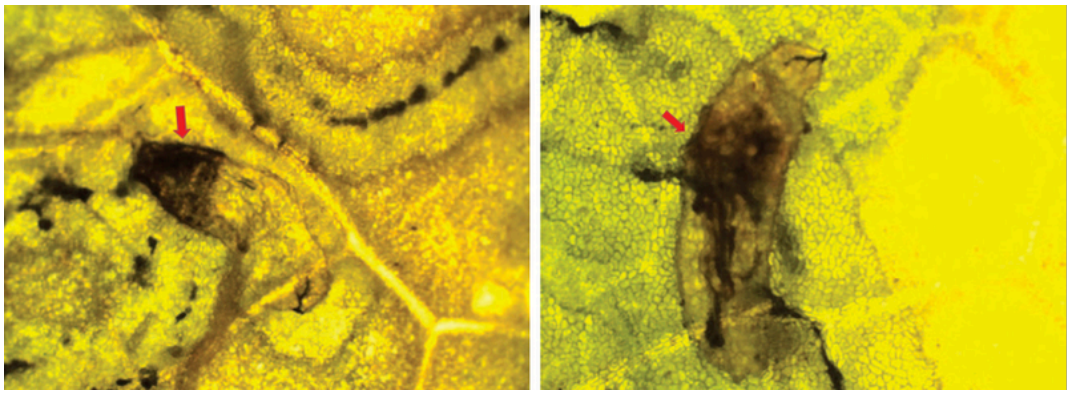
Appendix 3 - Figure 3. Details of the two types of collector pupae of mines and larvae pupa parasitoids. a) Collector on shelf: composed of metal shelf, wire for supporting leaves (1), PVC gutters (2) and pupae storage container (3); b) Cylindrical collector: composed of a 200 L metal cylinder, wire to support the leaves (1), plastic funnel (4) and pupae storage container (3).

Parasitoid breeding

The cages for parasitoids must have a structure similar to those described for the leafmining flies; however, the top face must be made of plastic or acrylic material. In this way, you can use this face to apply thin rows of pure honey using an entomological pin.

Considering the possibility of offering only the highlighted leaves, the size of the cages may be smaller, compared to that used for rearing the flies, especially in height. In the laboratory, *O. scabriventris* was multiplied in 60 cm x 50 cm base cages and 50 cm high. However, depending on the space to accommodate them, these dimensions may change.

Medium-sized larvae (second and beginning of third instars) are ideal for offering to the parasitoid *O. scabriventris*. The females of this braconid, as well as the majority of parasitoids of leafmining flies, also kill the larva to feed (Figure 4). The high number of predated larvae is undesirable, as it increases the cost of production, since the larva that could have been parasitized (resulting in a parasitoid) has been killed. At 25°C, *O. scabriventris* was found to prey on one larva for every three parasites (Costa-Lima 2011).



Appendix 3 - Figure 4. *Liriomyza sativae* larvae killed by predation of *Opius scabriventris* females. Arrows indicate area of necrotic tissue as a result of the introduction of the parasitoid ovipositor

To use the cut infested leaves in the parasitoid cages it is recommended to use a floral sponge to keep the leaves turgid. This type of sponge is normally sold for flower arrangements. It must be surrounded by plastic film, leaving only the upper face free (Figure 5). Use a stick with water to moisten the sponge, approximately 800 mL for a 22 cm x 10 cm x 6.5 cm block. The leaf petiole should be inserted into the sponge carefully so that the leaves do not overlap.



Appendix 3 - Figure 5. Cowpea leaves [*Vigna unguiculata* (L.) Walp] with larvae of *Liriomyza sativae* inserted in a moist floral sponge, wrapped with plastic film for exposure to parasitism by *Opius scabriventris*.
Photo: Tiago C. da Costa-Lima

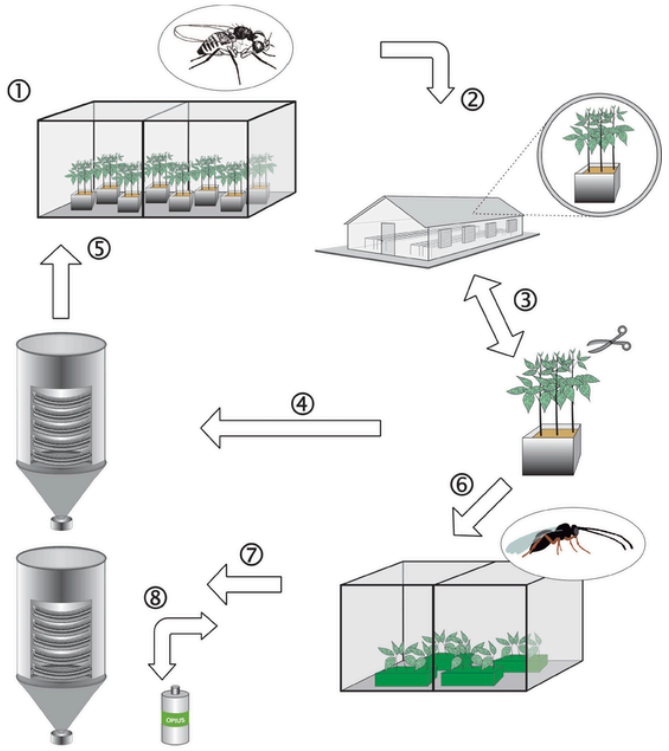
There is also an option not to use the detached leaves but to put the pots or trays with infested plants to the parasitoid cages. For this, they need to have dimensions similar to those described for the rearing of the leafmining flies.

The leaves with larvae of *L. sativae* are exposed for 24 hours to parasitoids. After that period, these must be exchanged. The removed leaves must be sent to a pupal collector similar to the one described above for the flies. Non-parasitized larvae generate flies, most of which emerge before parasitoids. However, there will be a part that will coincide with the emergence of the parasitoid. For the separation, it is recommended to release these in a cage and with the aid of an electric sucker, collect only the parasitoids.

In case of commercial purpose, part of these parasitoids will be directed to the packaging stage and the other part will return to the rearing facility. Depending on the demand for commercialization, the proportion of these destinations can be determined. All stages of the breeding system are summarized in Figure 6.

Production planning

Temperature variation can help with planning to delay or accelerate the production of flies / parasitoids. For this, the pupal phase is the easiest to manipulate the flies. With the use of the collectors it is possible to isolate only the pupae and store them in air-conditioned chambers with the desired temperatures. For *L. sativae*, between 30°C and 15°C the pupal stage lasts 7.5 and 23.9 days, respectively, without affecting viability (Table 1) (Costa-Lima *et al.* 2009).



Appendix 3 - Figure 6. Scheme of production of the parasitoid, *Opius scabriventris*, on the agromyzid, *Liriomyza sativae*. 1) Cage for plant infestation; 2) development of the egg and larva stages; 3) cutting the leaves with 2–3º instar larvae; 3) plants with cut leaves return to the greenhouse to be reused; 4) 30% of the leaves containing larvae sent to collector of pupae of miner-fly; 5) when they emerge, adult flies are added to the cages; 6) 70% the leaves directed to the parasitoid cages; 7) leaves exposed to parasitism sent to pupae collector; 8) when they emerge, the parasitoids are separated for the maintenance of the breeding and for packaging and commercialization.

Appendix 3 - Table 1. Average duration, in days, of the egg phases, larva, pupa and egg-adult period and viability (%) of the larvae and pupa phases of *Liriomyza sativae*, in cowpea [*Vigna unguiculata* (L.) Walp], in different temperatures (RH 50 ± 10% and photophase 14 hours).

°C	Egg (days)	Larva (days)	Pupa (days)	Egg-Pupa (days)	Larval survival (%)	Pupal survival (%)
15	6.7	10.1	23.9	40.9	80.1	79.7
18	5.6	7.1	15.1	28.0	85.4	80.0
20	4.7	6.2	14.6	25.7	88.5	84.1
22	4.2	5.7	11.3	21.4	85.2	83.3
25	2.8	4.9	8.9	16.5	84.8	78.8
28	2.5	4.2	8.0	14.3	87.7	79.7
30	2.0	4.1	7.5	13.7	86.5	82.1
32	1.6	3.8	7.3	12.9	79.5	40.1

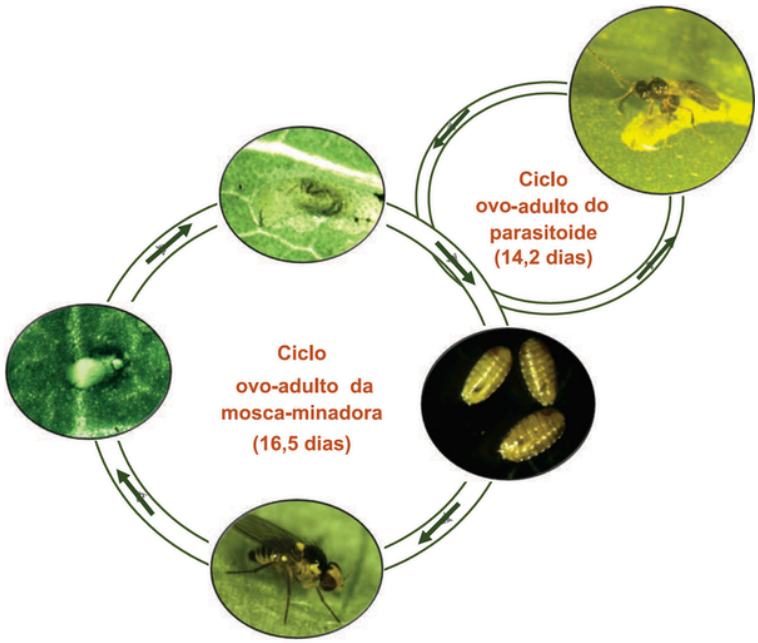
Source: Adapted from Costa-Lima *et al.* (2009).
For the parasitoid *O. scabriventris*, also in the range of 30°C and 15°C, there is no interference in pupal viability and the egg-adult period ranges from 13.3 to 29.8 days, respectively (Table 2) (Costa-Lima *et al.* 2014).

Appendix 3 - Table 2. Average duration, in days, of the egg-adult period and pupal viability (%) of *Opius scabriventris*, at different temperatures, on *Liriomyza sativae* in cowpea [*Vigna unguiculata* (L.) Walp] (RH of 50 ± 10% and photophase of 14 hours).

°C	Egg-adult period	Pupal viability (%)
15	29.84	86.1
18	24.6	89.4
20	22.54	92.3
25	14.27	92.9
30	11.21	92.3
32	13.33	6.6

Source: Adapted from Costa-Lima *et al.* (2014).

The adults of *O. scabriventris* have an average longevity of 36.5 days at 25 °C, with their peak of oviposition on the sixth day (Costa-Lima 2011). The use of lower temperatures may prolong its longevity, however, further studies are needed to verify its effect on parasitism. Therefore, production planning, packaging and shipping to the producer must consider these periods. For *O. scabriventris*, there is no interference in pupal viability between 30°C and 15°C, and the egg-adult period ranges from 13.3 to 29.8 days, respectively (Table 2) (Costa-Lima *et al.* 2014). For both species, from constant temperatures of 32 °C, pupal viability is reduced (Tables 1 and 2). The life cycle of *L. sativae* and its parasitoid, *O. scabriventris*, is shown in Figure 7.



Appendix 3 - Figure 7. Scheme of the egg-adult cycle of the leafmining fly, *Liriomyza sativae*, and its parasitoid, *Opius scabriventris*, with average duration at 25°C.

Commercialization

In countries that already offer parasitoids of leafmining flies, these are sold as adults in different quantities, with 250, 500 and 1,000 adults per package. Honey must be offered inside this package, however, it must be arranged in thin lines so that adults do not stick and die in the food. Adding a

substrate, such as folding paper strips, can help distribute the parasitoids in the packaging. The cover must contain voile fabric or other material that allows air to be exchanged and insects not to escape.

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Appendix 4 Quality control tests for *Dacnusa sibirica* and *Diglyphus isaea* (van Lenteren *et al.* 2003)

(a) Dacnusa sibirica Telenga (Hymenoptera: Braconidae)

Test conditions

Temperature:	22 ± 2°C
RH:	60 ± 5%
Light regime:	16L: 8D

Quality control criteria

Quantity	≥ number of live adults specified on the label; a weekly or batch-wise test.
Adult mortality	≤ 5% of the number of adults present in the container, based on three containers sampled and *n* = 500 or more; a weekly or batch-wise test.
Sex ratio	≥ 45% females; *n* = 100; conducted four times per year.
Fecundity	≥ 45 offspring per female within 3 days; *n* = 15; an annual test. Daily oviposition of single female wasps on brown beans (*Phaseolus vulgaris*) infested with sufficient *Liriomyza trifolii* and a source of carbohydrate (other hosts if *L. trifolii* is not available: *Liriomyza bryoniae* or *Chromatomyia syngenesiae*).

Description of testing methods

Fecundity Test: *Dacnusa sibirica* using *Chromatomyia* as host

Plant	*Sonchus oleraceus* grown from seed for *c*. 8 weeks.
Host leafminer	*C. syngenesiae*, a minimum of 100 second-instar larvae per plant (*c*. 8 days after eggs laid).
Apparatus	60 cm Perspex cube cages.
Day 0	Place a single female *Dacnusa* into a cage containing three plants infested with leafminer on 3 consecutive days and leave for 72 h.
Day 3	Remove the females from the cages. Leave the plants *in situ* until the leafminer larvae have pupated (*c*. 5 days later).
Day 8±	Cut plants off, save leaf and stem tissue containing pupae and discard the rest. Store pupae in a ventilated plastic box (*c*. 30 cm × 30 cm × 15 cm is ideal), lined with several layers of absorbent paper, at RH of 80% and 22°C for 21 days.

Day 29	Freeze the box for 24 h and count *Dacnusa* adults.

Fecundity Test: *Dacnusa sibirica* using *Liriomyza* as host

Plant	*Phaseolus vulgaris* grown from seed 2 weeks before starting the test.
Host leafminer	*Liriomyza bryoniae.*
Preparations	Two weeks before starting the test, sow *c*. 300 *P. vulgaris* seeds in small pots with soil. Ensure that the plants can grow under optimum conditions (sufficient light and nitrogen, no sciarid flies), as strong plants are required. After 2 weeks remove the tops of the plants, leaving only the spade leaves. About 500 1-day-old female *L. bryoniae* are then released on to the plants (the leafminers are provided with honey from the time they emerge from the pupae). Keep the plants in a greenhouse at about 22–25°C until the first larvae have reached second-instar (*c*. 7 days).
Day 0	Count the approximate number of larvae on the infested plants. Place as many plants in a cage as is necessary to provide about 150 larvae per parasite. Do not use plants with more than 30 larvae per leaf. Collect 15 female wasps from a bottle. Put one female in each cage.
Day 3	Put/fix a sleeve or ventilated box around the leaves to collect the larvae, which will pupate within about 5 days at 22°C. The RH must be 80%.
Days 5–8	Check that all the larvae have pupated. Before they hatch, collect all the pupae in a Petri dish and store them at 22°C and 80% RH.
Day 20 ±	Check that all the pupae have hatched and count the number of adults emerged after killing them in a freezer. Calculate the number of offspring produced per female during the 3-day test.

Design: J. Dale, P. Smytheman and R. Greatrex.

Coordinators: P. Smytheman, J. Dale and R. Greatrex.

(b) ***Diglyphus isaea*** **(Walker) (Hymenoptera: Eulophidae)**

Test conditions

Temperature:	25 ± 2°C
RH:	70 ± 10%
Light regime:	16L : 8D
Light intensity:	< 300 lx; direct lighting to be prevented – it affects the activity of females.

Quality control criteria

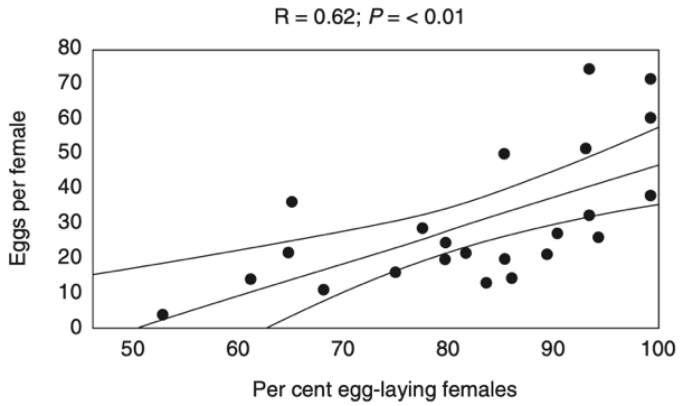
Quantity	≥ number of live adults specified on the label; a weekly or batch-wise test.
Adult mortality	≤ 8% of the number of adults present in the container, based on three containers sampled and *n* = 500 or more; a weekly or batch-wise test.
Sex ratio	Females ≥ 45% of live adults; *n* = 100; a batch-wise test.
Fecundity	70% of females laying eggs within 1 week in a Petri-dish test; *n* = 30; an annual test.

Description of testing methods

Fecundity	Isolate individual females, chosen at random from the container, in Petri dishes of *c*. 12 cm diameter. Use Parafilm to seal the dish. For each replicate, staple a brown bean leaf (*P. vulgaris* L.) infested by *c*. 10 L_2–L_3 larvae of *L. trifolii* or *L. bryoniae* to a slightly moistened filter-paper and place it in a dish. Add some honey droplets to the lid of the dish. Alternatively, *S. oleraceus* L. leaves infested by *C. syngenesiae* Hardy can be used. Replace the leaf daily for up to 7 days and check for presence of parasitoid eggs in the mines. Leaves can be stored prior to dissection for up to 24 h at 4–8°C. For each female the test is completed once the first parasitoid egg is found. At the end of 7 days, calculate the percentage of females that have laid eggs. Include the total females which died naturally. The number of females accidentally killed or lost should be omitted from the calculations, but must be recorded.

Design: G. Nicoli and S. Steinberg.

Coordinators: S. Steinberg, P. Smytheman and G. Burgio



Appendix 4 - Figure 1. Prediction of fecundity. The number of eggs laid per female in 7 days can be predicted from the relationship between this value and the percentage of egg-laying females (each replicate: n = 30 females). The fecundity data were obtained using the testing method described above (G. Nicoli, unpublished data).

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Appendix 6: Agromyzid leafminers, their hosts, and their parasitoid wasps within the Torres Strait Islands and Cape York Peninsula, with a focus on vegetable leafminer (*Liriomyza sativae*)

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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Contents

Introduction 3
Methods 4
Field trips 4
Host range confirmations for *L. sativae* 5
Specimen collections 5
Species identification 5
Assessing parasitism rates 5
Results 6
L. sativae geographic range 6
L. sativae host range in TSI and NPA 6
Other agromyzids 9
Parasitoid wasps 10
Parasitism rates 15
Discussion 16
Acknowledgements 17
References 17
Appendix 1. Longer term average parasitism rates 18
Appendix 2. Surveillance records for May 2018 20
Appendix 3. Database of agromyzids and parasitoids 27

Introduction

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. Nearly all agromyzid species are very host-specific but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world (Spencer 1973, 1990). This includes the tomato leafminer (*Liriomyza bryoniae),* the serpentine leafminer (*L. huidobrensis),* the vegetable leafminer (*L. sativae),* the American serpentine leafminer (*L. trifolii)*, and the pea leafminer (*Phytomyza horticola*), all of which are pests of numerous vegetable and ornamental crops and thus pose a significant quarantine threat to Australia. *Liriomyza sativae* has been recorded in Australia in far north Queensland (International Plant Protection Convention 2017). Following the detection of *L. sativae* in the Torres Strait Islands (TSI) in 2008 and Papua New Guinea in 2011 (Blacket et al. 2015), it was recorded at Seisia, Queensland, in the Northern Peninsula Area (NPA) in 2015 (International Plant Protection Convention 2017). Eradication of *L. sativae* from Torres Strait or Far North Queensland was not considered technically feasible, and phytosanitary measures are in place to prevent the further spread of the pest on mainland Australia (International Plant Protection Convention 2017). At the commencement of project MT16004, there was no evidence that *L. sativae* had spread outside of the town of Seisia in the NPA.

The most effective natural control of Agromyzidae comes from parasitoid wasps. Mortality results from parasitism, or by stinging and feeding by adult wasps. These pests have caused significant economic damage internationally and are a classic example of secondary pests. Outbreaks often occur when the use of a pesticide to reduce densities of a target pest species triggers subsequent outbreaks of leafminer species (Ridland et al. 2020). Major infestations/outbreaks of these polyphagous *Liriomyza* are mostly likely to occur when (1) natural enemies (primarily parasitoid wasps) are reduced in cropping systems by insecticides targeting other pests in the crop or targeting the leafminers themselves, or (2) naturally occurring parasitoids are not present in the first place (Ridland et al. 2020).

Australia has at least 50 species of parasitoid wasps that attack native leafminer flies. Of these, some species including *Hemiptarsenus varicornis*, *Diglyphus isaea* and *Opius* spp., are known to be particularly important for vegetable leafminer management overseas. In addition, some Indo-Australian species such as *Zagrammosoma latilineatum* may play important roles should exotic leafminers establish in Australia.

Our aim was to (1) conduct surveillance in TSI and along the Cape York Peninsula, Queensland (CYP) to look for any evidence of range expansion of *L. sativae*, with particular focus on the tourist routes between Cairns, Weipa and the Northern Peninsula Area (NPA) communities, (2) record species of parasitoid wasps currently targeting *L. sativae* within TSI and the NPA, and 3) conduct a survey of additional native or naturalized agromyzid leafminer species and their parasitoid communities along CYP and TSI.

Methods

Field trips

Four surveillance field trips targeting agromyzid leafminers, including *L. sativae*, were conducted between 2017 and 2019. Across all trips we sought to survey for *L. sativae*, looking for any evidence of geographic range or host range expansions, assess parasitism of *L. sativae*, and look for other reservoirs of parasitoids on native or naturalised agromyzid leafminers.

July 2017

We completed the first field survey in July 2017 on Thursday Island (TI) and the NPA, led by Northern Australia Quarantine Strategy (NAQS) scientists from Cairns, Queensland. Aims included (1) establishing connections with local NAQS officers, establishing good working relationships with the many political and cultural stakeholders in the region, and becoming familiar with the pest and the landscape, and (2) surveying for *L. sativae* and recording occurrence locations and host plant records. We met with the staff of the NAQS offices on TI and in Seisia (on the NPA) as well as several key stakeholder groups, including representatives from the Torres Strait Regional Authority, the Torres Shire Council, My Pathway (a national company providing education, training and employment services, which is involved in community garden activities on TI and the NPA), the Torres Strait Island Regional Council, the Kaurareg Native Title (Aboriginal) Corporation (representing the traditional owners of the Thursday Island group), Seisia enterprises Pty Ltd, the Northern Peninsula Area Regional Council, and the Apudthama Land Trust (representing traditional owners within the NPA). Surveillance for *L. sativae* was conducted along public roadsides, parks and private residences where we had permission. Any new local host preferences were recorded. Across all trips, we photographed any leaf mining observed, and recorded host plants.

May 2018

In May 2018, a 2.5 week survey trip along the CYP was conducted that included stops in Lakeland, Cooktown, Weipa, and the Thursday Island group of the Torres Strait (including TI, Horn Island, Roko Island, Zuna Island, and Albany Island). Aims included (1) surveillance for *L. sativae* and its host plants along the CYP and TSI, including the collection of leaf mine samples for possible molecular diagnosis, (2) a survey of parasitoids attacking *L. sativae* in the TSI and NPA, (3) a survey for possible reservoirs of parasitoid wasps in other native agromyzid species found along the CYP. We surveyed farms around Lakeland, major roadhouses and campgrounds along the CYP, major shipping ports in Cairns, Weipa, Seisia, TI and Horn Island, community gardens, and private residences where permission were obtained. At each site, we recorded any host plants observed and any signs of agromyzid mining (See **Figure 1**).

July/Aug 2018

In July & August 2018, we spent 3 weeks based on TI. This trip focussed on surveying the shipping ports of Seisia, TI and Horn Island, public roadsides and parks, community gardens, and private residences where permission were obtained. We also undertook a series of field experiments aimed at supporting surveillance tactics. Aims included (1) surveillance for *L. sativae* and its host plants in the TSI and NPA and (2) collection of leaf mine samples for molecular diagnosis.

May 2019

In May 2019, we spent 1 week based on TI to conduct follow up experiments and conduct surveillance on TI and the NPA. Additional aims included the quantification of parasitism rates within *L. sativae*.

Host range confirmations for L. sativae

During the May 2018 survey trip, all sites and host plants where *L. sativae* was observed were recorded. Across all other trips, *L. sativae* activity was only recorded when it was observed in new regions or potentially new host plants. Larvae were preserved into 100% ethanol to allow confirmation of each host plant via DNA identification. For mining damage that did not contain live larvae, empty leaf mine samples were preserved into 100% ethanol for analysis with a novel eDNA test (see Pirtle et al. 2020).

Specimen collections

Across all trips, but primarily during the May 2018 survey trip, any larvae of unknown species were maintained in rearing vessels to allow for adult emergence (either fly or parasitoid wasp) and morphological identification of specimens. Rearing vessels included rearing bags (a large sealed plastic bag containing numerous leaves, and with a higher risk of containing additional invertebrate species - thus parasitoids could be associated with species other than agromyzids) and petri dishes (containing less than five leaves each confirmed to contain only the target larval flies or wasps).

Species identification

Identifications by expert taxonomists were sought for a subset of specimens collected across all trips. Dr. Mallik Malipatil completed identifications for fly specimens, and Dr. Ryan Perry (University of California Riverside) and Dr. Roger A Burks (University of California Riverside) completed identifications for wasp specimens. All other identifications were conducted by the collectors (Dr. Elia Pirtle, Luke Halling and Sally Cowan), with assistance from Dr. Peter Ridland and Samantha Ward. All host plant identifications were made with assistance from Barbara Waterhouse.

Assessing parasitism rates

In May 2019, parasitism rates were assessed by randomly selecting plant leaves, inspecting each under a microscope, and recording the presence of healthy fly larvae, dead fly larvae, wasp larvae, and wasp pupae. Additionally, the emergence hole created by a wasp can typically be distinguished from that of a fly due to the presence of meconial pillars (Hendrickson and Barth 1978; Appendix 1).

Parasitism rates were calculated two ways, acknowledging that some mortality is driven by parasitism but may also be driven by host feeding (Ridland et al. 2020). To calculate the minimum parasitism rate, the number of mines containing signs of parasitism (taken as those with a wasp larvae visible on the back of a fly larvae (WL), or those containing a wasp pupa (WP)) was divided by the number of total mines (including all other mines including dead fly larvae on which no wasp larvae were visible (DL) and healthy fly larvae (HL)). However, dead fly larvae on which no wasp larvae were visible could still be a result of parasitoid activity, as a result of host feeding. Thus, to calculate the maximum mortality rate as a result of parasitoid wasp activity, the number of mines containing signs of parasitism (taken as those with a wasp larva visible on the back of a fly larva, or those containing a wasp pupa), or containing dead fly larvae with no signs of wasp larvae, was divided by the number of total mines (including all other mines including healthy fly larvae). To estimate a longer-term site average, we also characterized the exit holes of older leaf mines into

those with signs of a fly emergence versus those with signs of a wasp emergence. While a coarse measurement, this provided an indication of the longer-term impacts given mines persist as long as the leaf persists, meaning the random collection of mines at a given site could represent at least one month of fly and wasp activity.

Results

L. sativae geographic range

L. sativae were not detected south of the previously known distribution in Seisia on any of the four trips (See Appendix 2). However, the project team was able to confirm the presence of *L. sativae* on Zuna Island (where presence was previously only suspected) via the eDNA diagnostic test for empty leaf mines (see Pirtle et al. 2020).

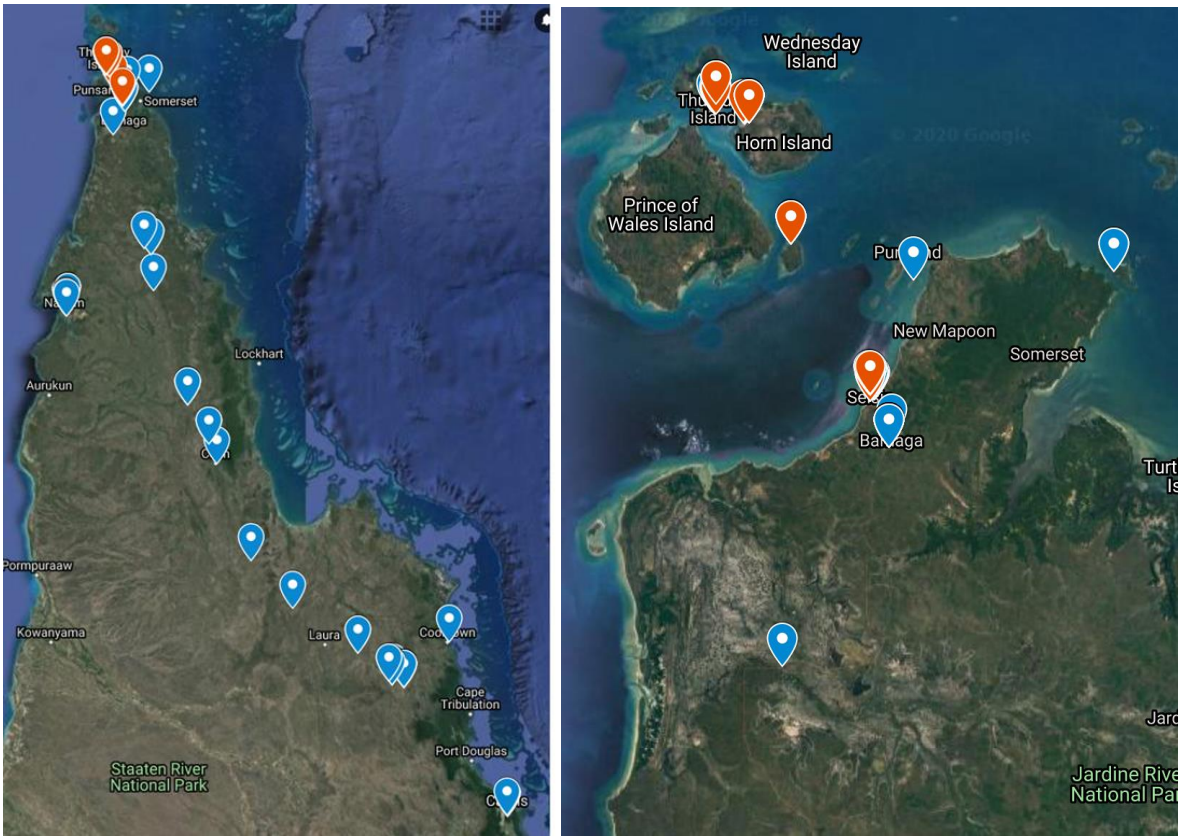


Figure 1. All survey locations, with presence based on the collection of larvae or adult flies or confirmations of old mining based on the eDNA test (red points) and absence (blue points) of *L. sativae* indicated.

L. sativae host range in TSI and NPA

Several new host species were recorded for *L. sativae* within the TSI and NPA (**Table 1**). In several cases, only empty leaf mines were detected, and these hosts could only be confirmed via the novel eDNA test developed within the project (Pirtle et al. 2020).

Table 1. Host plant records for *L. sativae* that had not been previously recorded within the TSI and NPA.

Host	DNA Confirmation	Notes	Photo of mining
Snakeweed (*Stachytarpheta jamaicensis*)	Y	Very light mining was found at the Tagai College Harmony Garden on Horn Island in May 2018; a sample was taken of a mine found to contain a larva and DNA confirmed this larva was *L. sativae*. Surveillance on Thursday Island in July 2018 did not yield any mines on snakeweed, however, surveillance on Thursday Island in May 2019 did yield a patch of snakeweed with at least 10 leaf mines. DNA diagnostics confirmed these mines were created by *L. sativae*. This species has been flagged with NAQS for future surveys.	
Wild passionfruit *(Passiflora foetida)*	Y	Mining was observed on wild passionfruit on Thursday Island in May 2019, and DNA diagnostics confirmed the mines were created by *L. sativae*. This is the first observation of *L. sativae* mining on wild passionfruit in the Torres Strait.	
Yellow alder (*Turnera ulmifolia)*	Y	Heavy mining was observed on a single yellow alder plant on Thursday Island in May 2019, and DNA diagnostics confirmed the mines were created by *L. sativae*. This is the first observation of *L. sativae* mining on yellow alder in the Torres Strait.	

Passionfruit *(Passiflora edulis)*	Y	Very light mining was observed on passionfruit on Thursday Island in July 2018, and DNA diagnostics confirmed the mines were created by *L. sativae*. This is the first observation of *L. sativae* mining on passionfruit in the Torres Strait.	
Basil *(Ocimum basilicum)*	Y	Several mines were observed on basil and Thai basil on Thursday Island in May 2018, and DNA diagnostics confirmed the mines were created by *L. sativae*. This is the first observation of *L. sativae* mining on basil in the Torres Strait.	
Chilli (*Capsicum* sp.)	Y	A single very old mine was observed on a chilli plant on Thursday Island in July 2018, and DNA diagnostics confirmed the mine was created by *L. sativae*. This is the first observation of *L. sativae* mining on chilli in the Torres Strait.	
Alternanthera ficoidea	N	A single empty mine was found on *Alternanthera ficoidea*. on Thursday Island in May 2018; this species has been flagged with NAQS for future surveys. The leaf mine was not retained for DNA diagnostics.	

Commelina benghalensis	N	A single empty mine was found in Seisia in May 2018 on *Commelina benghalensis*. This species has been flagged with NAQS for future surveys. The leaf mine was not retained for DNA diagnostics.	

Other agromyzids

At least four non-*L. sativae* agromyzid species were observed in the TSI and CYP (**Table 2**). Leaf mining activity caused by additional agromyzid species were regularly observed in *Alysicarpus* sp. and *Desmodium* sp. weeds and on *Scaevola taccada* plants across all four trips. Identifications were made via adults that were reared on the May 2018 survey trip. Two additional agromyzid species were detected in May 2018, including a species mining in eggplant, and another mining in *Milletia pinnata*. Full details of these specimens can be found in Appendix 3.

Table 2. Notes on non-*L. sativae* agromyzids reared during the May 2018 survey in the TSI and CYP and identified by Dr. Mallik Malipatil.

Host	Notes	Photo of mining
Ophiomyia solanicola	Active leaf mines were found on eggplant crops at a farm in Lakeland; pupation was occurring inside the mines (allowing immediate exclusion of *L. sativae* as a possibility); adults were successfully reared and were morphologically identified by Dr. Mallik Malipatil.	
Ophiomyia alysicarpi	Active leafmines were found on *Alysicarpus* and *Desmodium* weeds at survey stops along the entire Cape and into the Torres Strait; mint green pupae were observed developing inside the mines (allowing immediate exclusion of *L. sativae* as a possibility); adults were successfully reared and were morphologically identified by Dr. Mallik Malipatil.	

Ophiomyia cornuta	Active leaf mines were found on *Alysicarpus* and *Desmodium* weeds; adults were successfully reared from Roko Island and were morphologically identified by Dr. Mallik Malipatil.	
Unidentified agromyzid leafminer	Active leaf mines were found in a single seedling of *Milletia pinata* at Loyalty Beach, however adults could not be reared due to travel logistics.	

Parasitoid wasps

We confirmed at least 8 wasp species attacking *L. sativae* populations in the TSI and Seisia by recording wasp emergences from *L. sativae* larvae that had been isolated in rearing chambers. This included specimens provided by Luke Halling (NAQS) from 2017 collections in siratro (*Macroptilium atropurpureum*) from Horn Island. Specimens were identified morphologically by Dr. John La Salle, Dr. Ryan Perry (University of California Riverside) and Dr. Roger A. Burks (University of California, Riverside). Four of these 8 species were also reared from other agromyzid species collected along the Cape York Peninsula, along with at least an additional 5 additional parasitoid species. These additional species include *Opius* sp, which has been recorded in bulk invertebrate samples in the TSI in the past (Luke Halling pers. comm) and is therefore likely to be attacking *L. sativae* (though it has yet to be confirmed via direct rearing). These species are likely to play an important role in *L. sativae* control should it spread further south.

Liriomyza sativae

At least 8 species were reared from *L. sativae* (**Figure 2**). The most common species were *Zagrammosoma latilineatum, Atoposoma sp., Vagus* nr. *ambiguus* and *Hemiptarsenus varicornis.* Other species included *Closterocerus miribalis*, *Closterocerus* nr. *trifasciatus*, *Apleurotropis* (sp.) nr. *viridis*, *Neochrysocharis* sp., and *Melittobia australicus/hawaiiensis*.

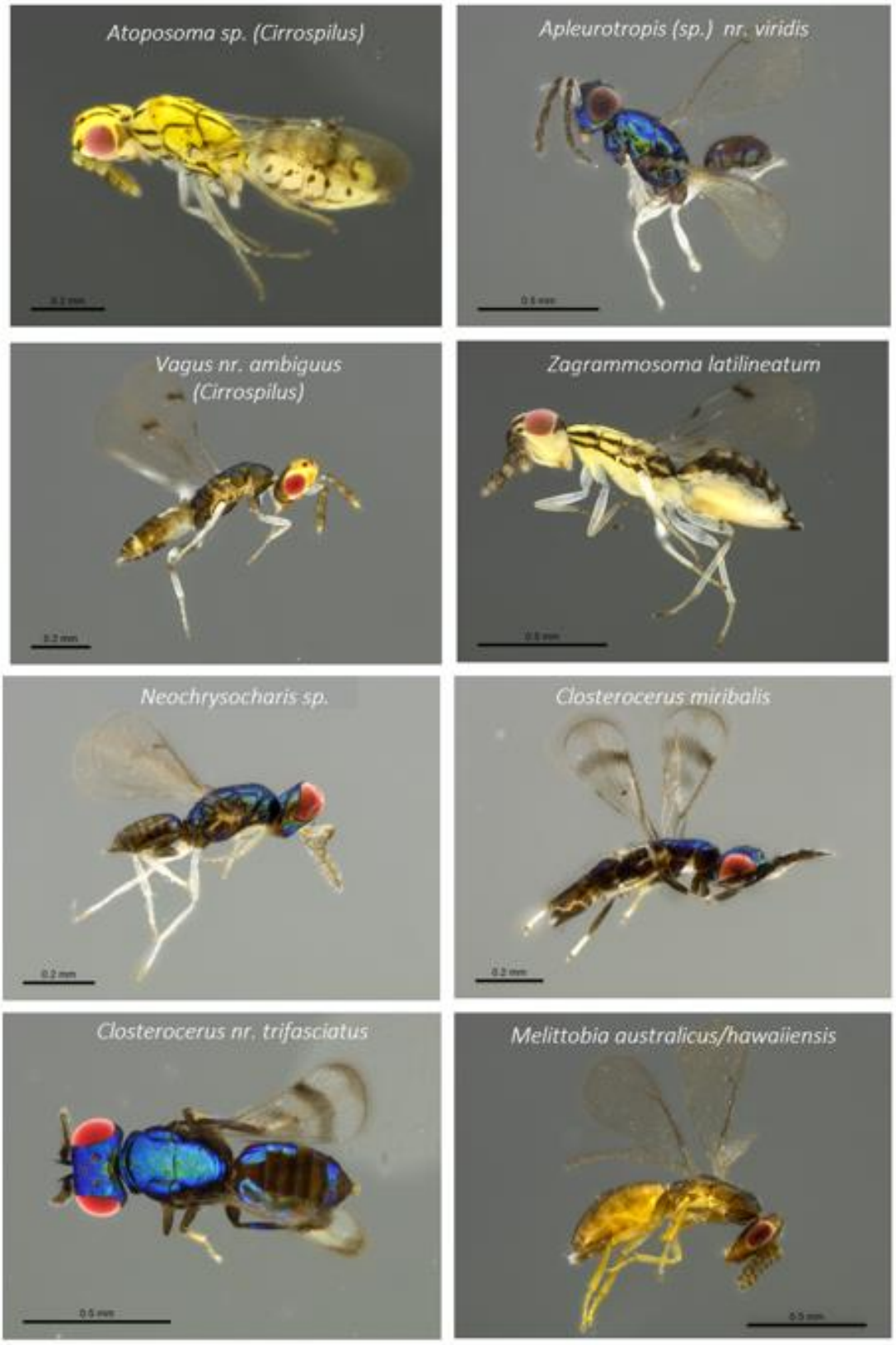


Figure 2. Parasitoid wasp species reared directly from *L. sativae* larvae in the TSI and NPA, and identified by Dr. Ryan Perry and Dr. Roger A. Burks.

Ophiomyia alysicarpi

The following species were reared from *Ophiomyia alysicarpi* on *Alysicarpus* sp. and/or *Desmodium* sp. weeds. Asterisks indicate identifications made by Dr. Ryan Perry and Dr. Roger A Burks (**Figure 3**), double asterisks represent records provided by Luke Halling from *Desmodium tortuosum* in TSI, all other identifications by Dr. Elia Pirtle, Dr. Peter Ridland, and Samantha Ward (**Figure 4**).

- *Zagrammosoma latilineatum*
- *Hempitarsenus varicornis*
- *Opius* sp.
- Encyrtidae*
- *Apleurotopis* sp.*
- *Vagus ambiguus**
- *Neochryocharis* sp.*
- Additional unidentified species x 3 (**Figure 4**)
- *Callitula* sp.**

Ophiomyia solanicola

The following species were reared from *Ophiomyia solanicola* on eggplant (*Solanum melongena*). Asterisks indicate identifications made by Dr. Ryan Perry and Dr. Roger A Burks (**Figure 3**), all other identifications by Dr. Elia Pirtle, Dr. Peter Ridland, and Samantha Ward (**Figure 4**).

- *Opius* sp.
- *Merismomorpha* sp.*
- *Callitula* sp.*
- *Neochryocharis* sp.*

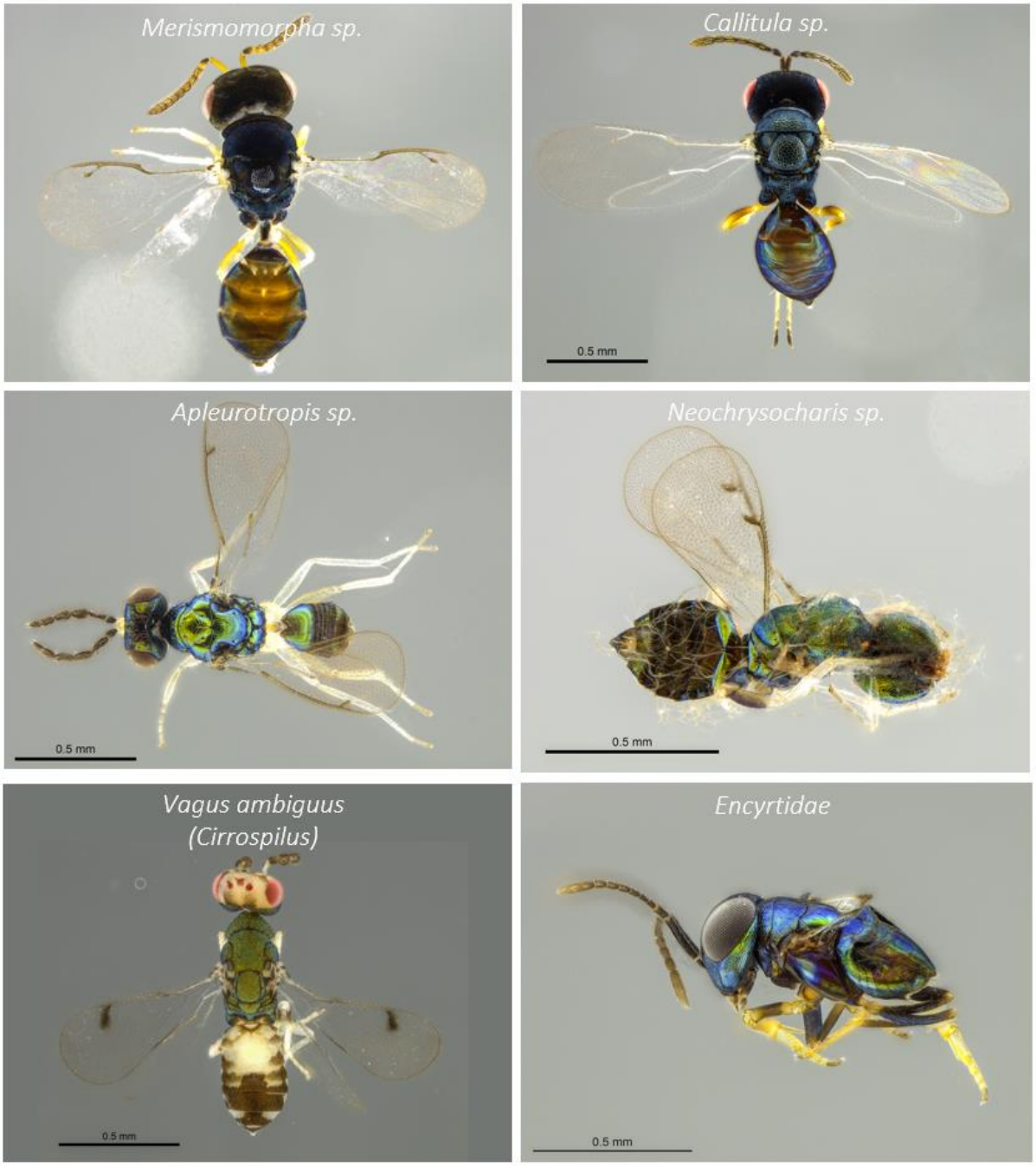


Figure 3. Parasitoid wasp species reared directly from *Ophiomyia alysicarpi* and *Ophiomyia solanicola* larvae in the TSI and CYP, and identified by Dr. Ryan Perry and Dr. Roger A Burks.



Figure 4. Additional parasitoid wasp species reared directly from *Ophiomyia alysicarpi* and *Ophiomyia solanicola* larvae in the TSI and CYP, tentatively identified by Dr. Elia Pirtle, Dr. Peter Ridland and Samantha Ward. Asterisk represents specimens that could have come from either siratro or *Alysicarpus* sp.

Parasitism rates

Parasitism rates on Thursday Island in May 2019 ranged from 50% to 80% in siratro and was 100% in a single patch of *L. sativae* mining discovered in snakeweed (Table 3).

Table 3. Parasitism rates recorded on Thursday Island in May 2019, calculated as number of mines containing signs of parasitism divided by total number of mines (dead or alive).

Location	Site	Host	Total mines*	DL	HL	WL	WP	minimum % parasitism**	maximum % mortality***
Thursday Island	Frog Gully garden	*Macroptilium atropurpureum*	76	26	24	9	17	52%	68%
Thursday Island	Hospital playground	*Macroptilium atropurpureum*	38	18	9	5	6	55%	76%
Thursday Island	Hargrave Hill	*Stachytarpheta jamaicensis*	15	8		3	4	100%	100%

Discussion

We found that *L. sativae* populations in Australia are already being attacked by at least 8 different parasitoids, one of which (*Hemiptarsenus varicornis*) is regarded as one of the most important sources of biocontrol overseas. We identified that two poorly studied Australian species, *Zagrammosoma latilineatum* and *Cirrospilus* sp., were most commonly found attacking *L. sativae* on TI, Horn Island and Seisia.

Although only preliminary, we found these wasps were able to parasitise up to 80% of *L. sativae* in different parts of the TSI and NPA. These high parasitism rates is likely a result of conditions well suited to healthy parasitoid populations and small *L. sativae* communities (relative to agricultural systems), including (1) patchy distributions of *L. sativae* host plants and the absence of large monocrops of hosts, (2) abundant reservoir agromyzids in commonly occurring weeds, and (3) very low insecticide usage in home, community and school gardens.

We found a high degree of overlap between parasitoids of *L. sativae* and *O. alysicarpi*. This overlap suggests *O. alysicarpi* is an important reservoir of *L. sativae* parasitoids. All three of the most common *L. sativae* parasitoids in TSI and the NPA have been found attacking *O. alysicarpi* in the NPA and Weipa. It is therefore likely that these parasitoids were already present in Seisia, being sustained by *O. alysicarpi*, well before the arrival of *L. sativae*, which would have arrived to a parasitoid community already primed to begin controlling them. The presence of this parasitoid community in Seisia may in part explain the surprisingly slow spread *L. sativae* since its arrival on the mainland.

We observed this 'wasp buffer' extending all the way down CYP to Cairns. Additionally, literature reviews conducted within the project (Ridland et al. 2020) show Australia has many native analogous flies that support at least 95 different wasp species. Some of these important species, including *Zagrammosoma* sp. and *Hemiptarsenus varicornis*, extend across large parts of Australia (Ridland et al. 2020).

It is evident that Australia is already equipped with diverse and abundant natural enemies, and there will be little need to import biocontrol candidates for *L. sativae* when/if it moves into key production regions. This work also suggests wasp parasitoids are not just important for pest management, but also act like 'tiny biosecurity officers', slowing the spread of exotics. There is a great opportunity to explore this service further, particularly in high risk entry and establishment areas in Australia.

Acknowledgements

We thank Dr. John La Salle for his assistance in wasp identifications. He was a dear friend to some involved in our project, and an irreplaceable source of knowledge and experience in the world of parasitic wasps. He is greatly missed.

We thank Dr. Mallik Malipatil, Luke Halling, Sally Cowan, Dr. Ryan Perry, Dr. Roger A Burks, Samantha Ward and Marianne Coquilleau for assistance with morphological identifications and provision of samples of leafminer flies and parasitoid wasps. We thank Barbara Waterhouse for assistance with host plant identifications.

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Appendix 1. Longer term average parasitism rates

Table S3. Long term parasitism rates recorded on Thursday Island in May 2019, calculated as number of mines containing signs of parasitism divided by total number of mines (dead or alive). To obtain a longer-term estimate, old mines were categorized as having previously had the emergence of a fly or a wasp, relying upon the fact that wasp emergences leave tell tail signs in the form of pupal fragment and myconial pillars (Figure S1). These signs may be visible for as long as one month after emergence. Any mines that did not have clear signs and could not confidently be categorized as fly or wasp were excluded from the analysis. To calculate the minimum parasitism rate, the number of mines containing signs of parasitism (taken as those with a wasp larvae visible on the back of a fly larvae (WL), those containing a wasp pupae (WP), and those containing signs of a wasp emergence in the past (EW)) was divided by the number of total mines (including all other mines including dead fly larvae on which no wasp larvae were visible (DL), healthy fly larvae (HL), and those containing signs of a fly emergence in the past (EF)). However, dead fly larvae on which no wasp larvae were visible could still be a result of parasitoid activity, as a result of host feeding. Thus, to calculate the maximum mortality rate as a result of parasitoid wasp activity, the number of mines containing signs of parasitism (WL + WP + EW), or containing dead fly larvae with no signs of wasp larvae, was divided by the number of total mines (including all other mines including HL + EF).

Location	Site	Host	Total mines	DL	HL	WL	WP	EF	EW	minimum % parasitism	maximum % mortality
Thursday Island	Frog Gully garden	*Macroptilium atropurpureum*	127	26	24	9	17	44	7	26%	47%
Thursday Island	Hospital playground	*Macroptilium atropurpureum*	66	18	9	5	6	15	13	36%	64%
Thursday Island	Hargrave Hill	*Stachytarpheta jamaicensis*	26	8		3	4	4	7	54%	85%
Seisia	Seaswift	*Macroptilium atropurpureum*	16					13	3	19%	19%

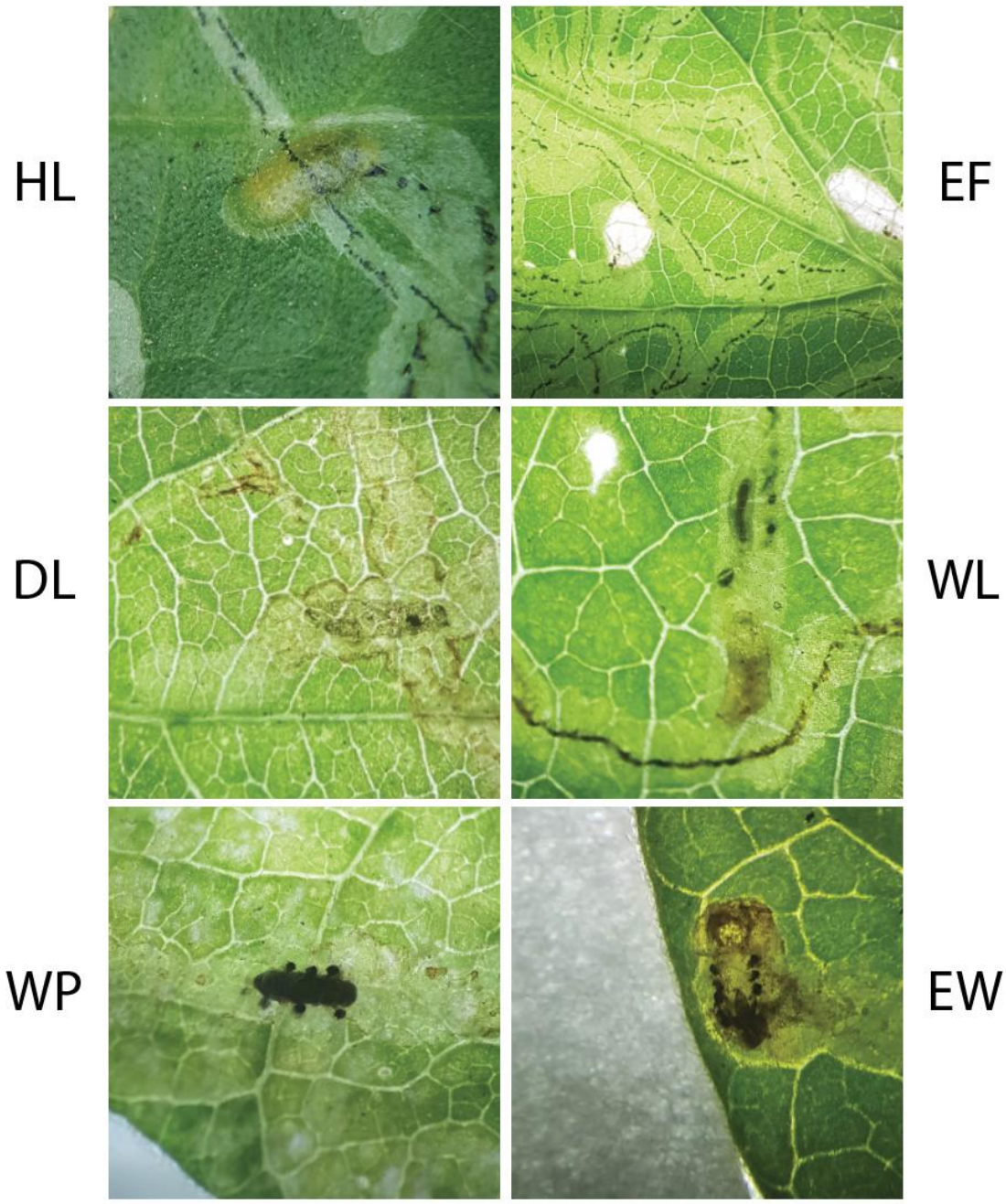


Figure S1. Signs of parasitism in empty leaf mines. HL = healthy fly larva; EF = empty mine with signs of fly emergence; DL = dead fly larva with no signs of wasp larva; WL = dead fly larva with visible wasp larva; WP = wasp pupa (with meconial pillars visible); WE = empty mine with signs of wasp emergence.

Appendix 2. Surveillance records for May 2018

Table S1. Survey records for May 2018. Confirmed observations of *L. sativae* are indicated in green and suspected (but unconfirmed) observations are indicated in yellow.

Location	Date	Lat	Lon	Plants	Signs of *L. sativae*?	Other signs of leafmining?
Cairns port	May-18	-16.95	145.77	*Macroptilium atropurpureum*	No	
	May-18	-16.95	145.77	*Macroptilium atropurpureum*	No	
	May-18	-16.94	145.77	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia alysicarpi*)
Turalba workers accommodation	May-18	-15.80	144.76	*Macroptilium atropurpureum*	No	
Lakeland caravan park	May-18	-15.86	144.85	*Macroptilium atropurpureum*	No	Lepidopteran leafmines
Cooktown Botanic Gardens	May-18	-15.47	145.26	Assorted native vegetation	No	
Lakeland Primary School	May-18	-15.86	144.86	Tomato	No	
	May-18	-15.86	144.86	Beets	No	
	May-18	-15.86	144.86	Rocket	No	
	May-18	-15.86	144.86	Lettuce	No	
	May-18	-15.86	144.86	Silverbeet	No	
	May-18	-15.86	144.86	Mint	No	
	May-18	-15.86	144.86	Sweet potato	No	Mines found and confirmed on the spot as lepidopteran (via dissection)
Lakeland organic farm #1	May-18	-15.86	144.85	Passionfruit	No	

	May-18	-15.86	144.85	Eggplant	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia solanicoli*)
	May-18	-15.86	144.85	Pumpkin	No	
Lakeland farm #2	May-18	-15.83	144.75	*Macroptilium atropurpureum*	No	Lepidopteran leafmines
	May-18	-15.83	144.75	Eggplant	No	No signs of any Agromyzid mines
	May-18	-15.83	144.75	Unidentified melon	No	
	May-18	-15.83	144.75	Marigold	No	
	May-18	-15.83	144.75	Beets	No	
	May-18	-15.83	144.75	Tomato	No	
	May-18	-15.83	144.75	Unidentified bean	No	Mines found and confirmed on the spot as lepidopteran (via dissection)
Lakeland farm #3	May-18	-15.80	144.72	Watermelon	No	
	May-18	-15.80	144.72	*Alysicarpus* weeds	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia alysicarpi*)
Coen Exchange hotel	May-18	-13.94	143.20	No hosts present	No	
Coen inspection centre	May-18	-13.77	143.13	Tomato	No	
	May-18	-13.77	143.13	Chilli	No	
	May-18	-13.77	143.13	Pumpkins	No	
Weipa campground	May-18	-12.64	141.86	Tomato	No	

	May-18	-12.64	141.86	Chilli	No	
	May-18	-12.64	141.86	Marigold	No	
	May-18	-12.64	141.86	Eggplant	No	
	May-18	-12.64	141.86	Basil	No	
	May-18	-12.64	141.86	Capsicum	No	
Weipa resident	May-18	-12.64	141.86	Marigold	No	
	May-18	-12.64	141.86	Pumpkin	No	
	May-18	-12.64	141.86	Tomato	No	
Weipa roadside Alakanum Ct	May-18	-12.64	141.86	*Alysicarpus/Desmodium* weeds	No	
Weipa roadside Alstonia St	May-18	-12.64	141.86	*Macroptilium atropurpureum*	No	
Weipa roadside Central Av	May-18	-12.64	141.87	*Macroptilium atropurpureum*	No	
Weipa roadside Duyfken Cres	May-18	-12.64	141.87	*Alysicarpus/Desmodium* weeds	No	
Weipa roadside Flinders St	May-18	-12.63	141.87	*Macroptilium atropurpureum*	No	
Weipa resident	May-18	-12.64	141.87	Marigold	No	
	May-18	-12.64	141.87	Cucumber	No	
	May-18	-12.64	141.87	Pumpkin	No	
	May-18	-12.64	141.87	Beans	No	
	May-18	-12.64	141.87	Tomato	No	
Weipa resident	May-18	-12.64	141.87	Assorted native plants	No	
Weipa roadside	May-18	-12.63	141.88	Yellow bells	No	
Weipa resident (nursery)	May-18	-12.63	141.88	Unidentified melon	No	
	May-18	-12.63	141.88	Sweet potato	No	Lepidopteran leafmines (dissection on the spot confirmed)
	May-18	-12.63	141.88	Snake bean	No	Lepidopteran leafmines (blotch style)
Weipa port	May-18	-12.67	141.86	*Macroptilium atropurpureum*	No	
	May-18	-12.67	141.86	*Macroptilium atropurpureum*	No	
	May-18	-12.67	141.86	*Macroptilium atropurpureum*	No	

	May-18	-12.67	141.86	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia alysicarpi*)
Weipa port 2	May-18	-12.67	141.87	*Macroptilium atropurpureum*	No	
	May-18	-12.67	141.87	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia alysicarpi*)
Moreton telegraph station	May-18	-12.45	142.64	Passionfruit	No	
Bramwell station	May-18	-12.14	142.62	Marigold	No	
	May-18	-12.14	142.62	Assorted ornamentals	No	
	May-18	-12.14	142.62	*Macroptilium atropurpureum*	No	
Agnara Corp. garden	May-18	-15.57	144.45	No hosts present	No	
Laura Roadhouse	May-18	-15.57	144.45	*Macroptilium atropurpureum*	No	
Hann River Roadhouse	May-18	-15.18	143.87	No hosts present	No	
Musgrave Roadhouse	May-18	-14.78	143.50	*Macroptilium atropurpureum*	No	
Bramwell Roadhouse	May-18	-12.09	142.56	No hosts present	No	
Archer River roadhouse	May-18	-13.44	142.94	Pumpkin	No	
	May-18	-13.44	142.94	Chilli	No	
	May-18	-13.44	142.94	Unidentified melon	No	
	May-18	-13.44	142.94	Unidentified bean	No	
	May-18	-13.44	142.94	New Guinea bean	No	
Jardine ferry crossing	May-18	-11.10	142.28	No hosts present	No	
Bamaga NPARC office	May-18	-10.89	142.39	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found

Bamaga Secondary School farm	May-18	-10.90	142.39	Tomato	No	
	May-18	-10.90	142.39	Chilli	No	
	May-18	-10.90	142.39	Radish	No	
	May-18	-10.90	142.39	Lettuce	No	
	May-18	-10.90	142.39	Capsicum	No	
	May-18	-10.90	142.39	Cucumber	No	
	May-18	-10.90	142.39	Bush bean	No	
	May-18	-10.90	142.39	Okra	No	
	May-18	-10.90	142.39	Watermelon	No	
	May-18	-10.90	142.39	Rockmelon	No	
	May-18	-10.90	142.39	Cabbage	No	
	May-18	-10.90	142.39	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Seisia nursery	May-18	-10.85	142.37	*Macroptilium atropurpureum*	No	
	May-18	-10.85	142.37	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Oasis accommodation	May-18	-10.85	142.37	Pumpkin	No	
	May-18	-10.85	142.37	Sweet potato	No	
	May-18	-10.85	142.37	Tomato	No	
	May-18	-10.85	142.37	Basil	No	
Seisia resident	May-18	-10.85	142.37	*Macroptilium atropurpureum*	Yes	
	May-18	-10.85	142.37	*Commelina benghalensis*	Unconfirmed	Single inactive mine found
	May-18	-10.85	142.37	Sweet potato	No	
Seisia roadside	May-18	-10.85	142.37	*Macroptilium atropurpureum*	Yes	
Seisia resident	May-18	-10.85	142.37	Bitter melon	No	
	May-18	-10.85	142.37	Ornamentals	No	
Seisia Sea Swift	May-18	-10.85	142.37	*Macroptilium atropurpureum*	Yes	
	May-18	-10.85	142.37	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Seisia Campground	May-18	-10.85	142.37	*Macroptilium atropurpureum*	Yes	
Zuna Island	May-18	-10.71	142.29	Bitter melon	No	
	May-18	-10.71	142.29	Paw Paw	No	
	May-18	-10.71	142.29	Passionfruit	No	

	May-18	-10.71	142.29	Citrus	No	
	May-18	-10.71	142.29	*Macroptilium atropurpureum*	Yes	No active mines found, DNA confirmed via empty leaf mine
	May-18	-10.71	142.29	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Albani Island	May-18	-10.73	142.60	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
	May-18	-10.73	142.60	Unidentified native tree	No	Agromyzid mines found (could be a *Tropicomyia*? Wrong mining form/pupae shape for *L. sativae*)
Roko Island	May-18	-10.74	142.41	*Scaevola taccada*	No	Agromyzid mines found (pupation inside leaf confirms it was not *L. sativae*; adults successfully reared and identified by Dr. Mallik Malipatil as *Ophiomyia cornuta*)
Horn Island	May-18	-10.59	142.25	*Macroptilium atropurpureum*	Yes	
	May-18	-10.59	142.25	*Macroptilium atropurpureum*	Yes	
	May-18	-10.59	142.25	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Harmony Garden Horn Island	May-18	-10.59	142.25	Marigold (*Tagetes* sp.)	Yes	Very heavy mining
	May-18	-10.59	142.25	Butternut pumpkin (*Cucurbita moschata*)	Yes	
	May-18	-10.59	142.25	Unidentified melon (Cucumber?)	Yes	
	May-18	-10.59	142.25	Unidentified melon (Zucchini?)	Yes	
	May-18	-10.59	142.25	Tomato (*Solanum lycopersicum*)	Yes	
	May-18	-10.59	142.25	*Stachytarpheta jamaicensis*	Yes	Very light mining, one mine with dead larvae, DNA confirmed
	May-18	-10.59	142.25	Beets	No	
	May-18	-10.59	142.25	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found

Frog Gully Garden Thursday Island	May/July 2018	-10.57	142.22	Snake bean (*Vigna unguiculata* ssp. *sesquipedalis*)	Yes	
	Jul-18	-10.57	142.22	Basil (*Ocimum basilicum*)	Yes	
	May/July 2018	-10.57	142.22	Eggplant (*Solanum melongena*)	Yes	
	May/July 2018	-10.57	142.22	Tomato (*Solanum lycopersicum*)	Yes	
	May-18	-10.57	142.22	Zinnia (*Zinnia elegans*)	Yes	
	May-18	-10.57	142.22	Cucumber (*Cucumis sativus*)	Yes	
	May-18	-10.57	142.22	Unidentified melon	Yes	
	May/July 2018	-10.57	142.22	Wild gooseberry (*Physalis angulata*)	Yes	
	Jul-18	-10.57	142.22	Passionfruit (*Passiflora edulis*)	Yes	Light mining detected in July, DNA confirmed
	Jul-18	-10.57	142.22	Chilli (*Capsicum* sp.)	Yes	Single very old mine detected, DNA confirmed
	May-18	-10.57	142.22	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Thursday Island Grand Hotel		-10.59	142.22	*Alternanthera ficoidea*	Unconfirmed	Single inactive mine found
Thursday Island Sea Swift	May-18	-10.58	142.22	*Macroptilium atropurpureum*	Yes	
	May-18	-10.58	142.22	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found
Thursday Island resident	May-18	-10.58	142.22	Thai Basil (*Ocimum basilicum*)	Yes	DNA confirmed
Thursday Island roadside	Jul-18	-10.58	142.22	*Alysicarpus/Desmodium* weeds	No	Agromyzid mines found

Appendix 3. Database of agromyzids and parasitoids

Table S2. Parasitoid wasp species reared and identified from *L. sativae* (indicated in green) and from other agromyzids during the May and July 2018 field surveys within Queensland. Associated hosts are given when an Agromyzid fly was reared from the same rearing vessel as a parasitoid wasp. Rearing vessels included: R = Rearing bag (a large sealed plastic bag containing numerous leaves); P = Petri dish (a sealed petri dish containing less than five leaves each confirmed to contain an immature fly or wasp). Collectors included: EP = Dr. Elia Pirtle; LH = Luke Halling; SC = Sally Cowan. Asterisks after a family, genus or species name indicate identification by an expert taxonomist: Dr. Mallik Malipatil for fly specimens, and Dr. Ryan Perry and Dr. Roger A Burks for wasp specimens. All other identifications were conducted by the collectors, with assistance from Dr. Peter Ridland and Samantha Ward.

Collection date	Location	Lat	Lon	Collector	Family	Genus	Species	Host plant	Associated Agromyzid host	Rearing vessel	Number
9/05/2018	Lakeland	-15.86	144.85	EP	Agromyzidae	*Ophiomyia*	*alysicarpi**	*Alysicarpus* sp.		R	8
9/05/2018	Lakeland	-15.86	144.85	EP	unknown			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	1
9/05/2018	Lakeland	-15.86	144.85	EP	Braconidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	2
9/05/2018	Lakeland	-15.86	144.85	EP	Encyrtidae*			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	4
9/05/2018	Lakeland	-15.86	144.85	EP	unknown	*Apleurotropis*	sp.*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	2
9/05/2018	Lakeland	-15.86	144.85	EP	unknown			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	1
9/05/2018	Lakeland	-15.86	144.85	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		R	1
9/05/2018	Lakeland	-15.86	144.85	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	1
9/05/2018	Lakeland	-15.86	144.85	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	1

9/05/2018	Lakeland	-15.86	144.85	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	1
9/05/2018	Lakeland	-15.86	144.85	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	1
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi**	*Alysicarpus* sp.		P	22
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	9
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	9
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	9
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	1
9/05/2018	Lakeland	-15.80	144.72	EP	unknown			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	0
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	30
9/05/2018	Lakeland	-15.80	144.72	EP	Braconidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	7
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	11
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	8
9/05/2018	Lakeland	-15.80	144.72	EP	Braconidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	6
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	3
9/05/2018	Lakeland	-15.80	144.72	EP	unknown			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	0
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	21
9/05/2018	Lakeland	-15.80	144.72	EP	Braconidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	6
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	P	2
9/05/2018	Lakeland	-15.86	144.85	EP	Agromyzidae	*Ophiomyia*	*solanicola*	*Solanum melongena*		R	16

9/05/2018	Lakeland	-15.86	144.85	EP	Braconidae	*Opius*	sp.	*Solanum melongena*	*Ophiomyia solanicola*	R	1
9/05/2018	Lakeland	-15.86	144.85	EP	Hymenoptera	*Merismomorpha*	sp.*	*Solanum melongena*	*Ophiomyia solanicola*	R	7
9/05/2018	Lakeland	-15.86	144.85	EP	Hymenoptera	*Callitula*	sp.*	*Solanum melongena*	*Ophiomyia solanicola*	R	7
9/05/2018	Lakeland	-15.86	144.85	EP	Hymenoptera	*Neochrysocharis*	sp.*	*Solanum melongena*	*Ophiomyia solanicola*	R	7
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		R	44
9/05/2018	Lakeland	-15.80	144.72	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		R	13
9/05/2018	Lakeland	-15.80	144.72	EP	Braconidae	*Opius*	sp.	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	14
9/05/2018	Lakeland	-15.80	144.72	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	8
9/05/2018	Lakeland	-15.80	144.72	EP	Hymenoptera			*Alysicarpus* sp.	*Ophiomyia alysicarpi*	R	1
12/05/2018	Weipa	-12.64	141.86	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Desmodium* sp.		P	1
12/05/2018	Weipa	-12.64	141.86	EP	Agromyzidae	*Ophiomyia*	*alysicarpi?**	*Desmodium* sp.		P	2
12/05/2018	Weipa	-12.64	141.86	EP	Agromyzidae	*Ophiomyia*	*alysicarpi?**	*Desmodium* sp.		P	1
12/05/2018	Weipa	-12.63	141.87	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	2
12/05/2018	Weipa	-12.63	141.87	EP	Eulophidae			*Alysicarpus* sp.		P	1
12/05/2018	Weipa	-12.67	141.86	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [1]		P	3
12/05/2018	Weipa	-12.67	141.86	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [1]		P	2
12/05/2018	Weipa	-12.67	141.87	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [1]		P	5
12/05/2018	Weipa	-12.67	141.87	EP	Eulophidae	*Vagus (Cirrospilus)*	*ambiguus**	*mixed hosts* [1]	*Ophiomyia alysicarpi*	P	4
12/05/2018	Weipa	-12.67	141.87	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*mixed hosts* [1]	*Ophiomyia alysicarpi*	P	1
12/05/2018	Weipa	-12.67	141.87	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*mixed hosts* [1]	no associated flies	P	1

12/05/2018	Weipa	-12.67	141.87	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Alysicarpus* sp.	no associated flies	P	1
14/05/2018	Bamaga	-10.90	142.39	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Alysicarpus* sp.	no associated flies	P	1
14/05/2018	Seisia	-10.85	142.37	EP	unknown	*Neochrysocharis*	sp.*	*Alysicarpus* sp.	no associated flies	P	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Alysicarpus* sp.	no associated flies	P	1
14/05/2018	Seisia	-10.85	142.37	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [2]		R	2
14/05/2018	Seisia	-10.85	142.37	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [2]		R	1
14/05/2018	Seisia	-10.85	142.37	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*mixed hosts* [2]		R	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*mixed hosts* [2]	no associated flies	R	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Atoposoma (Cirrospilus)*	sp.*	*Macroptilium atropurpureum*		P	5
14/05/2018	Seisia	-10.85	142.37	EP	unknown	*Apleurotropis*	sp.*	*Macroptilium atropurpureum*		P	1
14/05/2018	Seisia	-10.85	142.37	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Desmodium* sp.		P	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Macroptilium atropurpureum*		P	4
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Macroptilium atropurpureum*		P	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*		P	1
14/05/2018	Seisia	-10.85	142.37	EP	unknown			*Macroptilium atropurpureum*		P	1

14/05/2018	Seisia	-10.85	142.37	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.		P	1
14/05/2018	Seisia	-10.85	142.37	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*Desmodium* sp.	no associated flies	P	2
15/05/2018	Roko Island	-10.74	142.41	EP	Agromyzidae	*Ophiomyia*	*cornuta**	*Scaevola taccada*		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Tagetes* sp.		P	9
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Neochrysocharis*	sp.*	*Tagetes* sp.		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Cucumis sativus*		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Tagetes* sp.		P	3
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Cirrospilus*	*ambiguus*	*Tagetes* sp.		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Cucumis sativus*		P	3
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Tagetes* sp.		P	9
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Neochrysocharis*	sp.*	*Tagetes* sp.		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Tagetes* sp.		P	7
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Vagus* (*Cirrospilus*)	sp.*	*Tagetes* sp.		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Closterocerus*	*miribalis**	*Tagetes* sp.		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	unknown	*Melittobia*	*australicus / hawaiiensis**	*mixed hosts*		R	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*mixed hosts*[3]		R	1

16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Cucumis sativus*		P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Alysicarpus* sp.	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Agromyzidae	*Ophiomyia*	*alysicarpi**	*Alysicarpus* sp.	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Hemiptarsenus*	*varicornis*	*mixed hosts* [4]	no associated flies	R	2
16/05/2018	Horn Island	-10.59	142.25	EP	unknown			*mixed hosts* [4]	no associated flies	R	1
16/05/2018	Horn Island	-10.59	142.25	EP	Agromyzidae	*Ophiomyia*	*alysicarpi**	*Alysicarpus* sp.	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Alysicarpus* sp.	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*	no associated flies	P	1
16/05/2018	Horn Island	-10.59	142.25	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*	no associated flies	P	1

17/05/2018	Thursday Island	-10.58	142.22	EP	Agromyzidae	*Ophiomyia*	*alysicarpi*	*Alysicarpus* sp.	no associated flies	P	1
17/05/2018	Thursday Island	-10.58	142.22	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Cucumis sativus*	no associated flies	P	2
20/05/2018	Thursday Island	-10.58	142.22	EP	Eulophidae	*Zagrammosoma*	*latilineatum*	*Alysicarpus* sp. *or Macroptilium atropurpureum*	no associated flies	P	1
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Zagrammosoma*	*latilineatum* *	*Macroptilium atropurpureum*			
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Vagus* (*Cirrospilus*)	*ambiguus**	*Macroptilium atropurpureum*			
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Closterocerus*	sp.*	*Macroptilium atropurpureum*			
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Hemiptarsenus*	*varicornis*	*Macroptilium atropurpureum*		R	27
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Zagrammosoma*	*latilineatum*	*Macroptilium atropurpureum*		R	5
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	*Cirrospilus*	*ambiguus*	*Macroptilium atropurpureum*		R	6
16/06/2017	Horn Island	-10.59	142.25	LH	Eulophidae	Closterocerus	sp.	*Macroptilium atropurpureum*		R	3
2015	Darnley Island			LH	Eulophidae	*Hemiptarsenus*	*varicornis*	*Alysicarpus* sp.			2
2015	Darnley Island			LH	Braconidae	*Opius*	sp.	*Desmodium tortuosum*			1
2015	Boigu Island			LH	Braconidae	*Opius*	sp.	*Desmodium tortuosum*			3
2015	Boigu Island			LH	unknown	*Callitula*	sp.	*Desmodium tortuosum*			4
2015	Yorke Island			LH	unknown	*Herbertia*	sp.	*Gymnosporia inermis*			2

2015	Yorke Island	LH	Eulophidae	*Zagrammosoma*	*latilineatum*	*Ricinus communis*	1
2015	Yorke Island	LH	Hymenoptera	*Closterocerus?*		*Ricinus communis*	2
2015	Yorke Island	LH	Hymenoptera	*Closterocerus?*		*Ricinus communis*	1
2015	Yorke Island	LH	unknown	*Quadrastichus*	sp.	*Sesbania grandiflora*	1
2015	Warraber Island	LH	unknown	*Callitula*	sp.	*Desmodium tortuosum*	13
2015	Warraber Island	LH	Eulophidae	*Neochrysocharis*	sp.	*Desmodium tortuosum*	1
2015	Warraber Island	LH	Eulophidae	*Cirrospilus*	*ambiguus*	*Desmodium tortuosum*	1

[1] Rearing bag contained a mix of *Alysicarpus* sp. and *Desmodium* sp.

[2] Rearing bag contained a mix of *Alysicarpus* sp.*, Desmodium* sp. and *Macroptilium atropurpureum*

[3] Rearing bag contained a mix of *Tagetes* sp*. or Cucurbita moschata*

[4] Rearing bag contained a mix of *Alysicarpus* sp. and *Macroptilium atropurpureum*

Appendix 7: Review of chemical management options for polyphagous *Liriomyza* spp. in Australian horticultural crops

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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Table of Contents

1. Executive Summary 4

2. Introduction 9

3. Chemical options for L. sativae control 11

Table 1. Chemicals currently, or previously registered or permitted for Liriomyza spp. control worldwide. 12

3.1 Abamectin 13

Table 2. Efficacy of abamectin against different life stages of *L. sativae.* 14

Table 3. Label rates for different crop groups recommended to control *L. sativae* from abamectin products registered in the USA. 16

2.2 Cyromazine 16

Table 4. Efficacy of cyromazine against different life stages of *L. sativae.* 18

Table 5. Label rates for different crop groups recommended to control *L. sativae* from cyromazine products registered in the USA. 20

3.3 Spinosad 21

Table 6. Label rates for different crop groups recommended to control *L. sativae* from spinosad products registered in the USA. 22

Table 7. Efficacy of spinosad against different life stages of *L. sativae.* 24

3.4 Spinetoram 26

Table 8. Spinetoram registration in Australia, crop groups, pests and labelled rates. 27

3.5 Chlorantraniliprole 28

Table 9. Efficacy of chlorantraniliprole against different life stages of *L. sativae.* 29

Table 10. Label rates for different crop groups recommended to control *L. sativae* from chlorantraniliprole products registered in the USA. 31

3.6 Cyantraniliprole 32

Table 11. Label rates for different crop groups recommended to control *L. sativae* from cyantraniliprole products registered in the USA. 33

3.7 Emamectin benzoate 34

Table 12. Label rates recommended to suppress *L. sativae* from emamectin benzoate products registered in the USA. 34

Table 13. Efficacy of emamectin benzoate against different life stages of *L. sativae.* 35

3.8 Thiamethoxam 37

Table 14. Label rates for different crop groups recommended to control *L. sativae* from thiamethoxam products registered in the USA. 37

Table 15. Efficacy of thiamethoxam against different life stages of *L. sativae.* 39

3.9 Indoxacarb 41

3.10 Spirotetramat 41

3.11 Acetamiprid 42

3.12 Novaluron 43

3.13 Azadirachtin (Neem oil) 43

Table 17. Label rates for different crop groups recommended to control *L. sativae* from azadirachtin products registered in the USA. 44
Table 18. Efficacy of azadirachtin against different life stages of *L. sativae.* 45

3.14 Synthetic pyrethroids and organophosphates 50

4. Toxicity to beneficials 51
Table 19. Toxicity effect ratings on various lifecycle stages of four important parasitoid wasps of *Liriomyza spp. (Hemiptarsenus varicornis, Diglyphus isaea, Neochrysocharis formosa,* and *Opius* spp.). 53

5. Resistance status, mechanisms, cross resistance and implications for Australian control ... 56
Table 20. History of insecticide use on *Liriomyza* spp. in vegetables in Florida, USA. 57

5.1 Carbamates (1A) 58

5.2 Organophosphates (1B) 58

5.3 Cyclodiene organochlorines (2A) 59

5.4 Pyrethroids (3A) 59

5.5 Acetamiprid (4A) 60

5.6 Spinosyns (5) and diamides (28) 60

5.7 Avermectins (6) and cyromazine (17) 60

5.8 Novaluron (15) 61

5.9 Spirotetramat (23) 61

5. Pest management strategies 61

5.1 Chemical use 62

5.2 Resistance Management 63

5.3 Case Study: *L. sativae* 63
Figure 1. Chemicals presently used for pest control in Bowen and Bundaberg tomato crops (data collected based on consultation with local agronomists). 64

6. Eradication 65
Table 21. Treatment program for eradication of *L. trifolii* in glasshouse chrysanthemums. 67
Table 22. Treatment program for eradication of *L. trifolii* in plants outside of infested glasshouses. 70

7. Conclusions 71
Table 24. The active ingredients that could, or are being utilised for control of *Liriomyza* spp. present or at risk of invading Australia, and their known resistance status. 71
Table 25. A summary table detailing potential active ingredients that could be utilised for control for *Liriomyza* spp. present or at risk of invading Australia and their respective toxicity to four key parasitoids. Gaps in the table indicate areas where there is no data available. 72
Table 26. Labelled use of potential chemicals for registration for *L. sativae* control in Australia. Gaps in the table indicate areas where there are no registered products. 74

8. Recommendations 80

9. References 82

Appendix 1: Permits for the control of Liriomyza spp. on different host crops 90

1. Executive Summary

Exotic leafminer species of the genus *Liriomyza* (Diptera: Agromyzidae) represent a major risk to multiple Australian horticultural cropping sectors, particularly the vegetable, nursery and gardens, melon, potato and onion industries. There are six polyphagous *Liriomyza* flies which are considered` serious agricultural pests worldwide. They may be divided into two sub-groups: *L. huidobrensis, L. trifolii, L. sativae* and *L. brassicae*, originally discovered in the Americas, and *L. bryoniae* and *L. strigata* from Europe.

In 2015, *L. sativae* was detected on mainland Australia at the coastal town of Seisia on Cape York Peninsula. While *L. sativae* has not yet been detected in commercial horticultural production areas on the mainland, the closest commercial cropping area of Lakeland is 759 km south east of Seisia and is located on the major North-South dry season tourist route. This represents a major biosecurity risk for Australian horticulture. Two related species, *L. trifolii* and *L. huidobrensis* have also been recorded as close as Indonesia (CABI 2020). Recently, an incursion of *L. huidobrensis* was detected in NSW and QLD and eradication was subsequently deemed not technically feasible (AUSVEG 2020). All three species have broad host plant ranges and a propensity to evolve resistance to commonly used insecticides. It is therefore essential to understand the chemical and biological management of these leafminers, and to develop a portfolio of chemical permits ready for registration against these pests.

This review focuses on chemical management options for *L. sativae*, and the related exotic leafminer species, *L. trifolii* and *L. huidobrensis* currently found in South East Asia. This review has been written with a holistic view, taking into consideration several aspects of chemical control for *L. sativae, L. trifoli* and *L. huidobrensis*, resistance management planning, the role of beneficial wasps, and chemistries that may be used in a management and under an eradication scenario. We have also included data on the efficacy of insecticides on life stages for *L. sativae*, given its current close proximity to Australian agricultural regions.

Different chemicals have different levels of efficacy on *Liriomyza* spp. While some chemicals are effective as knockdown foliar sprays that kill adults (e.g. organophosphates), other products have translaminar activity (e.g. abamectin, spinosad, chlorantraniliprole and cyantraniliprole). For these translaminar chemistries, a foliar spray will not only kill adults but can also penetrate the leaf surface to target and kill the larval stages. Chemicals that have systemic activity (e.g. cyromazine, azadirachtin) can be applied as a soil drench, a seedling drench, or through drip irrigation, and will persist in the soil substrate and travel throughout the plant, targeting the pupal and larval stages. A multi-pronged management strategy that targets different leafminer life stages is needed, with a particular focus on chemicals that have translaminar and/or systemic activity, as the larvae develop entirely inside the protection of the leaf.

Two of the most effective insecticides for *Liriomyza* management are abamectin (IRAC Group 6A) and cyromazine (IRAC Group 17). These insecticides target the larvae inside the plant foliage as they both

have translaminar properties. Both insecticides have seen a long history of use, yet resistance has not been a major problem.

Resistance to synthetic pyrethroids has been observed in *L. sativae* populations in Vietnam. It is likely that Australian populations will either have resistance to synthetic pyrethroids or have the potential to evolve this resistance readily, as the likely pathway for entry into Australia is probably via Vietnam. There is also potential that other types of resistance may evolve in Australian *L. sativae* populations if care is not taken with chemical management. Therefore, it will be important to register several chemicals, from different IRAC MoA groups, to allow a resistance management strategy of chemical rotation to be put in place. Notably, *L. huidobrensis* and *L. trifolii* often display greater resistance than *L. sativae*.

As well as the current minor use permit for control of *L. sativae* using abamectin, there are several chemicals now permitted for *Liriomyza* spp. control in Australian crops, including emamectin benzoate, cyromazine, spirotetramat, spinetoram and dimethoate. There is also the potential for non-target control by other chemicals registered and used against other crop pests. However, registration of a larger range of chemicals with different MoA's, at a variety of rates, is needed in crops that are likely to be highly impacted by *L. sativae* populations. Pea and bean, leafy vegetable crops, and celery crops will be economically impacted both cosmetically as well as through yield reduction, and therefore new registrations of effective chemistries are necessary for these crops. The top five candidate chemicals for extended or new permit applications for *L. sativae* control are abamectin, emamectin benzoate, spinetoram, cyantraniliprole and indoxacarb.

Due to the cryptic nature of *Liriomyza* spp. immature life stages and the difficulty of targeting these with chemicals, as well as the potential for resistance evolution, beneficial organisms should be viewed as an essential part of any management strategy. Management plans across different crop groups should consider the suite of beneficials, especially parasitoid wasps. Ideally, chemistries that are less toxic to these beneficials will be selected for *L. sativae* control, however it will be important to have registrations for some of the less selective chemicals as well. Timing the use of the less selective chemicals to periods when parasitoid wasps are less likely to be active will lessen the impact of those chemicals on beneficial populations.

Eradication strategies may play a role if there are small outbreaks of *L. sativae* in isolated Australian horticultural cropping regions. Depending on the situation, eradication may not be cost beneficial and is less likely to be effective in warmer regions where *L. sativae* will have a shorter life cycle and host abundance is high. However, if outbreaks occur in southern Australia where winter temperatures will slow *L. sativae* population growth, swift implementation of an eradication strategy may help provide area freedom in certain circumstances (continued awareness activities will be important for early detection of *L. sativae* in those regions where eradication is more likely). In addition, it will be important to include weeds and roadside verges in the registration of any chemicals being considered for *L. sativae* eradication.

A summary of the information contained in this review is presented in the following table. This includes: pesticide toxicity against beneficials which attack *Liriomyza* spp., recorded *Liriomyza* spp. resistance and existing permit or label registration for potential host crops for *Liriomyza* spp. in Australia. Gaps in the table indicate areas where there are no data readily available and/or products are not registered in Australian crops.

Chemical	**MoA Group**	**Toxicity to beneficials**	**Resistance recorded for *Liriomyza* spp.**	**Labels/permits in Australia for *Liriomyza* or any other pest**	
				Food crops	**Non-food crops**
Organophosphates	1B	H-VH	Y	Y	O
Synthetic pyrethroids	3A	H-VH	Y	O	O
Acetamiprid	4A	L-H	N	N	O
Thiamethoxam	4A	L-H	Y	O	O
Spinosad	5	M-VH	Y	O	O*
Spinetoram	5	H-VH	Y	Y	O
Abamectin	6	L-VH	Y	Y	O
Emamectin benzoate	6	M-VH	N	Y	Y
Novaluron (IGR)	15	L-M	N	N	O
Cyromazine (IGR)	17	L-M	Y	Y	O
Indoxacarb	22A	L-M	N	O	O
Spirotetramat	23	L-M	N	Y	N
Cyantraniliprole	28	L-M	Y	O	O
Chlorantraniliprole	28	L-H	Y	O	O
Azadirachtin	-	L-H	N	N	O

Toxicity to beneficials: to account for variability in mortality between laboratory and field trials, pesticide toxicity ratings are categories into L, M, H and VH according to IOBC protocols, where observed mortality in laboratory and extended laboratory trials correspond to <30%, 30-79%, 80-98%, and >99% mortality, respectively, and semi-field and field trials correspond to <25%, 25-50%, 50-75%, and >75% mortality, respectively. Resistance: Y = yes, resistance recorded, N = no; no data available. Labels/permits in Australia: Y = there exists a permit/label for *Liriomyza* spp. for this chemical in Australia, O = there exists a permit for OTHER pests for this chemical in a *Liriomyza* spp susceptible crop(s), (thus it will be reasonably straightforward to obtain a permit that covers *Liriomyza* spp.*)*, N = there are no permits for ANY pests for this chemical in a *Liriomyza* spp susceptible crop(s), (thus it may be difficult to obtain a permit that covers *Liriomyza* spp). * Permitted as the insecticide in a bait concentrate.

During the course of this project, the following permits have been approved or are in preparation for the control of exotic *Liriomyza* spp.:

- Abamectin PER81876*.* For use against *L. sativae* in all Australian vegetable cropping regions; for use by anyone (rather than specialised personnel). Also extending this permit to cover lettuce crops, leafy brassica crops, and snow and sugar snap peas. Suitable for ongoing management.
- Emamectin benzoate PER87563. For use against *Liriomyza* spp. Suitable for ongoing management.
- Cyromazine PER83506/PER81867. For use against *L. sativae and L. huidobrensis* in selected vegetable crops, and for use against *Liriomyza* spp. in nursery stock and fruit trees. Note that in some cases, the crop needs to be destroyed so is only suitable for eradication in those situations. On other crops, it can be used for ongoing management.
- Spirotetramat. PER88640. For use against *Liriomyza* spp. Suitable for ongoing management.
- Spinetoram PER87878. For use against *Liriomyza* spp. Suitable for ongoing management.
- Cyantraniliprole. A permit has been drafted but has not been submitted to the APVMA to date. This permit was reviewed by the manufacturer who provided data to support its use.
- Diazinon. A draft application for a minor use permit has been developed but this has not been submitted to the APVMA as the product is not considered a good fit for inclusion in an IPM program. therefore, its use would only be for emergency/eradication purposes.
- Chlorpyrifos – A draft application for a minor use permit has not been developed as the product is not considered a good fit for use in an IPM program.
- Dimethoate PER89184 (secured via a GRDC funded project). For use against *L. sativae, L. trifolii* and *L. huidobrensis*. Suitable for ongoing management.

Recommendations from this review include:

- Apply for a minor use permit for cyromazine for the control of *L. trifolii* in vegetable crops were already labelled for use against *L. sativae* and *L. huidobrensis*.
- Apply for a minor use permit for abamectin for the control of *L. trifolii* and *L. huidobrensis* in vegetable crops were already labelled for use against *L. sativae.*
- Applying for minor use permits for indoxacarb for the control of *Liriomyza* spp. in vegetable crops where these products already have labelled uses.
- Applying for minor use permits for novaluron for the control of *Liriomyza* spp.
- Preparation of eradication permits (or ensure minor use permits include relevant information) for cyantraniliprole, spinetoram, thiamethoxam and several organophosphates (chlorpyrifos and diazinon). Chemical rates may be higher for the eradication permits compared with minor use permits, as crop destruction will form part of the eradication process. These permits should be linked to the contingency plans for *L. sativae, L. trifolii* and *L. huidobrensis*.
- Gathering data on current chemical management practices in crops that are economically vulnerable to *L. sativae*, *L. trifolii* and *L. huidobrensis* infestation in different growing regions in order to assess the non-target effects of these current practices on leafminer and parasitoid populations.
- Preparing region-specific management guidelines for *L. sativae*, *L. trifolii* and *L. huidobrensis* including the role of parasitoids, chemical rates and application methods for insecticides, selectivity of different chemistries against important beneficial species, cultural control methods, and strategies to reduce the risk of resistance evolution (currently underway).

- Genetic or phenotypic investigations of the resistance status of *L. sativae* and *L. huidobrensis* populations within Australia.

2. Introduction

Globally, the genus *Liriomyza* (Diptera: Agromyzidae), which contains members of the leafmining flies, are amongst the most economically important pests of vegetable and floriculture crops (Reitz, Gao, & Lei, 2013). There are currently approximately 440 species in the genus (Lonsdale, 2011, 2017; ITIS, 2016), although only 24 species are considered of economic importance (Spencer, 1973). Three species, *L. huidobrensis* (Blanchard), *L. sativae* Blanchard, and *L. trifolii* (Burgess), are perhaps the most significant. The success of these species as agricultural pests is linked with their biology and response to insecticides. These species are all highly polyphagous, have a high reproductive capacity, cause extensive damage in impacted crops, are highly invasive, and have become established in many agricultural regions throughout the world (Reitz et al., 2013). These species also have very fast developmental rates, with a generation completed in fewer than 20 days at optimal temperatures (Lanzoni, Bazzocchi, Burgio, & Fiacconi, 2002; Parkman, Dusky, & Waddill, 1989; Haghani, Fathipour, Talebi, & Baniameri 2006, , 2014). Thus, these species can complete multiple overlapping generations in a single cropping season. Perhaps, one of the increasingly important factors in their success as pests, is their ability to evolve resistance to insecticides (Parrella & Keil, 1984).

In 2015, *L. sativae* was detected at Seisia on the Cape York Peninsula in Australia. *L. sativae* is currently under control within the Cape York Peninsula Pest Targeted Quarantine Area. To date, the pest has not been detected outside the quarantined area. *L. huidobrensis* was detected in October 2020 in NSW and has since been found in QLD. Consultative Committee for Emergency Plant Pests has determined that *L. huidobrensis* is not technically feasible (AUSVEG 2020). *L. trifolii* is not currently present in Australia.

Jovicich (2009) modelled the potential future geographic distribution of *Liriomyza* spp. that pose high risks to Australian agriculture using CLIMEX v2 software. *Liriomyza sativae* and *L. trifolii* are both cold-intolerant, while *L. huidobrensis* is more cold-tolerant. All three species are therefore likely to overlap in their range in the east, south-east, south-west and elevated cooler regions of the north-east of Australia.

Liriomyza sativae is able to colonize a wide range of plants from nine plant families, while *L. huidobrensis* is reported from 14 plant families and *L. trifolii* the largest number of any *Liriomyza* spp., from 25 (Spencer, 1990; Appendices 1-3). Adult female *Liriomyza* puncture abaxial and adaxial leaf surfaces with their ovipositor to create sites for both sexes to feed on the plant exudates as well as oviposition sites (Bethke & Parrella, 1985; Weintraub et al., 2017). The plant volatiles induced by these punctures have recently been shown to promote the mating success of *L. huidobrensis* by attracting both sexes to host leaves (Ge, Li, Yang, Wei, & Kang, 2019).

The most economically important damage from these flies occurs from the mining activity of the larvae (Reitz et al., 2013). The female fly lays an egg in a puncture wound on a leaf. The egg hatches and the resulting larva feeds on the mesophyll inside the leaf. Larval feeding creates a mine between the upper

and lower leaf surfaces. *Liriomyza huidobrensis* can reportedly cause the most direct damage on host plants due to the large mines it creates in the spongy mesophyll of foliage and in petioles (Parrella, Jones, Youngman, & Lebeck, 1985). As the larva continues to feed, it tunnels in a twisting pattern through the leaf, and the mine expands in width as the larva matures. Just before pupation, the larva cuts an opening to exit its mine, and usually drops to the ground to pupate (Gao, Reitz, Xing, Ferguson, & Lei, 2017). Indirect effects may be experienced by growers because of quarantine restrictions that constrain international market access (Gitonga, Chabi-Olaye, Mithöfer, Okello, & Ritho, 2010). Feeding punctures in the plant leaf are small and generally of little concern except under high population pressure or in situations where the aesthetic value of a crop may be compromised (Reitz et al., 2013). Further, these feeding punctures have been shown to act as entry points for plant pathogenic bacteria (Matteoni & Broadbent, 1988) and fungi (Deadman, Khan, Thacker, & Al-Habsi, 2002). Transmission of several potyviruses has been demonstrated in the glasshouse via the contaminated ovipositors of females moving from plant to plant (Zitter & Tsai, 1977). Without control measures, losses of up to 100% of affected vegetable seedlings may occur in nurseries or in the field, however larger plants can sustain a considerable amount of leaf mining before there will be a significant reduction in yield (Jovicich, 2009). Exact levels of yield reduction are extremely hard to predict from pest damage symptoms, as old damage remains on the plant, so tends to be counted. Previous work has shown that *L. sativae* can cause losses of up to 70% in field-grown tomatoes and 40-70% in squash yields (Sharma, Durazo, & Mayberry, 1980).

Liriomyza spp. are considered secondary pests, as they are released from natural control when their enemies are eliminated (Metcalf & Luckmann, 1994). Parasitoid populations are able to reduce pest numbers below economic thresholds and when they are reduced in agroecosystems, outbreaks of *Liriomyza* spp. occur (Oatman & Kennedy, 1976; Ohno, Ohmori, & Takemoto, 1999). Consequently, there is a continual interest in identifying insecticides with low toxicity to *Liriomyza* parasitoids. As the different life stages of *L. sativae* inhabit very different niches, different chemical control methods are needed for adults, pupae, larvae and eggs. Contact insecticides which may kill adult *L. sativae* will be less effective against larvae and pupae, whereas systemic and translaminar insecticides can play a role in controlling the larval stages.

This review reports on the chemical control options and consolidates the relevant information in preparation for appropriate chemical permits for ongoing management of the vegetable leafminer (*L. sativae*) in Australian horticultural crops, including vegetables, melons, ornamentals and nursery stock (both vegetable and non-food seedlings). This review also looks at the chemical options in preparedness of an incursion by *L. trifolii* or *L. huidobrensis*. We also address the potential for insecticide resistance, the effect of chemicals on beneficial species, and investigate eradication for incursions into growing regions.

3. Chemical options for *L. sativae* control

The various life stages of *Liriomyza* spp. require different insecticidal modes for effective control. While contact insecticides are effective against adults (e.g. organophosphates), products with translaminar activity (e.g. abamectin, spinosad, cyantraniliprole) may be effective against both adults, and larvae, due to their leaf penetrance. Products with systemic activity (e.g. cyromazine, thiamethoxam) that can be applied as a soil drench, a seedling drench, or through drip irrigation, will persist in the soil substrate and travel throughout the plant, targeting the pupal and larval stages.

Over sixty different chemical products including insecticides, biological agents (e.g. entomopathogenic fungi), paraffinic, petroleum and plant oils, and mineral products are registered internationally for the control of *Liriomyza* leafminer species (Table 1). These chemicals span 18 different IRAC MoA groups and have varying levels of efficacy on the different life stages of leafminers. The majority of registered products are foliar sprays, with some products also registered for soil drenches or drip irrigation delivery, seed treatments, and seedling drenches. For effective insecticide resistance management, it is important to rotate chemicals from a range of MoA groups. Insecticides were also assessed based on their known efficacy against *Liriomyza* spp. and those actives that are already available within Australia. Also, as parasitoids can play an essential role in controlling leafminer populations (Hidrayani, Purnomo, Rauf, Ridland & Hoffmann, 2005; Weintraub et al. 2017), it is important to consider the selectivity of insecticides. Considering all of these factors and the fit with current pest management systems in Australia, thirteen chemical actives from eight different MoA groups were identified that are likely to be most effective in ongoing management of *L. sativae* in Australia: thiamethoxam and acetamiprid (IRAC Group 4A), spinosad and spinetoram (IRAC Group 5), abamectin and emamectin benzoate (IRAC Group 6), novaluron (IRAC Group 15), cyromazine (IRAC Group 17), indoxacarb (IRAC Group 22A), spirotetramat (IRAC Group 23), chlorantraniliprole and cyantraniliprole (IRAC Group 28) and azadirachtin (IRAC group UN).

Table 1. Chemicals currently, or previously registered or permitted for Liriomyza spp. control worldwide.

Chemical Group	**MoA group**	**Active ingredients**
Carbamates	1A	carbaryl, methomyl, oxamyl
Organophosphates	1B	acephate, chlorpyrifos, diazinon, dimethoate, malathion, methidathion, oxydemeton-methyl, phosmet, methamidophos, phorate, pyrazophos
Pyrethroids, Pyrethrins	3A	alpha-cypermethrin, beta-cyfluthrin, bifenthrin, cyfluthrin, cypermethrin, deltamethrin, esfenvalerate, fenpropathrin, gamma-cyhalothrin, lambda-cyhalothrin, permethrin, pyrethrins, zeta-cypermethrin
Neonicotinoids	4A	acetamiprid, clothianidin, dinotefuran, imidacloprid, thiacloprid, thiamethoxam
Butenolides	4D	flupyradifurone
Spinosyns	5	spinetoram, spinosad
Avermectins	6	abamectin, emamectin benzoate
Juvenile hormone analogues	7A	s-kinoprene
Pyriproxyfen	7C	pyriproxyfen
Nereistoxin analogues	14	thiosultap-sodium (dimehypo)
Benzoylureas	15	diflubenzuron, novaluron
Cyromazine	17	cyromazine
Diacylhydrazines	18	methoxyfenozide, tebufenozide
METI acaricides and insecticides	21A	fenpyroximate, tolfenpyrad
Oxadiazines	22A	indoxacarb
Tetronic and Tetramic acid derivatives	23	spirotetramat
Diamides	28	chlorantraniliprole, cyantraniliprole, flubendiamide
Oils and Plant extracts*	UN	azadirachtin (neem oil), paraffinic oil, petroleum oil, mineral oil, cedar oil, rosemary oil, peppermint oil, geraniol, extract of *Chenopodium ambrosiodes*
Biologicals*	UNF	*Chromobacterium subtsugae* strain PRAA4-It, *Isaria fumosorosea*, *Burkholderia* spp. Strain A396
Minerals*	UNM	diatomaceous earth (silicon), kaolin clay

* Compounds of unknown or uncertain MoA. Information collated from the Australian Pesticides and Veterinary Medicines Authority (APVMA; https://apvma.gov.au/, accessed 23 January 2020), Green Book (https://www.greenbook.net/, accessed 30 January 2020).

3.1 Abamectin

Abamectin is a member of the Avermectins (IRAC Group 6A). Abamectin is a mixture of avermectins containing avermectin B1a and avermectin B1b. These two components are insecticidal or anthelmintic compounds derived from the soil bacterium *Streptomyces avermitilis*. It is a neurotoxin that acts as a γ-aminobutyric acid (GABA) agonist and has translaminar activity (Lasota & Dybas, 1991; Gao et al., 2017). Most formulated products containing abamectin are of low toxicity to mammals and are not readily absorbed through the skin. Abamectin is nearly insoluble in water and has a strong tendency to bind to soil particles and is therefore quite immobile in soil and unlikely to leach or contaminate groundwater; it is rapidly degraded in soil, water, and undergoes rapid photodegradation.

Abamectin has high efficacy against *Liriomyza* species and is used widely overseas on various crops (e.g. Seal, Schuster, & Klassen, 2007; Weintraub & Horowitz, 1998). Due to the translaminar activity of abamectin, this product is particularly effective at controlling the larval stages of *L. sativae* (Table 2). A trial on celery against *L. huidobrensis* which compared synthetic insecticides (AGRI-MEK 0.15 EC (abamectin 1.9%), CITATION 75 WP (cyromazine 75%), TRACER 480 SC (spinosad 480 g/L), SYLGARD 309 (siloxylated polyether 76% + surfactant mixture 24%) and nematode (NEMASYS F (live nematodes, 50 million per pack) efficacy, showed that application of the high rates of abamectin, spinosad, or cyromazine showed the best control both in terms of mining and stippling damage (Hallet, Heal, & Bahlai, 2004). At harvest, application of all treatments, except the low rate of TRACER, produced significantly higher quality celery bunches than were harvested in the control plots (Hallet et al., 2004).

Abamectin is registered in a wide range of agricultural crops in Australia, mainly for the control of two-spotted mites. It is also permitted for use against *Liriomyza sativae* and *Liriomyza huidobrensis* in several agriculturally important hosts (Appendix 1). Additionally, a permit, which covers several actives including abamectin, is available for use against *Liriomyza* spp. leafminer in non-food nursery stock (Appendix 1). The rate ranges of abamectin registered in Australia are similar to those recommended for leafminer control in abamectin products registered in the USA (Table 3).

Table 2. Efficacy of abamectin against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Average mortality (SE)**	**Ref.***
Susceptible	Abamectin	Tween-80®	Foliar	larvae	0.04 (0.03 - 0.04)			a
Chorpyrifos resistant	Abamectin	25% petroleum oil	Foliar	larvae	0.04 (0.03 - 0.05)			a
Field collected	Abamectin	no	Foliar	1st instar	1.51 (1.19 - 1.89)			b
Field collected	Abamectin	no	Foliar	3rd instar	1.81 (1.4 - 2.2)			b
Field collected	Abamectin	no	Foliar	Adult	14.3 (9.5 - 19.4)			b
Field collected	Abamectin	25% petroleum oil	Foliar	1st instar	0.5 (0.1 - 0.8)			b
Field collected	Control	no		eggs			0.86 (0.65)	c
Field collected	Abamectin	no	Foliar	eggs		36	85.8 (1.09)	c
Field collected	Abamectin	no	Foliar	eggs		72	92.8 (0.95)	c
Field collected	Control	no		larvae			1.9 (0.89)	c

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Average mortality (SE)**	**Ref.***
Field collected	Abamectin	no	Foliar	larvae		36	100 (0)	c
Field collected	Abamectin	no	Foliar	larvae		72	100 (0)	c
Field collected	Control	no	14 days residual (lab)	larvae		0	1.8 (0.8)	c
Field collected	Abamectin	no	14 days residual (lab)	larvae		36	100 (0)	c
Field collected	Abamectin	no	14 days residual (lab)	larvae		72	100 (0)	c
Field collected	Control	no	14 days residual (field)	larvae		0	1.1 (0.7)	c
Field collected	Abamectin	no	14 days residual (field)	larvae		36	77.1 (0.8)	c
Field collected	Abamectin	no	14 days residual (field)	larvae		72	85.7 (0.9)	c

* Data taken from (a) Askari Saryazdi, Hejazi, Ferguson, & Rashidi, (2015), (b) Saberfar, Sheikhi Garjan, Naseri, & Rashid, (2012), and (c) Hossain & Poehling, (2009).

Table 3. Label rates for different crop groups recommended to control *L. sativae* from abamectin products registered in the USA.

Crop	**Proposed rate for control**	**WHP**	**Example products**
Cucurbit vegetables, Dry Beans, Fruiting vegetables, Herbs, Onions, Bulb vegetables, Potatoes, Tuberous and Corm vegetables, Leafy vegetables (except Brassica vegetables)	11 - 21 g a.i./ha	7 days 14 days Chives, Potatoes, Tuberous and Corm Vegetables 30 days Onions, Bulb crops	Epi-Mek® 0.15 EC (18 g/L) Reaper® ClearForm® (18 g/L) AgMectin® 0.15 EC (18 g/L) Borrada™ (18 g/L)
Ornamentals	11 g a.i./ha	N/A	Phoenix Merlin™(18 g/L) Ardent™ 0.15 EC Miticide/Insecticide (18 g/L)
Head Lettuce	13 - 16 g a.i./ha	7 days	Athena™ Insecticide (13 g/L)*
Soybeans	100 g a.i./100kg seed		Avicta® Complete Beans 500 (247 g/L)*
Spinach	8 - 16 g a.i./ha	40 days	Athena™ Insecticide (13 g/L)*

* Product contains a mixture of abamectin and other insecticides. Only the rates for abamectin are shown in this table.

2.2 Cyromazine

Cyromazine is an insect growth regulator (IRAC Group 17) that interrupts the development of larval stages. It has specific activity against dipterous larvae, and also has translaminar activity so it can target life stages living inside foliage (Yathom, Ascher, Tal, & Nemny, 1986; Gao et al., 2017). It can be applied as a soil drench or a foliar spray and is very efficacious at controlling the immature life stages of *L. sativae* but is less effective at controlling the adults (Table 4). It is almost non-toxic to mammals and birds, is rapidly degraded in soil and water, and undergoes rapid photodegradation. It is also stable to hydrolysis and has a low to moderate mobility in sandy or loam sand soils and is therefore unlikely to leach or contaminate groundwater.

Cyromazine is considered one of the most effective chemical control options for *Liriomyza* spp. (Leibee & Capinera, 1995; Weintraub, 1999; Reitz et al., 2013). A number of cyromazine products are registered in a variety of food crops for the control of *Liriomyza* spp. in the USA (Table 5).

Cyromazine is approved for the control of dipteran pest species that infest mushroom compost and affect animal husbandry in Australia, and several more recent permits include the use of cyromazine to control *Liriomyza* spp. larvae on non-food nursery stock and ornamentals, and to control *L. sativae* and *L. huidobrensis* on a variety of vegetable crops (Appendix 1).

Table 4. Efficacy of cyromazine against different life stages of *L. sativae*.

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Average score (SE)**	**Ref.***
Susceptible	Cyromazine	Tween-80®	Foliar	larvae	1.36 (1.23 - 1.51)			a
Chlorpyrifos resistant	Cyromazine	Tween-80®	Foliar	larvae	1.15 (1.02 - 1.28)			a
Lab culture	Cyromazine	Tween-80®	Foliar	1st instar	0.49 (0.43 - 0.56)			b
Lab culture	Cyromazine	Tween-80®	Foliar	larvae		0.25	75% (3.7) pupation	b
Lab culture	Control	no	Foliar	larvae			91.6% (2.3) pupation	b
Lab culture	Cyromazine	Tween-80®	Foliar	pupae		0.25	52.9% emergence	b
Lab culture	Control	no	Foliar	pupae			83.2% emergence	b
Field collected	Cyromazine	no	Foliar	1st instar	34.8 (24.9 - 46.2)			c
Field collected	Cyromazine	no	Foliar	3rd instar	38.4 (30.1 - 50.8)			c

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Average score (SE)**	**Ref.***
Field collected	Cyromazine	no	Foliar	Adult	1295 (864-1716)			c
Field collected	Cyromazine	25% oil	Foliar	1st instar	32.2 (25.2-46.2)			c
Field collected	Cyromazine	no	Foliar	oviposition		83	21.8 (11.0) eggs laid	d
Field collected	Control	no		oviposition			49.3 (5.0) eggs laid	d
Field collected	Cyromazine	no	Foliar	feeding		83	398 (82.8) punctures	d
Field collected	Control	no		feeding			444.9 (49.9) punctures	d
Field collected	Cyromazine	no	Foliar	larvae		83	94.3 - 99.2% mortality	d
Field collected	Cyromazine	no	Foliar	adults		83	2.5% mortality	d

* Data taken from (a) Askari Saryazdi et al., (2015), (b) Khorshidi, Hejazi, & Iranipour, (2017), (c) Saberfar et al., (2012) and (d) Tokumaru, Kurita, Fukui, & Abe, (2005).

Table 5. Label rates for different crop groups recommended to control *L. sativae* from cyromazine products registered in the USA.

Crop	**Proposed rate for control**	**WHP**	**Example products**
Capsicums, Tomatoes	140 g a.i./ha	0 days	Citation (750g/kg) Trigard (750g/kg)
Brassica Leafy Vegetables, Bulb Vegetables, Leafy Vegetables, Beans, Succulent and Dried Varieties, Cucurbits	140 g a.i./ha	7 days	Citation (750g/kg) Trigard (750g/kg)
Landscape ornamentals, container-grown ornamentals, greenhouse-, lathhouse-, and shadehouse-grown ornamental bedding plants and ornamental crops; and interiorscapes; and shore flies, in greenhouse ornamental crops and interiorscapes only	140 g a.i./ha	N/A	Citation (750g/kg)
Potatoes	140 g a.i./ha	17 days	Trigard (750g/kg)

3.3 Spinosad

Spinosad is a member of the Spinosyns group (IRAC Group 5). It is a bacterially derived insecticide consisting of two active compounds, spinosyns A and D. A member of the spinosyns group of nicotinic acetylcholine receptor (nAChR) allosteric modulators, spinosad is a broad spectrum, non-cumulative insecticide with contact and ingestion activity. It has a unique mode of action on the insect nervous system (Thompson, Dutton, & Sparks, 2000), killing insects through hyper-excitation of the nervous system. It also has translaminar, or local, systemic activity, penetrating leaf tissues and forming a reservoir of active ingredient within the leaf. Spinosad is readily degraded on soil or plants, primarily through photo- and microbial degradation (Thompson et al., 2000). It is approved for the control of several insect pest species in fruit crops and fruit trees, post-harvest/stored cereal grain, and some vegetables and fruits as well as animal husbandry in Australia. Appendix 1

A number of spinosad products are registered for the control of *Liriomyza* spp. in the USA (Table 6). Spinosad is efficacious at controlling the adult, larval and the egg life stages of *L. sativae* when applied as a foliar spray (Table 7), and also shows systemic effects against *L. huidobrensis* (Weintraub & Mujica, 2006). The use of a penetrating surfactant or mineral oil can increase the efficacy of spinosad significantly (Table 7), and 0.25 to 0.5% v/v emulsified crop oils or methylated crop oil plus organosilicone combination products are recommended on product labels in the USA, particularly for leafy vegetables. The spinosad product Entrust® is listed by the Organic Materials Review Institute for use in organic production in the USA (Table 7).

Table 6. Label rates for different crop groups recommended to control *L. sativae* from spinosad products registered in the USA.

Crop	**Proposed rate for control**	**WHP**	**Example products**
Cotton	75 - 102 g a.i./ha	28 days	Tracer (480 g/L) Entrust (240 g/L) Blackhawk (360 g/kg)
Quinoa	53 - 105 g a.i./ha	21 days	Entrust (240 g/L)
Bulb Vegetables	53 - 105 g a.i./ha	1 day	Success (240 g/L) Entrust (240 g/L)
Cucurbits	105 - 140 g a.i./ha	3 days 1 day (Cucumbers)	Success (240 g/L) Entrust (240 g/L)
Fruiting Vegetables, Okra and Leafy Vegetables	105 - 175 g a.i./ha	1 day	Success (240 g/L) Entrust (240 g/L)
Leaves of Legume Vegetables, Leaves of Root and Tuber Vegetables, Turnip Greens and Watercress	105 - 175 g a.i./ha	3 days	Success (240 g/L) Entrust (240 g/L)
Legume Vegetables	79 - 105 g a.i./ha	3 days (Beans, peas) 28 days (Dried legumes)	Success (240 g/L) Entrust (240 g/L) Blackhawk (360 g/kg)
Peppermint and Spearmint	70 - 175 g a.i./ha	7 days	Success (240 g/L) Entrust (240 g/L)
Root and Tuber Vegetables	70 – 105 g a.i./ha 79 – 175 g a.i./ha	2 days 3 days 7 days	Success (240 g/L) Entrust (240 g/L) Blackhawk (360 g/kg)
Spices (except Black Pepper)	105 - 175 g a.i./ha	14 days	Success (240 g/L) Entrust (240 g/L)
Ornamentals (Herbaceous and Woody), Growing Outdoors, in Nurseries (Including Conifer Seed	509 – 771 g a.i./ha	N/A	Entrust (240 g/L) Conserve (120 g/L)

Crop	**Proposed rate for control**	**WHP**	**Example products**
Orchards), or in Greenhouses			
Herbs	1.5 g a.i./L		Conserve (120 g/L)
Brassica vegetables	70 - 175 g a.i./ha	1 day	Success (240 g/L) Entrust (240 g/L)
Home Garden Use	1.5 g a.i./L	See commercial WHP	Conserve (120 g/L)

Table 7. Efficacy of spinosad against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Mortality % (SE)**	**Ref.***
Susceptible	Spinosad	Tween-80®	Foliar	larvae	0.83 (0.76 - 0.93)			a
Chlorpyrifos resistant	Spinosad	Tween-80®	Foliar	larvae	1.28 (1.14 - 1.46)			a
Field collected	Spinosad	no	Foliar	1st instar	4.4 (3.4 - 5.3)			b
Field collected	Spinosad	no	Foliar	3rd instar	12.1 (2.9 - 19.2)			b
Field collected	Spinosad	no	Foliar	Adult	13.7 (8.1-20.7)			b
Field collected	Spinosad	25% oil	Foliar	1st instar	1.9 (0.9-2.6)			b
Field collected	Control	no	Foliar	larvae		0	22.6 (1.3)	c
Field collected	Spinosad	no	Foliar	larvae		240	99.4 (0.3)	c
Field collected	Control	no	Foliar	eggs		0	0.9 (0.7)	d
Field collected	Spinosad	no	Foliar	eggs		240	0.5 (0.6)	d
Field collected	Spinosad	no	Foliar	eggs		480	1.4 (0.7)	d
Field collected	Control	no	Foliar	3rd instar		0	1.9 (0.9)	d
Field collected	Spinosad	no	Foliar	3rd instar		240	93.7 (0.6)	d
Field collected	Spinosad	no	Foliar	3rd instar		480	97.7 (1.3)	d
Field collected	Control	no	14 days residual (lab)	larvae		0	1.8 (0.8)	d

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**LC_{50} mg a.i./L (95% CI)**	**Conc. mg a.i./L**	**Mortality % (SE)**	**Ref.***
Field collected	Spinosad	no	14 days residual (lab)	larvae		240	91.4 (1.2)	d
Field collected	Spinosad	no	14 days residual (lab)	larvae		480	97.7 (1.3)	d
Field collected	Control	no	14 days residual (field)	larvae		0	1.1 (0.7)	d
Field collected	Spinosad	no	14 days residual (field)	larvae		240	53.9 (0.9)	d
Field collected	Spinosad	no	14 days residual (field)	larvae		480	62.6 (0.7)	d

* Data taken from (a) Askari Saryazdi et al., (2015), (b) Saberfar et al., (2012), (c) Hossain & Poehling, (2006b), and (d) Hossain & Poehling, (2009).

3.4 Spinetoram

Spinetoram is a member of the Spinosyns group (IRAC Group 5). Spinetoram is an insecticide developed from fermentation products of *Saccharopolyspora spinose*, and consists of two closely related active ingredients, spinosyn-J and spinosyn-L, at an approximate ratio of 3:1. A member of the spinosyns group (IRAC Group 5), along with spinosad, spinetoram was developed for the control of lepidopteran larvae, leafminers and thrips and has both contact and ingestion activity (Shimokawatoko, Sato, Yamaguchi, & Tanka, 2012). Spinetoram has both translaminar (penetration and movement within leaf tissues) and systemic activity, and therefore is available as a soil treatment, a chemigation treatment and a foliar treatment (Shimokawatoko et al., 2012). Spinetoram and spinosad are considered toxicologically equivalent. However, spinetoram has broader spectrum activity, increased insecticidal efficacy potency, and improved residual activity in the field than spinosad (Galm & Sparks, 2016). Due to this higher residual activity, there are longer withholding periods for certain crops, and important export and stock feeding regulations for treated crops in spinetoram products already registered in Australia. Spinetoram has low mobility in soil and leaching to groundwater is unlikely as it has a low solubility in water. It is degraded through a combination of biotic and abiotic mechanisms.

A spinetoram product (Radiant® SC) is registered for the control of dipterous leafminers in the USA (Table 8). Spinetoram has been shown to be efficacious at controlling both larval and adult *L. sativae*, and 100% mortality of early instar larvae was observed in a leaf dip assay with spinetoram at 23 ppm (Shimokawatoko et al., 2012). Spinetoram has also been shown to be highly efficacious at controlling the larvae of *L. trifolii*, when applied as a foliar spray (Seal et al., 2007; Hernández, Harris, & Liu, 2011 a; Hernández, Guo, Harris, & Liu, 2011b). Emulsified crop oils or methylated crop oil plus organosilicone combination products are the recommended adjuvants to aid leafminer control on the Radiant®SC product label.

Spinetoram is approved for the control of *Liriomyza* spp. in several legume crops comprising pea and bean species in Australia, as well as non-food nursery stock (Appendix 1).

Table 8. Spinetoram registration in Australia, crop groups, pests and labelled rates.

Example product	**Crop noted on label**	**Pests noted on label**	**Rate**	**WHP Harvest***	**WHP Grazing***
Delegate (250 g/kg)	Pome and Stone fruit, Grapes	*Helicoverpa* spp., Codling moth, Lightbrown apple moth, Grapevine moth, Oriental fruit moth, Looper, Pear and cherry slug, Western flower thrips	0.0075 – 0.5 g a.i./L	7 days	Do not graze
Success Neo Insecticide (120 g/L)	Avocados, Bananas, Coffee, Ornamentals, Grapes, Berry fruit, Citrus fruit, Kiwifruit, Pome fruit, Stone fruit, Subtropical and tropical fruit, Tree nuts,	*Helicoverpa* spp., Lightbrown apple moth, Grapevine moth, Citrus leafminer, Avocado leaf roller, Yellow peach moth, Sorghum head caterpillar, Carob moth, Sugarcane bud moth, Flower-eating caterpillars, small mango tipborer, large mango tipborer, Macadamia nutborer, Twig girdler, Pear and cherry slug, Western flower thrips, Red-banded thrips, Banana rust thrips,	0.06 – 0.12 g a.i./L	1 day 3 days 7 days	Do not graze
Success Neo Insecticide (120 g/L)	Culinary herbs, Brassica, Cucurbit, Fruiting, Leafy, Root and tuber, Stalk and stem, and Legume vegetables, Eucalyptus, Tea Tree, Chickpeas, Cotton, Forage Brassicas, Canola, Pulses, Soybean, Sweet Corn	*Helicoverpa* spp., Heliothis, Lightbrown apple moth, Loopers, Potato moth, Diamondback moth, Cucumber moth, Cabbage white butterfly, Cabbage cluster caterpillar, Cabbage centre grub, Cluster caterpillar, Western flower thrips, Chrysomelid beetle larvae, Pyrgo beetle	3 – 48 g a.i./ha	1 day 3 days, 14 days 28 days	3 days 14 days Do not graze

* Crops for export: Some crops for export to particular destinations outside of Australia may require a longer interval before harvest to comply with the residue standards of importing countries. This also applies to livestock for export that have been fed treated crops.

3.5 Chlorantraniliprole

Chlorantraniliprole is a member of the diamide group (IRAC Group 28). It is an anthranilic diamide insecticide that has a very specific mode of action on the muscle cells of sensitive insect species, and causes impaired regulation, paralysis and ultimately death (Cordova et al., 2006). It is a member of the diamide group (IRAC Group 28) and is active on chewing pest insects primarily by ingestion and secondarily by contact. In some situations where coverage is difficult to achieve, such as a closed canopy, dense foliage, plants with waxy leaf surfaces or less than optimum application equipment, an adjuvant may improve performance.

Chlorantraniliprole has both translaminar (penetration and movement within leaf tissues) and root systemic activity (mobile in the plant xylem and can move upward in the plant), and therefore can be applied as a foliar spray, through irrigation, or as a soil treatment. It is most effective against the immature life stages of *L. sativae* (Table 9). The compound is degraded through a combination of biotic and abiotic mechanisms, however, it is very persistent in soils. Leaching to groundwater is possible, although chlorantraniliprole has a low solubility in water.

A number of chlorantraniliprole products are registered for the control of *Liriomyza* species in the USA (Table 10).

Chlorantraniliprole is utilised for the control of several insect pest species in a range of horticultural crops (fruit and vegetables), broadacre crops (canola, pulses), coffee, grapes, and ornamentals in Australia. Only one chlorantraniliprole product, Coragen® Insecticide, is registered to control *Liriomyza* spp. in Australia in spinach and silverbeet crops. A permit, which covers several actives including a mix of chlorantraniliprole + thiamethoxam, is available for use against *Liriomyza* spp. leafminer for non-food nursery stock (Appendix 1).

Table 9. Efficacy of chlorantraniliprole against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Average score**	**LC_{50} mg a.i./L (95% CI)**	**Ref.***
field - chickpeas	Chlorantraniliprole	yes[1]	Foliar	larvae	100 mL/ha	1.17 - 1.32[3]		a
field - chickpeas	Control	no	Foliar	larvae	0	2.05 - 2.95[3]		a
field - chickpeas	Chlorantraniliprole	yes[1]	Foliar	empty mines	100 mL/ha	0.99 - 1.92[3]		a
field - chickpeas	Control	no	Foliar	empty mines	0	1.4 - 2.37[3]		a
field - chickpeas	Chlorantraniliprole	yes[1]	Foliar	adults	100 mL/ha	1.21 - 2.94[3]		a
field - chickpeas	Control	no	Foliar	adults	0	1.98 - 5.39[3]		a
field - chickpeas	Chlorantraniliprole	yes[1]	Foliar	% leaves damaged	100 mL/ha	8.7 - 11.4[3]		a
field - chickpeas	Control	no	Foliar	% leaves damaged	0	24.3 - 20.5[3]		a
lab cultured - beans	Chlorantraniliprole	yes[2]	Foliar	pupae formed	0.15 mg a.i./L	67.1% (.47)[4]		b
lab cultured - beans	Control	no	Foliar	pupae formed	0	91.6% (2.3)[4]		b

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Average score**	**LC_{50} mg a.i./L (95% CI)**	**Ref.***
lab cultured - beans	Chlorantraniliprole	yes[2]	Foliar	adults emerged	0.15 mg a.i./L	47.1%[4]		b
lab cultured - beans	Control	no	Foliar	adults emerged	0	83.2%[4]		b
lab cultured - beans	Chlorantraniliprole	yes[2]	Foliar	larvae			0.24 (0.22–0.26)	b
field - tomatoes	Control	no	Foliar	No. leaf mines		70 – 239[5]		c
field – tomatoes	Chlorantraniliprole	no	Foliar	No. leaf mines	70 g a.i./ha	22[5]		c

* Data taken from (a) López et al., (2016), (b) Khorshidi et al., (2017), and (c) DuPont, (2018).

[1] Ethylhexyl phosphate ester.

[2] Tween-80®.

[3] Average number *L. sativae* found per 40 leaves.

[4] *L. sativae* mortality % (SE).

[5] Average number of *L. sativae* found.

Table 10. Label rates for different crop groups recommended to control *L. sativae* from chlorantraniliprole products registered in the USA.

Crop	**Proposed rate for control (g a.i./ha) ****	**WHP Harvest**	**Example products ***
Cucurbit Vegetables	foliar: 44 - 110 soil: 73 – 110 drip: 73 – 100	1 day 30 days	Coragen® Insect Control (200 g/L) Durivo® (100 g/L) Besiege® Insecticide (100 g/L) Voliam Flexi® (200 g/kg) Voliam Xpress® (100 g/L)
Fruiting Vegetables	foliar: 44 - 110 soil: 73 – 110 drip: 73 – 100	1 day 5 days 30 days	Coragen® Insect Control (200 g/L) Besiege® Insecticide (100 g/L) Durivo® (100 g/L) Voliam Flexi® (200 g/kg) Voliam Xpress® (100 g/L)
Leafy Vegetables	foliar: 73 – 110 soil: 73 – 110 drip: 73 – 110	1 day 30 days	Coragen® Insect Control (200 g/L) Durivo® (100 g/L)
Legume Vegetables	foliar: 73 – 110	1 day 7 days (succulent) 21 days (dried)	Coragen® Insect Control (200 g/L) Besiege® Insecticide[1] (100 g/L)
Lettuce	foliar: 44 – 66	1 day	Besiege® Insecticide (100 g/L) Voliam Xpress® (100 g/L)
Tuberous and Corm Vegetables	foliar: 44 – 66 drip: 44 – 66 (potatoes)	14 days	Besiege® Insecticide (100 g/L)
Tropical Fruits	foliar: 74 – 111	1 day 10 days	Altacor® Insect Control (350 g/kg)

* Durivo® and Voliam Flexi® contain a mixture of chlorantraniliprole and thiamethoxam. Besiege® Insecticide and Voliam Xpress® contain a mixture of chlorantraniliprole and lambda-cyhalothrin.
** Proposed rate includes rates of chlorantraniliprole when included in a mixture product.

3.6 Cyantraniliprole

Cyantraniliprole, like chlorantraniliprole, is a member of the diamide group (IRAC Group 28). While chlorantraniliprole was originally developed for the control of lepidopteran species, cyantraniliprole has efficacy against sucking pests (whitefly, aphids) and Diptera. An anthranilic diamide insecticide, cyantraniliprole affects the muscle cells of sensitive insect species, causing impaired regulation, paralysis and death. In some situations where coverage is difficult to achieve, such as a closed canopy, dense foliage, plants with waxy leaf surfaces or less than optimum application equipment, an adjuvant may improve performance. Australian field trials with cyantraniliprole has shown efficacy with the inclusion of Hasten™ spray adjuvant. Cyantraniliprole has both translaminar (penetration and movement within leaf tissues) and root systemic activity, and therefore can be applied as a foliar spray, through irrigation, or as a soil treatment. When applied to the soil, cyantraniliprole is quickly taken up by the plant roots and translocated upwards via the xylem flow to control pests on the aerial parts of the plants. Cyantraniliprole is less persistent in the soil than chlorantraniliprole and is degraded through a combination of biotic and abiotic mechanisms. It is also more water soluble than chlorantraniliprole so is more available for local translocation upward within the xylem of the plant.

A range of cyantraniliprole products are registered for the control of *Liriomyza* species in the USA (Table 11). Cyantraniliprole has shown excellent efficacy against *L. trifolii* and *L. huidobrensis* (Trumble, 2009; Annan et al., 2011; Ferguson & DuPont, 2011).

A permit is currently available for use of Cyantranilirpole against *Liriomyza* spp. in non-food nursery stock. It is not registered for use as a soil treatment in Australia, as the oil-based formulation Benevia® is phytotoxic to plant seedlings. Cyantraniliprole is approved for the control of *Liriomyza* spp. in several fruit and vegetable crops (Appendix 1).

Table 11. Label rates for different crop groups recommended to control *L. sativae* from cyantraniliprole products registered in the USA.

Crop	**Proposed rate for control (g a.i./ha)**	**WHP Harvest**	**Example products ***
Cucurbit Vegetables	foliar: 54 - 149 soil: 99 – 197 drip: 146	1 day 7 days	DuPont™ Verimark® (200 g/L) Minecto™ Pro (135 g/L) DuPont™ Exirel® (100 g/L)
Fruiting Vegetables	foliar: 54 - 149 soil: 99 – 197 drip: 99 - 146	1 day 7 days	DuPont™ Verimark® (200 g/L) Minecto™ Pro (135 g/L) DuPont™ Exirel® (100 g/L)
Leafy Vegetables	foliar: 54 – 99 soil: 99 – 197	7 days	Minecto™ Pro (135 g/L) DuPont™ Verimark® (200 g/L)
Legume Vegetables	foliar: 99 – 149 soil: 99 – 197	1 day (succulent) 7 days (dried)	DuPont™ Verimark® (200 g/L) DuPont™ Exirel® (100 g/L)
Brassica vegetables	foliar: 99 – 149 soil: 99 – 197	1 day	DuPont™ Verimark® (200 g/L) DuPont™ Exirel® (100 g/L)
Tuberous and Corm Vegetables	foliar: 54 – 99	14 days	Minecto™ Pro (135 g/L)
Bulb vegetables	foliar: 69 – 99 soil: 99 – 197	1 day 7 days 30 days	DuPont™ Verimark® (200 g/L) Minecto™ Pro (135 g/L)
Ornamentals, greenhouses, nurseries, interior plantscapes and residential and commercial landscapes	foliar: 0.03 – 0.13 soil: 0.13 – 0.91 drip: 12.5 – 18.75	N/A	Mainspring® GNL

* Minecto™ Pro contains a mixture of cyantraniliprole and abamectin.

3.7 Emamectin benzoate

Emamectin benzoate is a member of the Avermectins (IRAC Group 6A). It is a novel semi-synthetic derivative of the natural product abamectin. It has excellent toxicity against a broad spectrum of lepidopterous pests (e.g. Lasota, Shelton, Bolognese, & Dybas, 1996) and the mode of action is similar to abamectin (GABA - and glutamate-gated chloride channel agonist). Emamectin benzoate is most effective when ingested, but also has contact efficacy. When sprayed on foliage, emamectin benzoate penetrates the leaf tissue and forms a reservoir within treated leaves, and this translaminar activity can therefore target larval *L. sativae* living inside foliage. It is relatively immobile in the environment due to a high degree of sorption to soil particles, and therefore is expected to mostly remain at the site of application until it degrades.

Emamectin benzoate is approved for the suppression of *Liriomyza* spp. in the USA (Table 12). The chemical controls all life stages of *L. sativae* (to at least 80% mortality; Table 13). A laboratory study showed that at a low concentration (0.002mg AI litre $^{-1}$), abamectin is more effective than emamectin benzoate against *L. huidobrensis*, although showed the same efficacy at a higher concentration (1mg AI litre $^{-1}$) (Ishaaya, Kontsedalov, & Horowitz, 2002). A laboratory study in Japan, showed that *L. trifolii* was less sensitive to emamectin-benzoate than its parasitoids, although *L. sativae* was as sensitive as its parasitoids (Matsuda & Saito, 2014). Another laboratory study in Japan, showed that the application of emamectin benzoate (and cartap, thiocyclam and spinosad) reduced the number of feeding and oviposition punctures made by female adults of *L. sativae* and *L. trifolli* (and *L. bryoniae*) on kidney bean leaves, and caused 78.9% and 100% egg mortality respectively (Tokumaru et al., 2005).

Emamectin benzoate is permitted for use against *Liriomyza* spp. in a range of Australian vegetable crops and non-food nursery stock (Appendix 1).

Table 12. Label rates recommended to suppress *L. sativae* from emamectin benzoate products registered in the USA.

Crop	**Proposed rate**	**WHP Harvest**	**Example products**
Brassica Vegetables, Fruiting Vegetables, Leafy Vegetables	11 – 17 g a.i./ha	7 days 14 days	Proclaim® Insecticide (50 g/kg)
Herbaceous and woody ornamental plants grown outdoors (in containers or in the ground), in commercial nursery production.	11 – 17 g a.i./ha	N/A	Enfold™

Table 13. Efficacy of emamectin benzoate against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Application**	**Life stage**	**Conc. mg a.i./L**	**Average score (SE)**	**LC_{50} ng/mg (95% CI)**	**Ref.***
field collected from pumpkin and tomato	Emamectin benzoate	Foliar	larvae	5	95.7 – 100% mortality		a
field collected from pumpkin and tomato	Emamectin benzoate	Foliar	adult	5	88% mortality		a
field collected from pumpkin and tomato	Emamectin benzoate	Foliar	eggs	5	78.9% mortality		a
field collected from pumpkin and tomato	Emamectin benzoate	Foliar	oviposition	5	1.3 (0.9) eggs laid		a
field collected from pumpkin and tomato	Control		oviposition		49.3 (5) eggs laid		a
field collected from pumpkin and tomato	Emamectin benzoate	Foliar	feeding	5	15 (4.8) punctures		a
field collected from pumpkin and tomato	Control		feeding		444.9 (49.9) punctures		a
lab culture on bean plants	Emamectin benzoate	Topical	adult females			0.166	b

***L. sativae* population**	**Chemical**	**Application**	**Life stage**	**Conc. mg a.i./L**	**Average score (SE)**	**LC_{50} ng/mg (95% CI)**	**Ref.***
						(0.054 - 0.390)	
lab culture on bean plants	Emamectin benzoate	Topical	adult males			0.071 (0.009 - 0.211)	b

* Data taken from (a) Tokumaru et al., (2005) and (b) Matsuda & Saito, (2014).

3.8 Thiamethoxam

Thiamethoxam is a systemic insecticide in the mode of action class of neonicotinoids (IRAC Group 4A). It has a broad spectrum of activity against many types of insects including aphids, whiteflies, thrips, rice hoppers, beetles, wireworms, psyllids, jassids, weevils, and leafminers. It is absorbed quickly by plants through translaminar activity, and acts to deter insect feeding in all plant parts. Thiamethoxam is absorbed through the insect stomach after feeding, or through direct contact. The insecticide affects the nicotinic acetylcholine receptors in the central nervous system, and eventually paralyses the muscles of the insects. It can be applied as a foliar spray, through drip irrigation or soil drenching as a soil treatment, or as a seed treatment, due to its systemic activity. The compound is degraded through hydrolysis, moving rapidly to sediment phase where degradation continues. While thiamethoxam has high potential soil mobility, it is not likely to leach readily in soil.

In the USA, Thiamethoxam can be applied as a foliar spray, through irrigation, or as a soil treatment for leafminer species control (Table 14) and is most effective against the immature life stages of *L. sativae* (Table 15).

Thiamethoxam is approved for the control of a broad range of insect pest species of various field crops in Australia, However, is only permitted for use against *Liriomyza* spp. leafminer as part of a mix of chlorantraniliprole + thiamethoxam for non-food nursery stock (Appendix 1).

Table 14. Label rates for different crop groups recommended to control *L. sativae* from thiamethoxam products registered in the USA.

Crop	**Proposed rate for control**	**WHP Harvest**	**Example products ***
Cucurbit Vegetables	foliar: 53 – 96 g a.i./ha soil/drip: 88 – 193 g a.i./ha seed: 0.25 – 0.75 mg a.i./seed	1 day 30 days	Actara® (250 g/L) Cruiser® FS (600 g/L) Platinum® (240 g/L) Flagship® (250 g/L)
Fruiting Vegetables	foliar: 56 – 98 g a.i./ha soil/drip: 88 – 193 g a.i./ha	1 day 5 days 30 days	Platinum® (240 g/L) Durivo® (100 g/L) Voliam Flexi® (200 g/kg)
Leafy Vegetables	soil/drip: 88 – 193 g a.i./ha seed: 0.5 – 1.2 mg a.i./seed	30 days	Platinum® (240 g/L) Durivo® (100 g/L) Cruiser® 70WS (700 g/L)
Legume Vegetables	seed: 25 – 50 g a.i./ 100kg seed	30 days	Seed Shield® Beans (255 g/L) CruiserMaxx ® (260 g/L)

Crop	**Proposed rate for control**	**WHP Harvest**	**Example products ***
Ornamental plants	foliar: 140 - 298 g a.i./ha soil/drip: 140 – 305 g a.i./ha		Meridian® (250 g/kg) Flagship® (250 g/kg)
Potatoes	seed: 0.042 – 0.06 g a.i./seed		Cruiser® FS (600 g/L)
Tuberous and Corm Vegetables	foliar: 44 – 53 g a.i./ha	7 days 14 days	Endigo® (141 g/L) Agri-Flex® (152 g/L)
Soybeans	seed: 50 g a.i./ 100kg seed	30 days	Seed Shield® Soybean (255 g/L) CruiserMaxx® (260 g/L)

Proposed rate includes rates of thiamethoxam when included in a mixture product.
* Durivo® and Voliam Flexi® contain a mixture of thiamethoxam and chlorantraniliprole. Endigo® contains a mixture of thiamethoxam and lambda-cyhalothrin. Agri-Flex® contains a mixture of thiamethoxam and abamectin.

Table 15. Efficacy of thiamethoxam against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc. mg a.i./L**	**Average score**	**LC_{50} mg a.i./L (95% CI)**	**Ref.***
Susceptible	Thiamethoxam	Tween-80®	Foliar	larvae			8.47 (7.29 - 9.80)	a
Chlorpyrifos-resistant	Thiamethoxam	Tween-80®	Foliar	larvae			9.93 (8.66 - 11.48)	a
Field collected	Thiamethoxam	no	Foliar	larvae	50	48.8 - 67.0% mortality		b
Field collected	Thiamethoxam	no	Foliar	pupae	50	52.0 - 77.1% mortality		b
Field collected	Thiamethoxam	no	Foliar	eggs	50	60.9% mortality		b
Field collected	Thiamethoxam	no	Foliar	adults	50	22.5% mortality		b
Field collected	Thiamethoxam	no	Foliar	eggs laid per 2 female adults	50	85.5 (10.3)		b
Field collected	Control	no	Foliar	eggs laid per 2 female adults		49.3 (5.0)		b

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc. mg a.i./L**	**Average score**	**LC_{50} mg a.i./L (95% CI)**	**Ref.***
Field collected	Thiamethoxam	no	Foliar	feeding punctures per 2 female adults	50	592.8 (100.7)		b
Field collected	Control	no	Foliar	feeding punctures per 2 female adults		444.9 (49.9)		b

* Data taken from (a) Askari Saryazdi et al., (2015) and (b) Tokumaru et al., (2005).

3.9 Indoxacarb

Indoxacarb is an oxadiazine insecticide with a main mode of action of blocking nerve sodium channels (IRAC Group 22A). It is highly effective at controlling lepidopteran pests, and also has some suppression activity against coleopteran, hemipteran and dipteran insects. Indoxacarb is both ingested and absorbed through the cuticle but is most lethal by ingestion. Inhibition of insect feeding occurs rapidly with pest knockdown typically in 1 to 3 days. It is absorbed into the waxy cuticle of leaves, with poor translaminar activity, which is increased with the use of a penetrating oil-based surfactant. Indoxacarb has no translocation in the plant and efficacy is impacted by growth dilution. It degrades rapidly in soils and has a low potential of leaching into groundwater.

In the USA, indoxacarb is registered as a foliar spray for the suppression of *Liriomyza* species in tomatoes (Table 16). Indoxacarb alone provided poor control of *L. trifolii*, however, in combination with a surfactant, Agri-Dex®, indoxacarb provided significant control of leafminer larvae and pupae (Seal, Betancourt, & Sabines, 2002). Similarly, Indoxacarb, in combination with certain other surfactants, was equally effective to the first, second and third instars of leafminer on 'Pod Squad' beans. While Indoxacarb alone did not affect emergence of the leafminer parasite, *Diglyphus* sp., when in combination with various surfactants, it significantly reduced leafminer parasitoids (Seal et al., 2002).

Indoxacarb is very effective against lepidopteran larvae and is registered in Australian field crops for the control of several different insect families, however it is only permitted for use against *Liriomyza* spp. leafminer within non-food nursery stock (Appendix 1).

3.10 Spirotetramat

Spirotetramat is an IRAC Group 23. It is derived from tetramic acid, a systemic material for the control of sucking insects in their immature stages, including aphids, scale insects, and whitefly. It produces growth inhibition of immature insects, and reduces the ability of insects to reproduce, resulting in mortality. It acts to inhibit the biosynthesis of lipids. Spirotetramat is toxic to aquatic organisms. It may have an adverse effect on predatory mites where IPM is practiced.

In a study on greenhouse grown gerberas in the USA, spirotetramat (Kontos) did not provide an acceptable level of leaf miner control for ornamental growers (Ludwig, 2011). In another trial in the USA on chrysanthemum (*Chrysanthemum indicum*), while no differences were found in the number of *L. trifolii* pupae collected, spirotetramat (Kontos) reduced the number of adults by almost half, suggesting that this compound may be used to control leafminers commercially (Vea & Palmer, 2018). It is however, not registered in the USA for control of *Liriomyza* spp.

Spirotetramat is registered in Australia for the control of a range of pests in vegetables and tree crops including *Myzus persicae* (green peach aphid), *Bemisia tabaci*/Biotype B (silverleaf whitefly) and *Aphis gossypii* (cotton aphid). It is also registered for use against *Liriomyza* spp. in a range of vegetables in Australia (Appendix 1).

3.11 Acetamiprid

Acetamiprid is a neonicotinoid from IRAC Group 4A. The neonicotinoids act on postsynaptic nicotinic acetylcholine receptors (nAChRs). In mammals, acetamiprid causes generalised, nonspecific toxicity and does not appear to have specific target organ toxicity (Environmental Protection Agency, 2002). Acetamiprid use poses minimal risk to fish and wildlife. It degrades rapidly by aerobic soil metabolism and poses low risks to the environment relative to most other insecticides (Environmental Protection Agency, 2002).

The stippling damage caused by *L. huidobrensis* on mustard greens is more economically damaging than mining damage, however trials in Canada showed that none of three insecticides tested - acetamiprid, novaluron and cyromazine - reduced stippling damage (Hallett & Heel, 2004a). Similarly, in spinach stippling damage is more economically damaging than mining damage. Testing the same products as for mustard greens, showed that all insecticides effectively reduced stippling damage levels, however the high rate of cyromazine was the best treatment as mining and stippling damage to leaves and damaged leaves per bunch were all than in the control (Hallett & Heel, 2004b).

In Romaine lettuce, while all treatments (high rate acetamiprid, low and high rate novaluron and registered rate of cyromazine), except the low rate of acetamiprid, significantly reduced mining damage, no treatment had lower stippling damage than the control (Hallett & Heel, 2004c). Harvest bunch damage was significantly lower in plots treated with the high rates of acetamiprid and novaluron, and the registered rate of cyromazine than in the control, and were considered the most effective treatments (Hallett & Heel, 2004c).

Acetamiprid has been shown to significantly reduced mining damage caused by *L. huidobrensis* in Romaine lettuce at low rates (though not at high rates), while having no effect on stippling damage (Hallett & Heel, 2004c). Similarly, acetamiprid did not reduce stippling damage in mustard greens (Hallett & Heel, 2004a), but did in spinach (Hallett & Heel, 2004b).

Acetamiprid is not registered in the USA against *Liriomyza* spp., and is currently not registered in Australia for *Liriomyza* spp.

3.12 Novaluron

Novaluron is a benzoylphenyl urea (IRAC Group 15) that inhibits chitin development (Ishaaya & Casida, 1974). It causes unsuccessful endocuticular deposition and disrupts moulting by the arthropod (Mulder & Gijswijk, 1973). Novaluron acts primarily via ingestion, but also has some contact and translaminar activities (Ishaaya., Kontsedalov, & Horowitz, 2003).

Novaluron has proved effective against *L. trifolii* in the USA when compared with abamectin, spinetoram, and lambda-cyhalothrin (e.g. Hernández, Harris, et al., 2011). Conversely, novaluron was ineffective against *L. trifolii* on tomato in Florida, USA (Seal et al., 2007). In Canada, three products, ASSAIL 70 WP (acetamiprid 70.35%), RIMON 10 EC (novaluron 100 g/L), and CITATION 75 WP (cyromazine 75%) were tested for their efficacy against *L. huidobrensis* on celery (Hallett & Heel, 2004d). Novaluron at the highest rate, based on lower mining damage indices per bunch and lower stippling damage indices, most effectively reduced *L. huidobrensis* damage to celery (Hallett & Heel, 2004d).

Novaluron is currently not registered in Australia for *Liriomyza* spp.

3.13 Azadirachtin (Neem oil)

Azadirachtin is a secondary metabolite present in neem seeds and is a naturally occurring substance that belongs to the tetranortriterpenoid organic molecule class. It is commonly used in garden use and organic growing. It is considered to be non-toxic to mammals and is not expected to have any adverse effects on non-target organisms or the environment. In insect pests, azadirachtin acts mainly as an antifeedant and growth disruptor, although depending on the life stage, insect death may not occur for several days. Azadirachtin degrades when exposed to light and water and has low mobility in the soil. No phytotoxicity has been observed when used as directed.

Azadirachtin is registered for the control of *Liriomyza* species in almost all field, food and ornamental crops in the USA as a foliar spray and soil drench (Table 17). As azadirachtin is an insect growth regulator, it is most effective on the larval stages of *L. sativae* and does not control adult insects (Table 18). Results are typically observed 3-7 days after first treatment. Azadirachtin, although not as effective as abamectin and spinosyn in controlling *L. trifolii* on 'Pod Squad' beans, when used together with several ionic and non-ionic surfactants, significantly reduced leafminer larvae (Seal et al., 2002). When used in rotation with management tools for *L. trifolii* in the USA, azadirachtin (and thiamethoxam) is thought to delay the development of tolerance or resistance to spinosan and abamectin (Seal et al., 2002).

Azadirachtin is a registered organic plant protection product for the control of various pests in ornamental and home garden crops in Australia (APVMA https://portal.apvma.gov.au/home; accessed March 2020). Additionally, a permit, which covers several actives including Azadirachtin, is available for use against *Liriomyza* spp. leafminer in non-food nursery stock (Appendix 1).

Table 17. Label rates for different crop groups recommended to control *L. sativae* from azadirachtin products registered in the USA.

Crop	**Proposed rate**	**WHP Harvest**	**Example products**
All outdoor plants including food and fiber crops, and trees	13 – 26 g a.i./ha	0 days (can be used the same day as harvest)	Neemix® 4.5 Insect Growth Regulator (46.73 g/L) Aza-Direct® (11.82 g/L) Molt-X® (33.55 g/L) Ecozin® Plus 1.2% ME (11.98 g/L)
Vegetables, Melons, Strawberries, and other food crops grown in greenhouses (or other cover) for transplanting to production fields. Bearing and non-bearing fruit and nut trees, grape vines, caneberries, and other small fruits	20 – 55 g a.i./ha	0 days (can be used the same day as harvest)	Neemix® 4.5 Insect Growth Regulator (46.73 g/L) Molt-X® (33.55 g/L) AzaGuard™ Botanical Insecticide/Nematicide (33.55 g/L) Aza-Direct® (11.82 g/L)
Ornamental Plants and Flowers, Ornamental Trees and Shrubs	30 - 201 mg a.i./L	0 days (can be used the same day as harvest)	Azatrol® EC Insecticide (11.82 g/L)

Table 18. Efficacy of azadirachtin against different life stages of *L. sativae.*

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Mortality % (SE)**	**Ref.***
Susceptible	Azadirachtin	yes	Foliar	larvae		2.7 (2.18 - 3.44)**	a
Chlorpyrifos-resistant	Azadirachtin	yes	Foliar	larvae		3.73 (3.20 - 4.35)**	a
Field collected	Control	no	Foliar	larvae		22.6 (1.26)	b
Field collected	Azadirachtin	no	Foliar	larvae	0.005 g a.i./L	93.7 (0.92)	b
Field collected	Azadirachtin	no	Foliar	larvae	0.01 g a.i./L	100	b
Field collected	Control	no	Soil drench	pupae	0	23.2 (1.11)	b
Field collected	Azadirachtin	no	Soil drench	pupae	0.1275 g a.i./L	63.8 (1.35)	b
Field collected	Azadirachtin	no	Soil drench	pupae	0.255 g a.i/L	86 (0.95)	b
Field collected	Azadirachtin	no	Soil drench	pupae	0.3825 g a.i./L	90.0 (0.78)	b
Field collected	Azadirachtin	no	Soil drench	pupae	0.51 g a.i./L	94.7 (1.05)	b
Field collected	Control	no	Foliar	eggs		0.86 (0.65)	c
Field collected	Azadirachtin	no	Foliar	eggs	0.005 g a.i./L	1.15 (0.82)	c
Field collected	Azadirachtin	no	Foliar	eggs	0.01 g a.i./L	1.55 (1.03)	c
Field collected	Control	no	Foliar	larvae		1.9 (0.89)	c
Field collected	Azadirachtin	no	Foliar	larvae	0.005 g a.i./L	76.65 (0.99)	c
Field collected	Azadirachtin	no	Foliar	larvae	0.01 g a.i./L	90.14 (0.87)	c

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Mortality % (SE)**	**Ref.***
unknown	Control	no	Foliar	larvae		11.3	d
unknown	Azadirachtin	no	Foliar	larvae	1 g /100mL	19.9	d
unknown	Azadirachtin	no	Foliar	larvae	5 g /100mL	50.1	d
unknown	Azadirachtin	no	Foliar	larvae	10 g /100mL	66.8	d
unknown	Azadirachtin	no	Foliar	larvae	15 g /100mL	91	d
unknown	Azadirachtin	no	Foliar	larvae	20 g /100mL	91.8	d
unknown	Control	no	Foliar	pupae		13.6	d
unknown	Azadirachtin	no	Foliar	pupae	1 g /100mL	87.4	d
unknown	Azadirachtin	no	Foliar	pupae	5 g /100mL	99.4	d
unknown	Azadirachtin	no	Foliar	pupae	10 g /100mL	100	d
unknown	Azadirachtin	no	Foliar	pupae	15 g /100mL	100	d
unknown	Azadirachtin	no	Foliar	pupae	20 g /100mL	100	d
Field collected	Control	no	soil drench laboratory	larvae		0.7 - 1.2	e
Field collected	Azadirachtin	no	soil drench laboratory	larvae	0.75 g/L	36.0 - 74.2	e
Field collected	Azadirachtin	no	soil drench laboratory	larvae	1.5 g/L	63.2 - 81.6	e
Field collected	Azadirachtin	no	soil drench laboratory	larvae	2.25 g/L	73.7 - 100	e
Field collected	Azadirachtin	no	soil drench laboratory	larvae	3.0 g/L	80.4 - 100	e
Field collected	Control	no	soil drench field	larvae		1.2 - 2.5	e

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Mortality % (SE)**	**Ref.***
Field collected	Azadirachtin	no	soil drench field	larvae	0.75 g/L	9.4 - 72.4	e
Field collected	Azadirachtin	no	soil drench field	larvae	1.5 g/L	40.6 - 82.8	e
Field collected	Azadirachtin	no	soil drench field	larvae	2.25 g/L	65.3 - 99.3	e
Field collected	Azadirachtin	no	soil drench field	larvae	3.0 g/L	69.5 - 99.2	e
Field collected	Control	no	soil drench	eggs		1.45 (0.59)	e
Field collected	Azadirachtin	no	soil drench	eggs	0.75 g/L	1.05 (0.56)	e
Field collected	Azadirachtin	no	soil drench	eggs	1.5 g/L	1.40 (0.73)	e
Field collected	Azadirachtin	no	soil drench	eggs	2.25 g/L	1.58 (0.81)	e
Field collected	Azadirachtin	no	soil drench	eggs	3.0 g/L	1.87 (0.86)	e
Field collected	Control	no	soil drench	1st instar larvae		1.06 (0.71)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	0.75 g/L	24.2 (1.4)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	1.5 g/L	73.7 (1.1)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	2.25 g/L	93.3 (1.6)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	3.0 g/L	98.7 (0.9)	e
Field collected	Control	no	soil drench	2nd instar larvae		1.5 (0.8)	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	0.75 g/L	7.6 (1.4)	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	1.5 g/L	46.6 (2.2)	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	2.25 g/L	57.9 (2.5)	e

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Mortality % (SE)**	**Ref.***
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	3.0 g/L	60.4 (1.7)	e
Field collected	Control	no	soil drench	3rd instar larvae		2.2 (1.1)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	0.75 g/L	6.4 (1.6)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	1.5 g/L	25.9 (2.1)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	2.25 g/L	55.6 (1.8)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	3.0 g/L	58.7 (1.4)	e
Field collected	Control	no	soil drench	eggs		22.2 (1.9)	e
Field collected	Azadirachtin	no	soil drench	eggs	0.75 g/L	100	e
Field collected	Azadirachtin	no	soil drench	eggs	1.5 g/L	100	e
Field collected	Azadirachtin	no	soil drench	eggs	2.25 g/L	100	e
Field collected	Azadirachtin	no	soil drench	eggs	3.0 g/L	100	e
Field collected	Control	no	soil drench	1st instar larvae		23.6 (3.6)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	0.75 g/L	99.4 (0.6)	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	1.5 g/L	100	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	2.25 g/L	100	e
Field collected	Azadirachtin	no	soil drench	1st instar larvae	3.0 g/L	100	e
Field collected	Control	no	soil drench	2nd instar larvae		21.8 (0.5)	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	0.75 g/L	95.5 (2.3)	e

***L. sativae* population**	**Chemical**	**Adjuvant**	**Application**	**Life stage**	**Conc.**	**Mortality % (SE)**	**Ref.***
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	1.5 g/L	100	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	2.25 g/L	100	e
Field collected	Azadirachtin	no	soil drench	2nd instar larvae	3.0 g/L	100	e
Field collected	Control	no	soil drench	3rd instar larvae		22.3 (1.9)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	0.75 g/L	86.4 (3.0)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	1.5 g/L	99.0 (1.0)	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	2.25 g/L	100	e
Field collected	Azadirachtin	no	soil drench	3rd instar larvae	3.0 g/L	100	e
Lab cultured	Azadirachtin	Tween-80®	foliar	larvae	5 mg a.i./L	33.6	f
Lab cultured	Control	Tween-80®	foliar	larvae		7.7	f
Lab cultured	Azadirachtin	Tween-80®	foliar	pupae	5 mg a.i./L	94.2	f
Lab cultured	Control	Tween-80®	foliar	pupae		16.8	f

* Data taken from (a) Askari Saryazdi et al., (2015), (b) Hossain & Poehling, (2006a), (c) Hossain & Poehling (2009), (d) Costa et al., (2016), (e) Hossain, Poehling, Thöming, & Borgemeister, (2008), and (f) Khorshidi et al., (2017). ** Indicates LC_{50} mg a.i./L (95% CI)

3.14 Synthetic pyrethroids and organophosphates

Synthetic pyrethroids (IRAC Group 3A) and organophosphates (IRAC Group 1B) are effective at controlling susceptible populations of *L. sativae*. Although the short persistence time and lack of plant tissue penetration of some pyrethroids (e.g. permethrin) mean these products are ineffective at controlling larvae; foliar sprays are lethal to adult stages of susceptible populations (Bartlett & Powell, 1981). There is variation in the efficacy of different organophosphate compounds for *L. sativae* control, in particular, fenitrothion is not very effective compared with chlorpyrifos (Tokumaru et al., 2005). Currently, there are 13 synthetic pyrethroids and nil organophosphate products registered for the control of *L. sativae*, *L. huidobrensis* and *L. trifolii* in the USA (www.greenbook.net; accessed 20 February 2020). The synthetic pyrethroids include 11 permethrin products, one zeta-cypermethrin/avermectin B1 product and one bifenthrin/avermectin B1 product.

In Australia, these two chemical groups are widely registered across our agricultural cropping systems, in multiple plant groups for many invertebrate pests. However, both chemistries have a broad spectrum of action and are often lethal to beneficial insects and disrupt IPM systems (see section on toxicity to beneficials).

Pursuing new registrations of organophosphate and pyrethroid insecticides is also problematic given the regulatory pressures that currently exist on these older, broad-spectrum pesticides. Concerns about toxicity and associated potential risks to the public, occupational health and safety and the environment has prompted the APVMA to review the usage of over 20 chemicals from these chemical groups. Registrations have been cancelled for several organophosphates (e.g. methamidophos, fenthion) and label amendments and restricted usages have been applied to other chemicals (e.g. bifenthrin, dimethoate, diazinon). Registration of chemicals from these classes against *L. sativae* would risk future review, and even potential withdrawal of permits or labels.

Along with these continuing regulatory pressures, many invertebrate pests have evolved resistance to these two chemical groups, and *Liriomyza* leafminers are no exception. There are well documented issues of resistance in several *Liriomyza* species, and field and laboratory studies have shown evidence for the evolution of resistance to both synthetic pyrethroids (cypermethrin, permethrin, and fenpropathrin) and organophosphates (chlorpyrifos) in *L. sativae* populations (Mason & Johnson, 1987; Johansen, Tuan, Oanh, & Nordhus, 2003; Askari Saryazdi, Hejazi, Rashidi, & Ferguson, 2014; Askari Saryazdi et al., 2015).

While these two groups of insecticides may have some use in initial eradication and area freedom programs against *L. sativae* in Australia, it is not recommended to pursue registration of these products for ongoing management.

4. Toxicity to beneficials

Parasitoid wasps provide important natural control of *Liriomyza* spp., and in the absence of insecticides can keep populations at low levels (Harding, 1965; Trumble, 1981; Liu, Kang, Lei, & Hernández, 2011). The integrated use of beneficial organisms and other control measures, such as cultural controls, will be essential for ongoing control of *Liriomyza spp.* populations in commercial crops. In food crops, where foliage disfigurement does not reduce the economic yield of the crop, it may be possible for *Liriomyza* populations to be effectively suppressed below an economic threshold by parasitoid wasps (Gao et al., 2014).

There are many well documented cases where repeated applications of insecticides against *Liriomyza* leafminers have reduced parasitoid populations and resulted in subsequent pest outbreaks (Oatman & Kennedy, 1976; Saito, Oishi, Ikeda, & Sawaki, 1992; Hidrayani et al. 2005; Liu et al., 2011). Indeed, most broad-spectrum synthetic insecticides developed since the 1940s are highly toxic to parasitoids of *Liriomyza* spp. (Hidrayani et al., 2005; Oatman & Kennedy, 1976; Saito, Ikeda, & Ozawa, 1996; Schuster, 1994). Those with high toxicity to parasitoids include carbamates, organochlorines, organophosphates and pyrethroids, which are also insecticides that show limited efficacy against *Liriomyza* spp. (Hara, 1986; Hidrayani et al., 2005).

Consequently, it is important to examine how insecticides selected for *Liriomyza* spp*.* control may disrupt natural parasitoid populations. Parasitoids that will potentially be the most important in aiding *Liriomyza* spp*.* control in Australia include *Hemiptarsenus varicornis*, *Neochrysocharis* spp., *Zagrammosoma latilineatum*, *Diglyphus isaea*, *Closterocerus mirabilis*, *Aprostocetus* spp., *Opius* spp., *Trigonogastrella parasitica* and *Callitula* spp. (Ridland, Umina, Pirtle, & Hoffmann, 2020). Toxic effects of suggested chemicals for *L. sativae* control against these species are summarised in Table 30. Unfortunately, data are only available for four of these target parasitoids: *H. varicornis*, *D. isaea*, *N. formosa* and *Opius* spp. In particular, cyromazine and cyantraniliprole are known to be selective to hymenopteran parasitoids and are therefore important options to consider for control of *Liriomyza* spp*.* in Australia. Cyromazine is a growth regulator specific to Diptera, and therefore does not directly affect the development of parasitic Hymenoptera. Cyromazine will lead to the mortality of the *Liriomyza* spp. larvae before parasitoids complete their development, and will reduce host numbers. However, these effects are thought to be complementary and enhance the action of parasitoids (Reitz et al., 2013). There has not been any research on the effect of novaluron on natural enemies of *Liriomyza* leafminers. However, the effect of novaluron was tested on a parasitoid, *Trichogramma pretiosum* Riley of the Mediterranean flour moth, *Ephestia kuehniella* (Zeller), which showed that novaluron did not affect the adult parasitoids, but interfered with immature development within the host egg (Bastos, Almeida, & de Suinaga, 2006). Spinetoram also negatively affects parasitoid immature stages, and previous research has shown that this insecticide is harmful to natural enemies (Hossain & Poehling, 2006b). However, a subsequent trial in Texas, USA showed that spinetoram was no more toxic to the parasitoid complex

compared to the untreated control (Hernández, Harris, et al., 2011). The authors suggested that additional studies are required to elucidate the toxicity of spinetoram to parasitoids of *Liriomyza* spp.

As well as the selectivity of the chemical, application method can have an effect on how disruptive insecticides are to parasitoid wasps. Generally, application of chemicals via drip irrigation systems (e.g. PMR diamides) and soil drenches are less disruptive than foliar sprays (Weintraub, 1999; Tran, Takagi, & Takasu, 2005; Liu et al., 2011).

Excessive chemical use for *Liriomyza* control will also affect beneficial insects beyond specialist leafminer parasitoid species. For example, spinetoram has been shown to be highly toxic to predatory mites and honey bees (Biondi et al., 2012). However, products such as chlorantraniliprole have been shown to have negligible effects on numerous beneficial organisms (e.g. the predatory mite *Typhlodromus pyri* and the parasitic wasp *Aphidius rhopalosiphi*) (Dinter, Brugger, Bassi, Frost, & Woodward, 2008). In closed horticultural systems such as greenhouses, where supplementary populations of parasitoid wasps (e.g. *Eretmocerus* and *Aphidius*), predatory mites and lacewings are released, it will be important to choose selective chemicals to register for *Liriomyza* control. Products containing azadirachthin, chlorantraniliprole, novaluron and cyromazine are the least disruptive to beneficial species overall, followed by indoxacarb, cyantraniliprole, thiamethoxam, abamectin, spinetoram and emamectin benzoate. Products containing synthetic pyrethroids and organophosphates will be the most disruptive to current IPM systems. Differences in application of some chemicals will affect how disruptive/selective they are (Table 30).

Table 19. Toxicity effect ratings on various lifecycle stages of four important parasitoid wasps of *Liriomyza spp. (Hemiptarsenus varicornis, Diglyphus isaea, Neochrysocharis formosa,* and *Opius* spp.).

		Hemiptarsenus varicornis		***Diglyphus isaea***			***Neochrysocharis formosa***		***Opius* spp.**	
Chemical	**MoA Group**	**L**	**A**	**L**	**P**	**A**	**L**	**A**	**L**	**A**
Organophosphates	1B		VH [1,2]	H-VH [1,2,3,4,5]		VH [1,4,5]		VH [1]		VH [5,6]
Synthetic pyrethroids	3A		VH [1]	H-VH [1,3,4,5,7,8]		VH [3,4,5,7,8]		H-VH [1,9]		VH [5,6]
Acetamiprid	4A			H [4,5,10]		L-H [4,5,9,10,11]				L [12]
Thiamethoxam	4A			H [5, 13]		H [5,8,14]			L [12]	
Spinetoram	5			VH [5]		VH [5,11]		H-VH [9]		
Spinosad	5			H [4,15]		H-VH [4,8,11,16]	M [17]		VH [17]	VH [5,18]
Abamectin	6	M [19]	L [2]	M [4,5,19]	VH [19]	H-VH [2,4,5,7,8]	M-H [17]	VH [9]		VH [5,12,17,20]
Emamectin benzoate	6		VH [1]	M [5]		VH [1,3,5]		VH [1]		
Novaluron	15			M [5]		L [5,8]		L [9]		
Cyromazine	17	L [19]		L-M [2,4,5,19,21]	L [19]	L-M [4,5,19,21]				L [2,5,22]
Indoxacarb	22A			M [4,5]		L [4,5]				L [12]
Spirotetramat	23			M [5]	L [4]	L-M [5,8]				
Chlorantraniliprole	28			L [5]		L [5,11]				
Cyantraniliprole	28			L [5]		H [11]				
Azadirachtin	-			L [4,5]		L-M [4,5,8]	L [17]		H [17]	M [18]

where L = larvae, P = pupae, and A = adult. To account for variability in mortality between laboratory and field trials, pesticide toxicity ratings are categories into L, M, H and VH according to IOBC protocols, where observed mortality in laboratory and extended laboratory trials correspond to <30%, 30-79%, 80-98%, and >99% mortality, respectively, and semi-field and field trials correspond to <25%, 25-50%, 50-75%, and >75% mortality, respectively. Unless specified, the toxicity rating within the table has been derived from spray application methods. Footnotes within the table detail where toxicity ratings were derived from, as well as the contact type (if applicable; direct, residual or indirect contact) and application method.

[1] Matsuda & Saito, 2014; direct contact via micropipette application

[2] Prijono, Robinson, Rauf, Bjorksten & Hoffmann, 2004; residual contact via coated glass vial method application

[3] Ozawa, Saito & Ikeda, 1998

[4] Biobest Database; spray application

[5] Koppert Database; spray application

[6] Sterk et al., 1999; residual contact via dried spray application

[7] Guantai et al., 2015; residual contact via coated glass vial method application

[8] Abraham, Braman, Oetting & Hinkle, 2013; residual contact via dried spray application

[9] Hernández, Guo, et al., 2011; direct and residual contact via spray applications

[10] Biobest Database reports a L toxicity rating when acetamiprid was applied through irrigation, and similarly, a L toxicity rating is reported by Koppert Database when applied through a drench application.

[11] Kim, Ahn, Ha, Lim & Lee et al. 2018; direct contact via spray application

[12] Araujo, Nogueira, & Bezerra, 2015; residual contact via spray application

[13] Koppert Database reports an H toxicity rating when thiamethoxam was applied through a drench application.

[14] Koppert Database reports a M toxicity rating when thiamethoxam was applied through a drench application

[15] Biobest Database reports a H toxicity rating when spinosad was applied through irrigation.

[16] Biobest Database reports a L toxicity rating when spinosad was applied through irrigation.

[17] Hossain & Poehling, 2006b; indirect contact via dried spray application

[18] Viñuela et al., 2001; residual contact via dried spray application

[19] Bjorksten & Robinson, 2005; larvae: indirect contact via leaf dip method, pupae: direct contact via dipping method

[20] In a field trial run by Hidrayani et al. (2005), no significant difference in parasitism between control and abamectin treated plots was reported

[21] Biobest Database and Koppert Database report L toxicity ratings when cyromazine was applied through irrigation and drench application, respectively.

[22] Koppert Database reports a L toxicity rating when cyromazine was applied through drench application.

5. Resistance status, mechanisms, cross resistance and implications for Australian control

In many countries, reliance upon insecticides as the main control method for leafminers has led to insecticide resistance evolving in several *Liriomyza* species. It is apparent that there is a difference in insecticide susceptibility among *Liriomyza* spp, therefore accurate identification will be important (Reitz et al., 2013). Similarly, understanding the resistance status of a *Liriomyza* spp. populations entering Australia will be important in order to manage this pest. Comparative toxicity data showed that *L. trifolii* was more tolerant to synthetic organic insecticides than *L. sativae* Blanchard (Parrella & Keil, 1985). A study in the USA showed that *L. trifolii* populations are much less susceptible to methamidophos than either *L. sativae*, or *L. langei* in order of decreasing tolerance (Parrella & Keil, 1985). Similarly, *L. trifolii* populations in China are considerably less susceptible to abamectin and cyromazine than are populations of *L. sativae* (Gao, Reitz, Wei, Yu, & Lei, 2012). Conversely, in Japan*, L. sativae* populations were less susceptible to several commonly used insecticides than were local populations of *L. trifolii* (Tokumaru et al., 2005). Israeli populations of the invasive *L. huidobrensis* appear more tolerant to some commonly used insecticides than sympatric populations of *L. trifolii* (Weintraub, 2001).

Resistance in *L. sativae* has been documented for three different IRAC MoA groups: 1B (organophosphates), 3A (synthetic pyrethroids) and 6 (avermectins). However, only resistance to pyrethroids has been observed in the field: 5-28-fold fenpropathrin resistance in *L. sativae* populations in Iran (Askari Saryazdi et al., 2014), 12.5-fold permethrin resistance in Hawaii (Mason & Johnson, 1987), and 14-19-fold cypermethrin resistance in Vietnam (Johansen et al., 2003). In the other cases, resistance through artificial selection has been observed in laboratories, including chlorpyrifos resistance in Iran (Askari Saryazdi et al., 2015), and abamectin resistance in China (Wei, Lei, Nauen, Cai, & Gao, 2015).

L. trifolii has evolved resistance to most chemicals registered for leafminer control, including chemicals from IRAC MoA groups 1A (Carbamates), 1B (chlorpyrifos, demeton, methamidophos, parathion-methyl, pyrazophos), 2A (toxaphene), 3A (bifenthrin, cypermethrin, permethrin), 3B (DDT), 5 (spinosad), 6 (abamectin) and 17 (cyromazine) (Ferguson, 2004; Gao et al., 2017; APRD, 2018). Recently there have also been field reports of resistance to IRAC MoA Group 28 (cyantraniliprole and chlorantraniliprole) in populations of *L. trifolii* in Florida tomato crops (S. Ferguson, personal communication, 2017). This apparent faster development of resistance by *L. trifolii* compared to other species, is thought to be due largely to its comparatively higher reproductive capacity and the exposure of confined populations to heavy insecticide pressure in enclosed greenhouses (Parrella, Allen, & Morishita, 1981; Parrella, Keil, & Morse, 1984).

A timeline of the development of resistance for *L. sativae* and *L. trifolii* in Florida, as well as the actives used is shown in Table 20. Control failures in the mid-1970s coincided with the displacement of

L. sativae by *L. trifolii* (Leibee, 1981; Parrella & Keil, 1984; Leibee & Capinera, 1995; Ferguson, 2004; S. Ferguson personal communication, 2017).

Table 20. History of insecticide use on *Liriomyza* spp. in vegetables in Florida, USA.

Insecticide	**Date first used**	**Effective field life (yr)**	**Year of control failure**
Nicotine sulfate	<1945	?	
Chlordane	1947	11	1958
Toxaphene	1947	5	1952
Parathion	1948	10	1958
Diazinon	1958	3	1961
Azinphosmethyl	1961	13	1974
Dimethoate	1961	13	1974
Naled	1961	13	1974
Oxamyl	1975	2	1977
Methamidophos	1977	4	1981
Permethrin	1978	2	1980
Cyromazine	1983	ongoing	n.a.[1]
Abamectin	1990	ongoing	n.a.[1]
Spinosad	1997	ongoing	n.a.[1]
Spinetoram	2009	ongoing	n.a.
Chlorantraniliprole	2008	ongoing	n.a.[2]
Cyantraniliprole	2013	ongoing	n.a.[2]
Cyclaniliprole	2018	ongoing	n.a.
Thiomethoxam	1999	ongoing	n.a.

The table was modified from Parrella & Keil (1984).
[1] some resistant *Liriomyza trifolii* populations have been identified (Ferguson, 2004).
[2] some resistant *Liriomyza trifolii* populations have been identified (S. Ferguson personal communication, 2017).

L. huidobrensis populations rapidly develop resistance to the particular conventional insecticides used in different countries, and therefore resistance profiles differ globally (Weintraub et al., 2017). The adults became resistant to insecticides as a result of spraying another pest. In the 1970s in South America, the gelechid potato moth, *Tuta (Scrobipalpula) absoluta* (Meyrick), was exposed to considerable insecticide application and the non-pestiferous leafminer was drenched in the process (Chavez & Raman, 1987). This exposure imposed selection pressure on *L. huidobrensis,* and by the time the leafminer was carried to Europe and further abroad, adults had developed resistance to many conventional insecticides. In the United Kingdom, resistance of *L. huidobrensis* (and *L. trifolii*) is reported to most carbamate, organophosphate, and pyrethroid insecticides (Macdonald, 1991). Currently, only translaminar and/or systemic larvicides are effective against this pest (Reitz et al., 2013).

Detailed incidences of insecticide resistance for all three *Liriomyza* spp., where available, are provided below.

5.1 Carbamates (1A)

Mechanism of resistance

Resistance of *L. trifolii* to carbamates is considered to be metabolic through the enhanced detoxification by monooxygenases (https://www.irac-online.org/pests/liriomyza-trifolii/; accessed 5 March 2020).

5.2 Organophosphates (1B)

Mechanism of resistance

Synergism and biochemical assays have indicated esterases play a key role in metabolic resistance of *L. sativae* to chlorpyrifos (Askari Saryazdi et al., 2015). A laboratory selected strain of *L. sativae* showed 40-fold resistance to chlorpyrifos compared with a susceptible strain after 23 generations, although in the absence of selection pressure chlorpyrifos resistance was unstable (Askari Saryazdi et al., 2015). The target enzyme of chlorpyrifos, acetylcholinesterase (AChE), was also more active in the chlorpyrifos-resistant strain compared with the susceptible strain. This suggests both overactivity of metabolic enzymes and reduction in target site sensitivity are joint factors contributing to chlorpyrifos resistance in *L. sativae,* however, the molecular mechanism of modified AChE in *L. sativae* is still unknown (Askari Saryazdi et al., 2015). Interestingly, adult *L. sativae* had lower resistance to chlorpyrifos than larval stages, and higher activity of detoxification enzymes in larvae were observed, compared with adults. This discrepancy is potentially related to the different diets of the two life stages (Askari Saryazdi et al., 2015).

In *L. trifolii*, resistance was thought to be due to an insensitive target acetylcholinesterase (Broadbent & Pree, 1989). Although the authors ruled out enhanced metabolism due to glutathione-S-transferases or esterases through the addition of the synergists, DEM and TPP, they could not rule out enhanced metabolism by mixed function oxidase activity as these assays were not performed (Broadbent & Pree, 1989). The mechanism of resistance is now known to be oxidative and hydrolytic metabolism (https://www.irac-online.org/pests/liriomyza-trifolii/; accessed 5 March 2020).

Despite the high levels of resistance shown by *L. trifolii*, reversion to susceptibility to organophosphates and pyrethroids has been shown to occur within a few generations (within 1 year) (Broadbent & Pree, 1989; Parrella & Trumble, 1989).

Cross resistance

Chlorpyrifos resistant *L. sativae* exhibited no cross-resistance to other insecticides (abamectin, azadirachtin, chlorantraniliprole, cyromazine, fenpropathrin, spinosad, thiamethoxam) except for diazinon, another organophosphate from the same IRAC MoA group as chlorpyrifos (Askari Saryazdi et al., 2015).

A greenhouse population of *L. trifolii* highly resistant to pyrazophos exhibited cross-resistance to the phosphorothioate insecticides demeton, chlorpyrifos, and triazophos (Broadbent & Pree, 1989).

5.3 Cyclodiene organochlorines (2A)

Mechanism of resistance

L. trifolii shows metabolic resistance to this class of chemicals (https://www.irac-online.org/pests/liriomyza-trifolii/; accessed 5 March 2020).

5.4 Pyrethroids (3A)

Mechanism of resistance

L. sativae collected from sites in Vietnam where cypermethrin resistance was detected were found to have a mutation in the sodium channel gene associated with pyrethroid resistance (knockdown resistance; *kdr*) (Johansen et al., 2003). Previous studies have also shown that *L. sativae* is capable of evolving resistance to synthetic pyrethroids within 11-26 generations, depending on the dosage and repeat application timing (Mason, Tabashnik, & Johnson, 1989). Synergism and inhibitor tests with fenpropathrin resistant *L. sativae* populations from Iran suggest glutathione S-transferases and carboxyl esterases are not involved in fenpropathrin resistance in this species, rather P450 monooxygenases are the main metabolic enzymes involved (Askari Saryazdi et al., 2014). Esterases also have a major role in resistance of *L. sativae* to fenpropathrin (Askari Saryazdi et al., 2014).

Liriomyza trifolii from California and Florida, USA displayed, in topical application studies, resistance to permethrin, methyl parathion, methamidophos (organophosphate), and a mixture of methyl parathion and permethrin (Keil & Parrella, 1990). Oxidative metabolism was implicated as possible resistance mechanisms but did not account for all resistance to permethrin. The authors suggested that a nerve insensitivity factor, *kdr-like*, was also likely to be involved in producing permethrin resistance. In another population from Florida, USA, moderate resistance was reported for permethrin and methamidophos (organophosphate), but declined over three months of colonisation to levels similar to that of a susceptible colony. Assays with synergists showed that oxidative and hydrolytic metabolism may have contributed to resistance in this population (Keil & Parrella, 1990).
Resistance to synthetic pyrethroids is the type of resistance most likely to be present in the *L. sativae* population currently found in Australia, given that pyrethroid resistance has been detected in Vietnam, and is suspected in Indonesian populations of *L. sativae* (Hidrayani et al., 2005). This potential resistance risk further negates the need for synthetic pyrethroid registrations against *L. sativae* in Australia and emphasises the need for the registration of multiple chemical classes to reduce the risk of further resistance evolving.

Cross resistance

Californian and Florida populations of *Liriomyza trifolii* that displayed resistance to permethrin and methamidophos were also cross-resistant to DDT and cypermethrin (Keil & Parrella, 1990).

In Hawaii, despite only a short history of pyrethroid use, high levels of tolerance to fenvalerate and permethrin were detected in field populations of *L. trifolii,* with lower levels in *L. sativae* (Mason, Johnson, & Tabashnik, 1987). It was speculated that the tolerance/resistance appeared as a result of cross-resistance to longer used organochlorine insecticides, which have a similar mode of action to pyrethroids (Mason et al., 1987).

5.5 Acetamiprid (4A)

There are no reports of resistance to any *Liriomyza* sp. against this compound, however it is not registered in the USA for *Liriomyza* spp.

5.6 Spinosyns (5) and diamides (28)

Mechanism of resistance

Resistance to spinosyns (IRAC MoA Group 5) and diamides (IRAC MoA Group 28) has been documented in *L. trifolii* (Ferguson, 2004; Gao et al., 2017). In most cases of insecticide resistance in *L. trifolii* examined to date, resistance has been unstable, with reversion to susceptibility occurring in 5-10 generations when reared in the absence of insecticide selection pressure (Gao et al., 2017). However, once resistance has evolved to permethrin in *L. trifolii* the resistance levels decline slowly, with field resistance still high enough to render maximum field rates ineffective after 15 generations without exposure (Parrella & Trumble, 1989). Altered target site resistance is through a G275E mutation in the α6 subunit of nAChR (https://www.irac-online.org/pests/liriomyza-trifolii/; accessed 5 March 2020).

Cross resistance

As there is no cross-resistance among spinosysns and diamides, they can be used in rotation for leafminer control in a resistance management strategy (Gao et al., 2017).

5.7 Avermectins (6) and cyromazine (17)

Mechanism of resistance

After 22-25 generations, laboratory-selected *L. sativae* populations showed resistance to abamectin at 40-50 times a susceptible population (Wei et al., 2015). A significant difference in glutathione S-transferases enzyme activity was found between larvae of abamectin-resistant strains and the susceptible strain. Glutathione S-transferases enzyme activity was increased by 5.6–6.6 fold in the resistant strains, possibly indicating a role of this enzyme in metabolic abamectin resistance in *L. sativae* (Wei et al., 2015).

Resistance to cyromazine showed that reversion to susceptibility occurred within eight generations in a laboratory strain and that field efficacy was restored within two seasons of reduced exposure (Schuster & Everett, 1983). Cyromazine resistance was again reported in *L. trifolii* from celery in Florida in 1990 (Leibee & Capinera, 1995; Ferguson, 2004). Resistance mechanisms in *L. trifolii* are still somewhat unknown. Under field conditions, it was hypothesised that resistance to abamectin would probably involve an ingestion detoxification mechanism in larvae (Cox, Remick, Lasota, & Dybas, 1995). RAPD-PCR methods have identified various oligonucleotides that are present in abamectin and cyromazine resistant populations and absent in susceptible populations (Ferguson & Pineda, 2010). *L. trifolii* resistance to cyromazine, abamectin and spinosad were reported to be unstable in several populations collected from ornamentals in the USA (Ferguson, 2004). Cyromazine resistance in *L. trifolii* appears to be expressed as an incompletely recessive trait, and not sex-linked. Backcrossing suggests that cyromazine resistance is conferred by a major gene (Leibee & Capinera, 1995).

Cross resistance

There is no cross-resistance to cyromazine in abamectin resistant strains of *L. sativae* or *L. trifolii* (Ferguson, 2004; Wei et al., 2015). As cyromazine and abamectin belong to different IRAC MoA groups, (17 and 6, respectively), cross-resistance between both compounds is unlikely. However, *L. sativae* strains that were exposed to selection pressure for cyromazine resistance in the laboratory exhibited low-level (6.4- to 9.4-fold) cross-resistance to abamectin (but remained susceptible to cyromazine) (Wei et al., 2015).

5.8 Novaluron (15)

There are no reports of resistance to any *Liriomyza* sp. against this compound.

5.9 Spirotetramat (23)

There are no reports of resistance to any *Liriomyza* sp. against this compound, however it is not registered in the USA for *Liriomyza* spp.

5. Pest management strategies

For crops where leaf mining does not disfigure produce, an IPM program promoting beneficial species (parasitoid wasps in particular) is likely to be the most effective way to control *Liriomyza spp*. In the USA, IPM programs used across many crop groups focus on (i) using uninfested seedling transplants, (ii) cultural controls including destroying weed hosts and ploughing senescent crops, (iii) not planting new crops adjacent to old crops, (iv) avoiding broad-spectrum chemical use for leafminer control and other pest control, and (v) using economic thresholds to delay and reduce chemical sprays to allow colonising parasitoid populations to build up (Johnson et al., 1980; Leibee & Capinera, 1995). In crops where leaf

mining causes a cosmetic economic impact, it will still be essential to promote healthy parasitoid populations for *L. sativae* control. The critical time period will be during crop establishment. Early crop mining at low levels should not be significant to crop yield, so it will be important to delay harmful chemical sprays at this stage to allow parasitoids to build up.

Ongoing management strategies for *Liriomyza* should consider biological and cultural controls first, with chemical intervention used when needed based on regular field monitoring.

Cultural controls could include destruction and deep ploughing of old vegetable fields, a host-free period (a 2 month summer period during which important host crops are not grown), avoiding the use of broad-spectrum insecticides, and monitoring *L. sativae* population numbers so that insecticides are applied only when needed. Programs of this kind have successfully managed *Liriomyza* leafminer species in Florida vegetable crops for many years (Gao et al., 2017).

It will also be important to consider how current management practices will affect establishment of *L. sativae* and associated parasitoid populations.

5.1 Chemical use

Liriomyza spp. are notoriously difficult to control with chemical application. Sprays that target adults often are not effective at controlling the immature life stages, as leafminer eggs and larvae develop inside leaves, and mature in the soil. In situations overseas, following establishment, *Liriomyza* leafminers frequently achieve significant pest status on vegetable and ornamental crops. This has been attributed to the use of ineffective insecticides, evolution of resistance, difficulties in targeting chemical control at the protected immature life stages (egg, larvae and pupae), and lack of effective natural enemies in new areas of infestation (Rauf, Shepard, & Johnson, 2000). It is therefore important to have multiple-pronged management strategies, targeting different life stages, in order to reduce the economic impact of these pests.

When chemical control is used, it should be targeted at the immature life stages, and therefore insecticides with translaminar and/or systemic activity are necessary (e.g. abamectin, emamectin benzoate, cyromazine, spinosad, spinetoram, cyantraniliprole, chlorantraniliprole and thiamethoxam). As well as targeting the immature life stages, some of these products also have efficacy against adults. It is important to apply chemicals (either as foliar sprays or soil applications) so they have activity when larvae will be actively feeding in their mines, before they drop out of the mines to pupate, and therefore monitoring to ensure populations are still increasing in crops is essential before applying chemicals. Pupal counts, either by using pupal trays to catch larvae as they exit leaves or by collecting pupae from mined leaves (after 7 days) and counting the larvae/pupae in the trays per week is an effective means of estimating the growth or decline of a leafminer population (Johnson, Oatman, Wyman, & Van Steenwyk, 1980; Foster, 1986).

Internationally, thresholds of economic damage have been developed for some *L. sativae* and *L. trifolii*: 30 and 60 mines/leaf decreased yield by 10% and 20%, respectively in greenhouse-grown tomatoes, and chemical control is recommended if new mines are seen and 25% of the surface of old leaves of pink and kidney beans were mined. Chemical control is also recommended for catches of 15 or more pupae between rows of celery plants, but this is dependent on leafminer species (Jovicich, 2009).

5.2 Resistance Management

Resistance management guidelines developed overseas for *Liriomyza* spp. include the judicious use of insecticide treatments (avoiding unnecessary chemical applications) and rotation between a number of insecticides from different IRAC MoA groups to mitigate resistance evolution (Reitz et al., 2013; Gao et al., 2017). The same insecticide should not be applied in more than three consecutive applications (preferably only two). This use pattern lessens insecticide selection pressure and minimises selection against successive generations with the same product (Reitz et al., 2013; Gao et al., 2017). In particular, recommendations of mixtures or the rotation of cyromazine and abamectin should be considered carefully, as this could increase the evolution of metabolic enzyme-mediated cross resistance (Wei et al., 2015).

5.3 Case Study: *L. sativae*

Two key growing areas in Queensland that may be of risk to *L. sativae* incursion are Bowen and Bundaberg. The key crops grown in these regions include tomatoes, capsicums, chillies, French and runner beans, sweet corn, capsicum, zucchini, melons, sweet potato, herbs, cucumbers, okra, eggplants, pumpkins, snow peas and potatoes. In particular, tomatoes are a major economic crop for each region, and would be impacted by *L. sativae.*

A summary of the chemicals that are currently used to manage pests in tomato crops in the Bowen and Bundaberg regions, and how they might affect *L. sativae* and one of its parasitoids, *Diglyphus isaea* that is expected to play a crucial role in controlling this pest in Australia (Ridland et al., 2020), is shown in Figure 1. Many of the insecticides already being applied to control pests will potentially provide some non-target control of establishing *L. sativae* populations, including abamectin, spinetoram, spinosad, chlorantraniliprole, cyantraniliprole, thiamethoxam, indoxacarb and organophosphate sprays. However, several of the products currently being used in these systems are toxic to parasitoids.

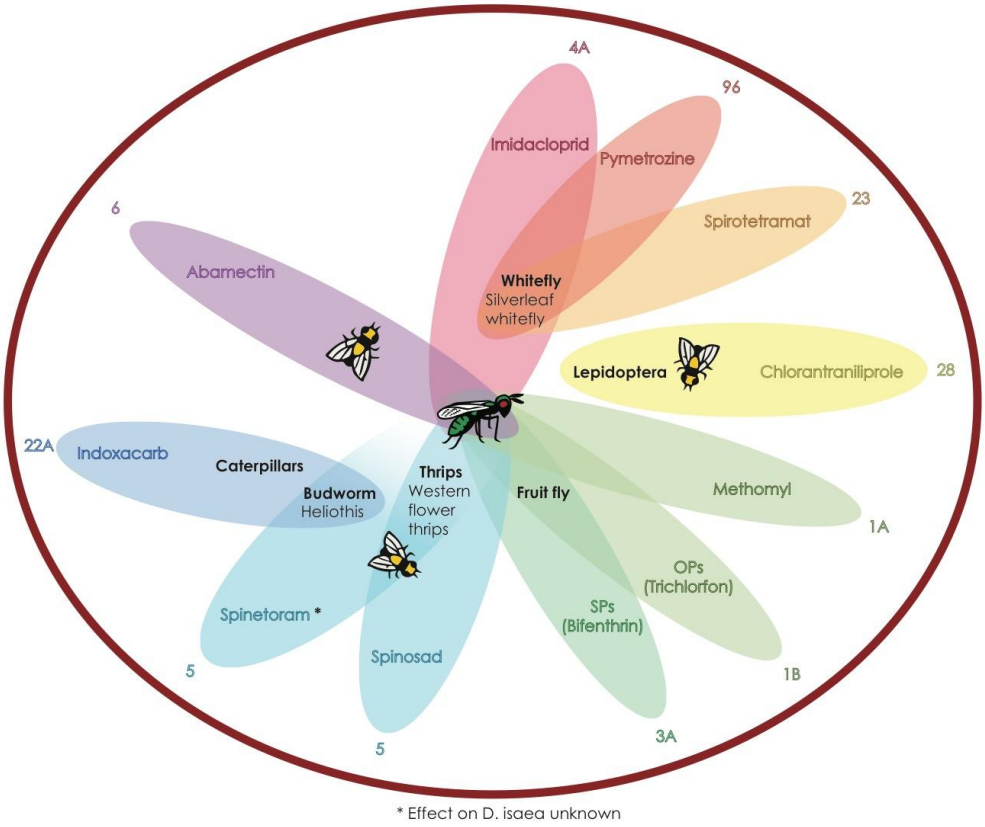


Figure 1. Chemicals presently used for pest control in Bowen and Bundaberg tomato crops (data collected based on consultation with local agronomists). Colours indicate different chemicals and the pests they target. Colours overlapping with the flies indicate instances where non-target control of *L. sativae* may occur, while colours overlapping with the wasp indicate instances where products may be harmful to the parasitoid, *D. isaea*, or other endemic parasitoid species.

As well as considering how current management scenarios will affect *L. sativae* and associated parasitoids, it will be essential to look at how chemical recommendations for leafminer may impact the current pest and beneficial communities. This will be particularly challenging when considering pests that have evolved resistance to multiple chemicals (e.g. *Tetranychus urticae, Helicoverpa armigera, Myzus persicae, Bemisia tabaci* and *Frankliniella occidentalis*). The use of some insecticides may be restricted to certain times or 'windows' during the year, and therefore different chemicals, biological or cultural controls will be needed when targeting *Liriomyza* spp. at these times.

Control of *Liriomyza* spp. in protected or covered situations (i.e. in glasshouses and polyethylene-covered greenhouses and tunnels) will be important. *Liriomyza* populations have been found to survive cold winters by staying inside protected and heated greenhouse environments (Jovicich, 2009). The use of chemicals in protected cropping also differs to field-grown crops, with more extensive OH&S

requirements for personnel using chemicals within an enclosed space, and restrictions on the types of chemicals that can be applied to plants grown in certain conditions, such as hydroponically.

6. Eradication

There are no published examples of eradication of *L. sativae*. However, a protocol for the disinfestation of glasshouse production sites against *L. sativae* was developed by the European and Mediterranean Plant Protection Organisation in 2009. It includes a regime of foliar sprays using chemicals from different IRAC MoA groups with different activities (e.g. translaminar movement, contact activity) (OEPP/EPPO, 2009).

There are several examples of eradication for both *L. trifolii* (e.g. Finland: Rautapää, 1984; United Kingdom: Bartlett & Powell, 1981) and *L. huidobrensis* (e.g. United Kingdom: Cheek, MacDonald, & Bartlett, 1993; Ireland: EFSA Panel on Plant Health, 2012). The broader principles of these eradications also apply to *L. sativae*. In the United Kingdom, *L. trifolii* has been successfully eradicated from multiple incursions (Bartlett & Powell, 1981; Powell, 1981; MacDonald, 1991). Outbreaks of *L. trifolii* occurred in commercial nurseries in England and Wales between 1977 and 1980. An eradication strategy was devised to control all stages, and where possible to use insecticides from different IRAC MoA groups.

The eradication strategy required:

1. Identification of all sources of infested plants.
2. Finding all current outbreaks and new introductions. Crops inspected daily for signs of infestation, involving a slow careful scan of foliage, so even slight damage could be detected. Yellow sticky traps were also used to monitor for adults.
3. Statutory treatment with insecticide and, where necessary, destruction of individual infested plants or, in extreme cases, of entire beds.
4. Soil sterilisation of all infested beds after flowering,
5. Prohibition on the movement of cuttings and flowers from infested nurseries (NB: the sale of cut flowers was normally permitted provided all damaged leaves were removed and burnt).

Eradication programs continued for more than a month, due to the protracted emergence of adults under certain conditions. Weekly visits were made to each infected nursery to inspect the eradication notice, and if infestation continued beyond 3 weeks, nurseries were required to include weed control, sticky traps and crop destruction as part of the eradication program. Weekly visits continued after no further detection of live infestation, and nurseries declared infestation free if two visits were made where no live leafminers were detected. Once a nursery was declared 'infestation free', two more visits were made at monthly intervals, and if no further infestations arose, these routine inspections reduced to twice annually. The programs were sometimes required to be repeated for 2-3 months before eradication was achieved. Eradication measures rarely required the destruction of infested crops but frequently required the selective destruction of infested imported plants. Where the eradication

strategy was correctly followed, eradication of *L. trifolii* infestations from glasshouses usually occurred within a few weeks (Powell, 1981). Table 20 summarises the eradication treatment program for eradication of *L. trifolii* in glasshouse chrysanthemums.

Table 21. Treatment program for eradication of *L. trifolii* in glasshouse chrysanthemums.

Treatment	**Insecticide (IRAC MoA group)**	**Rate**	**Timing**
1. Persistent systemic applied to soil to kill larvae in young growth	Aldicarb* (1A) Oxamyl (1A)	5.6 kg a.i./ha 0.1 g a.i./plant	14-28 days after planting out 10-14 days after planting out and 21 days later
2. Foliar sprays to kill larvae in older leaves	Diazinon (1B) Heptenophos* (1B) Triazophos* (1B)	126 g a.i./500L 37.5 mL a.i./100 L 30 mL a.i./100 L	5 - 7 day intervals 5 day intervals 10 - day intervals (max 3 apps)
3. Knockdown space treatment to kill adults (fumigation or fog)	Lindane* (2A) Nicotine* (4B) Permethrin (3A) Resmethrin* (3A)	20.5 g a.i./280 m^3 200 g a.i./1250 m^3 220 mL a.i./ha 73 mL a.i./ha	7 day intervals 7 day intervals 4 day intervals then 6 day intervals 3 day intervals
4. Soil treatment to kill pupae in beds	Lindane* (2A) Steam sterilisation Methyl bromide (8A)	12.6 mL a.i./100 L 93^oC to 200 mm 3-4 hr 1 kg/10 m^2	Drench at 14 day intervals Once when bed cleared Fumigation of empty glasshouse
5. Soil treatment to kill pupae in pots before marketing	Fenitrothion (1B) Permethrin (3A)	5 mL a.i./ 4 L 1 mL a.i./4 L	24 hours before marketing 24 hours before marketing
6. Knockdown space treatment of empty glasshouse to kill adults	Methyl bromide (8A) Lindane* (2A) Nicotine* (4B) Permethrin (3A)	40 g/m^3 20.5 g a.i./280 m^3 200 g a.i./1250 m^3 220 mL a.i./ha	Fumigation of empty glasshouse for 14 hours by any insecticide in list.

Treatment	Insecticide (IRAC MoA group)	Rate	Timing
	Resmethrin* (3A)	73 mL a.i./ha	

Taken from Bartlett & Powell (1981).

* Indicates insecticides that are not/no longer approved/registered in Australian crops.

While most infestations were contained in glasshouses and protected cropping situations, several situations where the infestation spread outside of the protected cropping structure occurred (Bartlett & Powell, 1981; Powell, 1981). In one case, all infected weeds outside were treated and killed and inspections were made of surrounding farms. Affected crops including chrysanthemums, celery and tomatoes were detected up to 800 metres from the original outbreak. Chrysanthemum crops were subjected to a full eradication program (summarised in Table 33), but celery and tomato crops only had monitoring traps, foliar sprays and soil sterilisation with methyl bromide (Bartlett & Powell, 1981). In another case, eradication was considered impracticable because of the many infested host plants of different species that were present (Powell, 1981). Measures were taken to prevent reinfestation of glasshouses, including monitoring adults, treating infested plants with pesticides, and banning/restricting movement of visitors (Powell, 1981). Due to the impracticality of surveying all known host plants outdoors, inspections for infestations were limited to: (i) a restricted number of preferred hosts in roadside verges, hedges, fields and gardens within about 4.5 km from the originally infested glasshouse, once in mid-summer and again in early autumn, and (ii) all potential host plants in a limited area surrounding the originally infested glasshouses as often as possible from mid-summer through to the end of autumn. In this example, no positive identification of *L. trifolii* was made from hosts found more than 500 m from the original infestation, and therefore the survey area was not extended (Powell, 1981). Spread was apparently limited by physical barriers such as roads boarded by high hedges, banks or trees channelling wind, and other windbreaks (Powell, 1981). *L. trifolii* populations declined as the weather cooled down, even though survival occurred when minimum temperatures of 1.9°C were recorded in early autumn, and active infestations and adults were trapped throughout autumn. No further infestations were observed following the winter, or throughout the next year.

In these examples from the UK, eradication of *L. trifolii* was achieved, but only because growers rigidly followed the eradication strategy. Destruction of entire crops was not often required; however, destruction of individual plants was required when mines, feeding and oviposition punctures were observed. Cost benefit analysis of the eradication procedures found that eradication was justified in these situations (Powell, 1981). The total cost of eradicating one glasshouse/outdoor infestation at Efford in 1979 was GBP36,000, which equates to around AUD240,000 today.

The chemical management elements of an eradication program presented in Table 20 may be applicable if infestations are detected in protected cropping situations outside of the current detection area of *L. sativae* in Australia, or if an incursion of *L. trifolii* or *L. huidobrensis* occurs. These chemical controls would need to be supplemented with cultural controls, and relevant crop destruction and decontamination protocols as per PLANTPLAN: Australian Emergency Plant Pest Response Plan (PHA, 2017). Similarly, the elements of an 'outside' chemical eradication program presented in Table 21 for *L. trifolii* may be applicable if infestations or incursions are detected outside of protected cropping situations. Chemicals that are registered or permitted for use in Australian crops will need to be substituted for outdated chemistries in these original eradication programs. The success of eradication will also depend on the susceptibility of invasive *Liriomyza* populations to available insecticides. It is

likely that populations will be resistant to several pesticides, and therefore eradication programs should not be reliant on these products (MacDonald, 1991; Reitz et al., 2013).

Table 22. Treatment program for eradication of *L. trifolii* in plants outside of infested glasshouses.

Treatment	**Insecticide (IRAC MoA group)**	**Rate**	**Methods**
Weed Control		Mowing, hoeing, rotavating fallow areas and ploughing after harvest to remove all weeds. Herbicide applied where possible.	
Home Gardens	Chlorpyrifos (1B) Trichlorfon (1B)	1 L a.i in 4500 L/ha 2.72 kg a.i. in 9000 L/ha	Drench Foliar spray and drench
Grass swards and headlands	Lindane* (2A) Chlorpyrifos (1B)	3.1 L a.i. in 9000 L/ha 312 mL a.i in 4500 L/ha	Drench Drench
Navy beans, Roadside verges	Trichlorfon (1B)	2.72 kg a.i. in 9000 L/ha	Foliar spray and drench
Runner beans	Heptenophos* (1B) Trichlorfon (1B)	440 mL a.i. in 1125 L/ha 2.72 kg a.i. in 9000 L/ha	Foliar spray Foliar spray and drench
Tomatoes	DDT* (3B) Trichlorfon (1B)	1.1 L a.i. in 1125 L/ha 2.72 kg a.i. in 9000 L/ha	Foliar spray Foliar spray and drench

Taken from Powell (1981).
* Indicates insecticides that are not/no longer approved/registered in Australian crops.

Eradications may be successful in parts of southern Australia where winter conditions may slow or halt *L. sativae* or *L. trifolii* population growth, however it is unlikely that the climate conditions in northern Australia will be limiting to either species population growth, as was observed in the UK examples. Eradication of *L. huidobrensis* which is more cold tolerant than both species could be more challenging in southern regions, however it is expected to be less tolerant of the hotter conditions across much of northern Australia. To date, there are no examples of successful eradication programs against *Liriomyza* leafminer species in warm-climate regions, and the expectations of Jovicich (2009), was that it would be difficult, if not impossible to eradicate *Liriomyza* spp. from Australian regions that have favourable environments for the establishment and presence of numerous hosts. Cost benefit analysis for different areas of Australia that have the potential for *L. sativae, L. trifolii and L. huidobrensis* outbreaks will help to determine if eradication is worthwhile in each instance.

7. Conclusions

Fifteen chemicals were assessed for their potential to be used in the control of *L. sativae, L. trifolii and L. huidobrensis* in Australia, covering recorded insecticide resistance against these pests (Table 24), and toxicity to several key natural enemies of *Liriomyza* spp. (Table 25). A summary of the Australian label registrations and permits of these chemicals in crops that are known to be economically affected by *L. sativae, L. trifolii*, or *L. huidobrensis* is provided in Table 26.

Table 24. The active ingredients that could, or are being utilised for control of *Liriomyza* spp. present or at risk of invading Australia, and their known resistance status.

		Resistance status
Chemical	**MoA Group**	***Liriomyza spp.***
Organophosphates	1B	Y
Synthetic pyrethroids	3A	Y
Acetamiprid	4A	N
Thiamethoxam	4A	Y
Spinosad	5	Y
Spinetoram	5	Y
Abamectin	6	Y
Emamectin benzoate	6	N
Novaluron (IGR)	15	N
Cyromazine (IGR)	17	Y
Indoxacarb	22A	N
Spirotetramat	23	N
Chlorantraniliprole	28	Y
Cyantraniliprole	28	Y
Azadirachtin #	-	N

Can be used for organically grown produce in the USA.
Resistance status of *Liriomyza* spp. (Y = resistance documented; N = No data available).

Table 25. A summary table detailing potential active ingredients that could be utilised for control for *Liriomyza* spp. present or at risk of invading Australia and their respective toxicity to four key parasitoids. Gaps in the table indicate areas where there is no data available.

		Toxicity to Parasitoids			
Chemical	**MoA Group**	***Hemiptarsenus varicornis***	***Diglyphus isaea***	***Neochrysocharis formosa.***	***Opius* spp.**
Organophosphates	1B	VH	H-VH	VH	VH
Synthetic pyrethroids	3A	VH	H-VH	H-VH	VH
Acetamiprid	4A		L-H[1]		L
Thiamethoxam	4A		H[2]	H	L
Spinosad	5		H-VH[3]	M	VH
Spinetoram	5		VH	H-VH	
Abamectin	6	L-M[4]	M-VH[5]	M-VH[6]	VH
Emamectin benzoate	6	VH	M-VH[7]	VH	
Novaluron (IGR)	15		L-M[8]	L	
Cyromazine (IGR)	17	L	L-M[9]		L[10]
Indoxacarb	22A		L-M[8]		L
Spirotetramat	23		L-M		
Chlorantraniliprole	28		L		
Cyantraniliprole	28		L-H[11]		
Azadirachtin #	-		L-M[12]	L	M-H

To account for variability in mortality between laboratory and field trials, pesticide toxicity ratings are categories into L, M, H and VH according to IOBC protocols, where observed mortality in laboratory and extended laboratory trials correspond to <30%, 30-79%, 80-98%, and >99% mortality, respectively, and semi-field and field trials correspond to <25%, 25-50%, 50-75%, and >75% mortality, respectively. Footnotes within the table detail where toxicity ratings were derived, as well as the application method. Where a toxicity rating range is expressed (e.g. H-VH), varying effects are recorded either within or between life stages. Differing toxicity

ratings between life stages or through pesticide application method are captured in the footnotes below. Potential for resistance to occur in *L. sativae* are categorised as L = low, M = medium, and H = high. IGR = Insect Growth Regulator.

[1] A L-H toxicity rating range is reported for *D. isaea* adults when treated with acetamiprid, but only an H rating is reported for larvae. When applied through irrigation and drench application methods, L toxicity ratings are reported for both adults and larvae.

[2] When thiamethoxam is applied through a drench application, toxicity ratings for *D. isaea* adult and larvae are M and H, respectively.

[3] Spinosad has an H and L toxicity rating when applied through irrigation for *D. isaea* larvae and adults, respectively.

[4] Abamectin has a L toxicity rating for adult *H. varicornis*, but a M rating for larvae.

[5] Abamectin has M, VH, and H-VH toxicity ratings to *D. isaea* larvae, pupae, and adults, respectively.

[6] Abamectin has a M-H toxicity rating for *Neochrysocharis formosa* larvae, and a VH rating for adults.

[7] Emamectin benzoate has a VH toxicity rating for adult *D. isaea*, but a M rating for larvae.

[8] Indoxacarb and novaluron have a L toxicity rating for adult *D. isaea*, but a M rating for larvae.

[9] For larva and adult *D. isaea*, L toxicity ratings are reported for irrigation and drench application of cyromazine.

[10] L toxicity ratings are reported for cyromazine drench application for adult *Opius* spp.

[11] Cyantraniliprole has a H toxicity rating for adult *D. isaea*, but a L rating for larvae.

[12] Azadirachtin has a M toxicity rating for adult *Opius* spp., but an H rating for larvae.

Table 26. Labelled use of potential chemicals for registration for *L. sativae* control in Australia. Gaps in the table indicate areas where there are no registered products.

Permit = permits currently exist for this chemical in Australia. Within rates = recommended rates to control *L. sativae* lie within the currently labelled use rates for this chemical. Twice rates = recommended rates to control *L. sativae* are less than twice the currently labelled use rates for this chemical. Rates too low = recommended rates to control *L. sativae* are greater than twice the currently labelled use rates for this chemical.

Crop	Cyromazine	Abamectin	Emamectin benzoate	Spinosad	Spinetoram	Spirotetramat	Cyantraniliprole	Chlorantraniliprole	Dimethoate	Thiamethoxam	Indoxacarb	Azadirachtin
Onions		Permit 81876					twice label rates					
Leeks		Permit 81876	within rates		twice rates		twice label rates					
Spring, shallots		Permit 81876			twice rates		twice label rates					
Lettuce		twice rates	within rates		twice rates	Permit 88640		rates too low			twice rates	
Silverbeet and spinach		Permit 81876	within rates		twice rates			rates too low			twice rates	
Asian vegetables		twice rates	within rates		twice rates			rates too low			twice rates	

Crop	Cyromazine	Abamectin	Emamectin benzoate	Spinosad	Spinetoram	Spirotetramat	Cyantraniliprole	Chlorantraniliprole	Dimethoate	Thiamethoxam	Indoxacarb	Azadirachtin
Cabbages		Permit 81876	within rates		twice rates			rates too low			within rates	
Broccoli			within rates		twice rates			rates too low			within rates	
Cauliflower			within rates		twice rates			rates too low			within rates	
Brussels sprouts			within rates		twice rates			rates too low			within rates	
Radish		Permit 81876	within rates		twice rates							
Swedes and turnips		Permit 81876	within rates		twice rates							
Beetroot		Permit 81876	within rates		twice rates							
Potatoes		Permit 81876			twice rates		twice label rates	rates too low				
Carrots		Permit 81876			twice rates							
Parsnips		Permit 81876	within rates		twice rates							
Zucchini and squash		Permit 81876			twice rates		twice label rates	rates too low				

Crop	Cyromazine	Abamectin	Emamectin benzoate	Spinosad	Spinetoram	Spirotetramat	Cyantraniliprole	Chlorantraniliprole	Dimethoate	Thiamethoxam	Indoxacarb	Azadirachtin
Pumpkins		Permit 81876					twice rates	rates too low				
Cucumbers		Permit 81876			twice rates		twice rates	rates too low				
Melons		Permit 81876			twice rates		twice rates	rates too low				
Peas (green)					within rates			rates too low				
Snow and sugar snap peas		twice rates	within rates		within rates	Permit 88640		rates too low				
Beans		Permit 81876			within rates	Permit 88640		rates too low				
Tomatoes		Permit 81876	within rates		twice rates	Permit 88640	twice rates	rates too low		rates too low	within rates	
Capsicums and chillies		Permit 81876	within rates		twice rates	Permit 88640	twice rates	rates too low			within rates	
Eggplants		Permit 81876	within rates		twice rates	Permit 88640	twice rates	rates too low			within rates	
Celery		Permit 81876	within rates		twice rates	Permit 88640		rates too low			within rates	
Rhubarb*						Permit 88640						

Crop	Cyromazine	Abamectin	Emamectin benzoate	Spinosad	Spinetoram	Spirotetramat	Cyantraniliprole	Chlorantraniliprole	Dimethoate	Thiamethoxam	Indoxacarb	Azadirachtin
Herbs					twice rates	Permit 88640						
Pulses									Permit 89184			
Ornamentals (non-food)	Permit 83506				twice rates							rates too low
Nursery stock (food)								rates too low		rates too low		
Nursery stock (non-food)	Permit 83506						within rates	rates too low		rates too low	twice rates	
Home garden use		labelled use		labelled use	labelled use					labelled use		labelled use

*not a known host of *Liriomyza sativae, L. trifolii* or *L. huidobrensis*

Due to potential issues surrounding insecticide resistance of all three *Liriomyza* spp., synthetic pyrethroids are not considered to be viable long-term control options. Synthetic pyrethroids are also highly toxic to the parasitoid wasps of *Liriomyza* spp., as well as other important beneficial species that are important in many Australian crops (Table 35). Due to resistance concerns, it will be important to register several different chemicals (from different IRAC MoA groups) against *Liriomyza spp.* in important crop groups, to allow a resistance management strategy of chemical rotation to be put in place. A variety of chemicals that target the different life stages of *Liriomyza spp.* will also be important (e.g. translaminar and systemic chemicals).

Internationally, two of the most effective insecticides for *Liriomyza* spp. management are abamectin (IRAC Group 6A) and cyromazine (IRAC Group 17) (Reitz et al., 2013). These insecticides target the larvae inside the plant foliage as they both have translaminar properties. Spinosyns have also been effective against *Liriomyza* spp and similar to abamectin and cyromazine, have translaminar properties, enabling them to target larvae (Reitz et al., 2013). All three insecticides have seen well over two decades of use, yet resistance has not been a major problem (Ferguson, 2004).

Due to the importance of beneficial organisms (especially parasitoid wasps), any future decisions around chemical permits and/or registrations for *Liriomyza* spp. control should consider the possible non-target effects of each product. Furthermore, management plans in different crop groups should be developed that consider the beneficial community aligned with that crop group. Ideally chemistries that are less toxic to these beneficials will be selected for *Liriomyza spp.* control (e.g. cyromazine, azadirachtin and cyantraniliprole), however it will be important to have registrations for a broader array of chemicals. Timing the use of less selective chemicals during periods when parasitoid wasps are less likely to be active will lessen the impact of those chemicals. Most broad-spectrum synthetic insecticides that are highly toxic to parasitoids of *Liriomyza* spp. (Hidrayani et al., 2005; Oatman & Kennedy, 1976; Saito et al., 1992; Schuster, 1994) including carbamates, organochlorines, organophosphates and pyrethroids, are also insecticides that show limited efficacy against *Liriomyza* spp. (Hara, 1986; Hidrayani et al., 2005). Therefore, these types of insecticides should be used with great caution in systems where *Liriomyza* spp. are key pests (Reitz et al., 2013).

All chemicals examined in this review have registered labels or permits for use in Australian crops, although the use of azadirachtin and cyromazine is only allowed in ornamentals and/or non-food crops (Table 36). Abamectin, chlorantraniliprole and cyromazine already have labels or permits for the control of leafminer flies in certain crops. In addition, there will potentially be a degree of non-target control of *L. sativae* (and other *Liriomyza* spp. that may incur) in many crops given growers already use many of the chemicals that are efficacious for leafminer control to target other pests. For example, seedling drenches and drip irrigation treatments of chlorantraniliprole and thiamethoxam are common in many vegetable crops, and the persistence of these systemic chemicals may control immature life stages in the soil (pupae) and leaves (larvae). The rate ranges registered to control *Liriomyza* spp. overseas lie within the current registered rates on Australian labels (or permits) for abamectin, cyromazine,

indoxacarb and azadirachtin, although in Australia these chemicals do not cover all important crop groups for *L. sativae*. The lowest overseas rates for control of *Liriomyza* spp. with spinetoram, cyantraniliprole, emamectin benzoate and thiamethoxam overlap with some of the highest registered rates for these products in Australian crops. The registered rates in Australian crops for spinosad and chlorantraniliprole are much lower than the recommend rates from overseas to control *Liriomyza* spp. Registration of higher rates in various crop groups would be necessary for these products.

Control of *Liriomyza* spp. in ornamental crops and in non-food nursery stock by cyromazine should be possible under permit 83506. This permit allows use of cyromazine in protected cropping situations. Any further permit applications for the control of *Liriomyza* spp. in protected cropping will need to address the specific OH&S needs of chemical use in these situations.

Eradication strategies may play a role initially if there are small outbreaks of *L. sativae*, or other *Liriomyza* spp. in isolated Australian horticultural cropping regions. Eradication is likely to be expensive and is unlikely to work effectively, particularly in warmer regions of Australia. If outbreaks occur in southern Australia where winter temperatures will slow or even stop *L. sativae* and *L. trifolii* population growth, or in protected cropping systems (i.e. greenhouses), swift application of an eradication strategy may help provide area freedom in certain circumstances. Some organophosphate chemicals are efficacious against *Liriomyza* spp., and emergency permits for eradication use for these chemicals (e.g. chlorpyrifos) would be worthwhile. It will be important to include weeds and roadside verges in the registration of any chemicals being considered for *Liriomyza* spp. eradication. Target weeds and native plants for control would include those from plant families that are hosts of *Liriomyza spp.* (Spencer, 1990; Jovicich, 2009).

8. Recommendations

1. Apply for a minor use permit for cyromazine for the control of *L. trifolii* in the following vegetable crops: Broccoli, Fruiting vegetables – cucurbits, Fruiting vegetables – other than cucurbits (excluding mushrooms and corn), Head lettuce, Legume vegetables, Root and tuber vegetables, Stalk and stem vegetables.

2. Apply for a minor use permit for abamectin for the control of *L. trifolii* in the following vegetable crops: Fruiting vegetables, including cucurbits (excl. sweet corn and mushrooms), leafy vegetables (excl. lettuce), Legume vegetables, Root and tuber vegetables, Bulb onions, Cabbage (head), Celery, Rhubarb, bulb vegetables (incl. leeks and spring onions).

3. Apply for a minor use permit for indoxacarb for the control of *Liriomyza* spp. in brassica, fruiting vegetables and potentially leafy vegetable crops (pending availability of supporting data). The recommended rates for *L. sativae* control falls within the current labelled use of indoxacarb in the brassica and fruiting vegetable crops, and the recommended rates for *L. sativae* control fall within twice the current labelled use of indoxacarb in the leafy vegetable crops.

4. Current labelled rates of thiamethoxam products in Australian crops are less than half the recommended rates for *L. sativae* control. While permit applications for this product against *L. sativae, L. trifolli* or *L. huidobrensis* is not recommended at this time, due to a lack of residue data, the ongoing use of these products in crops that will be impacted by these pests may provide some incidental control. It would be prudent to consider this when devising chemical management guidelines.

5. Apply for a minor use permit for acetamiprid and novaluron for *Liriomyza* spp..

6. There are several products currently available that could provide control of *L. sativae* in home gardens. Including *L. sativae*, *Liriomyza* spp. or 'leafminer flies' on the labels of home garden use products that contain abamectin, spinosad, spinetoram, thiamethoxam or azadirachtin would provide a range of products for control of *L. sativae* in home gardens.

7. Prepare emergency/eradication permits: Several permit applications have been prepared by Plant Health Australia and may be held by Hort Innovation, or State administration bodies and should cover relevant crop groups. Chemical rates may be higher for the emergency/eradication permits compared with minor use permits, as crop destruction will form part of the eradication process. These permits should be linked to contingency plans for *Liriomyza* spp. preparedness and management. It should be noted that due to the impact of organophosphates on beneficial insects these pesticides should only be used for the eradication of *Liriomyza* spp. rather than as part of an ongoing management regime.

8. Gather information on current chemical management practices in crops that are economically vulnerable to *L. sativae* in different growing regions in order to assess the likely non-target effects of these practices on future leafminer (and parasitoid) populations.

9. Understanding the insecticide resistance status of Australian populations of *L. sativae* and *L. huidobrensis* would be valuable. In the absence of an established genetic diagnostic for this species, the most likely approach to determine resistance is to conduct laboratory phenotypic bioassays. Given the present (restricted) location of *L. sativae* in Australia, and in the absence of a known 'insecticide-susceptible' population, this is challenging. There will be greater opportunities for phenotypic assays on SLM in Australia. Molecular research is warranted and should initially focus on known resistances in closely related species (e.g. mutations in the *kdr* gene known to confer resistance to synthetic pyrethroids).

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Appendix 1: Permits for the control of *Liriomyza* spp. on different host crops

Insecticide control options for different commercial host plant groups of vegetable leafminer, *Liriomyza* spp. and notes where suitable pesticides are permitted and approved by the APVMA for control on each host crop. Blank cells denote where suitable pesticides have not yet been approved or identified. Refer to the APVMA permit database (https://portal.apvma.gov.au/permits) or the most up to date information on current leafminer permits.

Table 27. Insecticides control options for different crop groups, as of December 4, 2020.

Relevant Crop group	**Crop/host plant**	**Pesticide**	**Permit number(s)/ status**	**Target pest (as per permit)**	**Expiration date**	**Withholding information/notes**	**Suitable for management or eradication[1]**
Brassica vegetables	Bok choy; pak-choy; turnip	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Broccoli	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication

[1] Some pesticides will be more suitable for eradication than for the ongoing management of the pest. This is because either they will need to be used at high rates to be effective, there are potential resistance issues, or the pesticide is not likely to fit into existing IPM systems as well as alternative pesticides. Other chemicals provide good control of the pest and are also compatible with IPM systems and will be more suitable for the ongoing management of the pest. To highlight this, pesticides are noted for either eradication, or for management and eradication.

Brassica vegetables	Broccoli	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Brussels sprouts	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Brussels sprouts	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cabbage	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication

Brassica vegetables	Cauliflower	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cauliflower	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Indian mustard	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Bulb vegetables	Garlic	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Garlic	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Bulb vegetables	Leek	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	30 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Shallots	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Shallots	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication

Bulb vegetables	Spring onion	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Cereal grains	Barley	No suitable pesticides identified					
Fruit trees (non-bearing)	Any trees that will not bear fruit for the next 12 months	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Cucumber	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits –	Pumpkin (e.g. butternut)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae,*	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

including melons)				*L trifolii, L. huidobrensis*)			
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Eggplant	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables	Okra*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication

(other than cucurbits)							
Fruiting vegetables (other than cucurbits)	Okra*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Okra*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management

Herbs	Basil	No suitable pesticides identified					
Herbs	Oregano	No suitable pesticides identified					
Herbs	Parsley[2]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management
Head lettuce	Lettuce (Head type only)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Lettuce	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Leafy vegetables (including	Spinach	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication

[2] Note: Parsley is not a recorded host for *L. sativae* but may be a host of other *Liriomyza* spp. and is listed as one of the crops that the permit covers.

brassica leafy vegetables)							
Leafy vegetables (including brassica leafy vegetables)	Spinach	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Leafy vegetables (including brassica leafy vegetables)	Spinach and silverbeet	Chlorantraniliprole	PER87631	*Liriomyza* spp. (including*: L. sativae, L brassicae, L. huidobrensis*)	30 June 2024	3 days (harvest) 7 days (grazing or cutting for stock food)	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	7 days (harvest) 7 days (grazing and hay)	Management
Legume vegetables	Faba bean, broad bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Legume vegetables	Faba bean, broad bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spinetoram	PER87878	*Liriomyza* spp.	28 February 2023	3 days (harvest); 14 days (grazing and hay) Do not allow dairy cattle to graze treated forage	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest); 3 days (grazing and hay)	Management

Legume vegetables	Red bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Red bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Not classified	Field mustard	No suitable pesticides identified					
Not classified	Lucerne	No suitable pesticides identified					
Nursery stock (non-food)	Various non-food nursery plants (e.g. chrysanthemum etc.)	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Nursery stock (non-food)	Seedlings, tubes and plugs, potted colour, trees and shrubs, foliage plants, palms, grasses, fruiting	Abamectin Azadirachtin Cyromazine Emamectin Chlorantraniliprole + thiamethoxam Cyantraniliprole	PER88977	Leafminers (*Liriomyza* spp.) including Vegetable leafminer	30 November, 2022	Not required when used as directed	Management or eradication

	plants (non-bearing), cut flowers and ornamentals.	Indoxacarb Spinetoram		(*Liriomyza sativae*)			
Oilseeds	Cotton	Spinetoram has been identified as suitable for the control of Vegetable leafminer and other *Liriomyza* spp. on cotton as overseas and Australian use patterns are comparable. However, no permit has been applied for this use to date					
Pulses	Pulses	Dimethoate	PER89184	*Liriomyza sativae, Liriomyza trifolii, Liriomyza huidobrensis*	31 March 2025	Harvest: Do not harvest for 14 days after application. Grazing: Do not graze or cut for stock food for 14 days after application.	Management
Root and tuber vegetables	Beets (beetroot, silver beet, sugar beets)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Beets (beetroot, silver beet, sugar beets)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Carrot	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Carrot	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Root and tuber vegetables	Potato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Potato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Potato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Radish	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Radish	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Celery	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication

Stalk and stem vegetables	Rhubarb[3]	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication

[3] Note: Rhubarb is not a recorded host for *L. sativae or L. huidobrensis* but may be a host of other *Liriomyza* spp. and is part of the 'Stalk and Stem Vegetable Crop Group' that the permit covers.

Appendix 8: C&E Outputs developed throughout the project

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Madeleine Quirk [1], Dr. Elia Pirtle [2]

[1] AUSVEG, [2] Cesar Australia

December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Table 1. Industry communication and engagement

Output	Target audience	Output location
Information webpage	Industry, community, government	www.ausveg.com.au/mt16004
Project information flier*	Industry and community	https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#project_flyer
Species factsheet	Industry and community	https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#leafminer_awareness
Standard VLM PowerPoint presentation	Industry	Extension resource package www.ausveg.com.au/mt16004
Exotic leafminers speed drawing	Industry	https://www.youtube.com/watch?v=F-gRuC9TaO0&t (2020) https://www.youtube.com/watch?v=oNvMXQGuAYM&feature=youtu.be (2018)
Northern Peninsula Area/ Torres Strait Island grower and wasp guides*	Community	https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#torres_strait_npa
Full length project webinar	Industry	https://www.youtube.com/playlist?list=PL4cJZzAUfvKB03Pvihu6E-P5mR3sS4dXy
Webinars	Industry	http://www.soilwealth.com.au/resources/videos-and-apps/biosecurity-lessons-in-planning-and-response-for-the-vegetable-industry-webinar-recording/ (national) https://www.youtube.com/playlist?list=PL4cJZzAUfvKAtqZd-vEbGlIuA8pB1caqj (Western Australia) https://www.youtube.com/watch?v=G_pAcZuDRdg&t=333s (Western Australia) https://www.youtube.com/watch?v=u4p-bfY9gZ0&t=2314s (Tasmania) https://www.youtube.com/watch?v=St4AopGdcV0&t=249s (Melbourne) https://www.youtube.com/watch?v=5b4g09hhVHU (melons industry update) https://www.youtube.com/playlist?list=PL4cJZzAUfvKArW1Rd0JZIAYZL21iptD-W (Gippsland)

Output	Target audience	Output location
		https://www.youtube.com/playlist?list=PL4cJZzAUfvKCuPGOIyFiBINAjYt-muXwA (Queensland)
Webinars	Community	Webinar for Bundaberg Fruit and Vegetable Growers' association
Doorstep science video	Industry	Native parasitoid wasps: tiny biosecurity officers https://youtu.be/frtIc22zVmc
Leafminer preparedness guide*	Industry	https://ausveg.com.au/app/uploads/2020/07/1303CR2_Preparedness-guide_FINAL_150620.pdf
Leafminer management guide*	Industry	https://ausveg.com.au/app/uploads/2020/07/1303CR2_Management-guide_FINAL_150620.pdf
Regional response plans*	Industry	https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#leafminer_management
Information package to support GIA's PestID identification tool*	Industry	Updated biological and management information and imagery relevant to VLM, ASLM, SLM, *L. brassicae, L. chenopodii P. syngenesiae,* and beneficial parasitoid wasps provided to GIA to update PestID guide listing for each species https://www.pestid.com.au/
Project articles Included in *Vegetables Australia* magazine (reach: 6000), Potatoes Australia magazine (reach: 3000), Greenlife Industry Australia e-magazines, WA Grower magazine, Lockyer Valley Growers Association, and Melon News	Industry and community	Horticulture industries get active in preparing for exotic leafmining pests: https://ausveg.com.au/app/uploads/publications/Vegetables-Australia_November-December-2017_Web.pdf (page 12) Community gardens provide surveillance and sampling opportunities for vegetable industry: https://ausveg.com.au/app/uploads/publications/Vegetables-Australia_March-April-2018_Web-1.pdf (page 32) Enhancing future preparedness for the vegetable leafminer: https://ausveg.com.au/app/uploads/2019/02/Vegetables-Australia_July-August-2018_Web-29.pdf (page 56) Serpentine leafminer: A threat to the potato industry: https://ausveg.com.au/app/uploads/2019/02/Potatoes-Australia-Feb-Mar-2019-Serpentine-leafminer.pdf Repeated in Greenlife Industry Australia's e-magazine

Output	Target audience	Output location
		American serpentine leafminer: A threat to horticulture: https://ausveg.com.au/app/uploads/2020/01/American-Serpentine_VA-Summer.pdf Vegetable leafminer under the microscope in far north Queensland: https://ausveg.com.au/app/uploads/2019/08/VLM-article-Vegetables-Australia.pdf How parasitoid wasps are helping to fight the leafminer battle https://ausveg.com.au/app/uploads/2020/04/AUSVEG_VegetablesAustralia_Autumn_2020_WEB-150DPI_F01v1.pdf (page 54) Repeated in Melon News magazine (print only) Preparing for exotic leafminers: a threat to Australian horticulture: https://ausveg.com.au/app/uploads/2019/08/VLM-article-Vegetables-Australia.pdf Lockyer Valley Growers Association Newsletter (2020) VLM pest profile Keep an eye out for the vegetable leafminer: WA Grower magazine (Autumn 2020) https://issuu.com/vegetableswa/docs/wagrower_autumn_20_lr__2_ VLM update Bundaberg 'Fresh Pickings' e-magazine
e-Newsletters	Industry	*Several frontline e-bulletins were published throughout the project. The Frontline e-Bulletins are distributed to more than 2900 stakeholders.* <u>2017</u> • Announcement of new Hort Innovation funded project - MT16004 - RD&E Program for control, eradication and preparedness for Vegetable Leafminer (2017-2020) <u>2018</u> • Upcoming workshop - creating value from vegetable waste and managing vegetable leafminer • Upcoming events - Tyabb VIC and Bundaberg QLD <u>2019</u>

Output	Target audience	Output location
		• Event: Putting R&D into practice farm walk and talk Greater Sydney • Upcoming pest and disease workshops in Queensland - Ayr and Bowen • Upcoming potato pest and disease workshops in Virginia SA (23 August 2019) and Devonport TAS (30 August 2019) <u>2020</u> • Upcoming workshops in Bundaberg QLD and Darwin NT • Upcoming webinar - Learn about Exotic Leafminers (repeated) • Learn about exotic leafminers with this new video • Suspect serpentine leafminer (*Liriomyza huidobrensis*) detection in Western Sydney, NSW • Upcoming Gippsland-focused webinar *Several e-Newsletters were published in the AUSVEG Weekly Update throughout the project. The Weekly Update is distributed to more than 4,300 stakeholders.* <u>2017</u> • New project tackling vegetable leafminer • Project update - vegetable leafminer edition of InfoVeg Radio now available • AUSVEG in the media <u>2018</u> • VegNET workshop to look at creating value from veg waste and managing leafminer • VegNET workshop gives growers hands-on experience in pest surveillance • Werribee field day to show Victorian growers the future of farming • VegNET shows growers the future of farming in Werribee • Leafminer research update • Learn more about the vegetable leafminer with informative video <u>2019</u> • Vegetable leafminer project partners investigate serpentine leafminer, a threat to the potato industry • Leafminer preparedness project predicts potential pest populations • AUSVEG engagement and extension team heads to the Northern Territory • Biosecurity workshop helps Atherton growers identify problem pests • E.E. Muirs and Sons Brassica and Leafy Vegetables Agronomy and Sales Workshop in Werribee

Output	Target audience	Output location
		• Vegetable leafminer under the microscope 2020 • Exotic pest profile: American serpentine leafminer • Event: Biosecurity and beneficials workshop • Project update: vegetable leafminer • Webinar: Biosecurity and plant health update • TPP, fall armyworm and exotic leafminers webinar series • Exotic leafminers webinar series • How parasitoid wasps are helping fight the leafminer battle • Learn about exotic leafminers with this new video! • Watch: Exotic leafminers video! • Report suspect serpentine leafminer • Upcoming Gippsland-focused webinar *An AUSVEG SA article was distributed.* 2020 • R&D spotlight: keep an eye out for exotic leafminers
Web articles*	Industry	http://cesaraustralia.com/latest-news/all/preparing-for-the-vegetable-leafminer-an-emerging-biosecurity-threat-to-australian-agriculture http://cesaraustralia.com/latest-news/all/far-north-queensland-wasp-collections-have-exciting-implications-for-management-of-the-new-vegetable-leafminer-pest-in-australia http://cesaraustralia.com/latest-news/all/a-surveillance-toolkit-in-development-for-detecting-vegetable-leafminer http://cesaraustralia.com/latest-news/all/we-re-all-abuzz-about-our-tiny-biosecurity-officers http://cesaraustralia.com/latest-news/all/environmental-dna-is-improving-early-detection-surveillance-for-a-notorious-exotic-pest-the-vegetable-leafminer http://cesaraustralia.com/sustainable-agriculture/pestfacts-south-eastern/issue-10-19-october-2020/the-serpentine-leafminer-detected-in-australia/

Output	Target audience	Output location
		http://cesaraustralia.com/latest-news/all/the-serpentine-leafminer-get-up-to-speed-on-this-recently-detected-exotic-pest
Podcasts and radio	Industry	Project overview: https://soundcloud.com/ausveg/episode-13-infoveg-radio-october-2017-vegetable-leafminer Project results - melon industry specific: https://www.listennotes.com/podcasts/talking-melons/exotic-leafminers-by-dr-elia-YEDGg46tueS/ 2 x interviews with TSIMA 4MW Radio (Torres Strait radio)
Social media	Industry and community	Notable Twitter statistics: Serpentine leafminer article - reach - 6925 impressions Biosecurity workshop in Darwin - reach - 2,742 impressions
Industry presentations and grower visits	Industry	• Devonport (TAS), October 2017 • Perth (WA), October 2017 • Bowen (QLD), November 2017 • Bundaberg (QLD), November 2017 • Tyabb (VIC) March 2018, 9 growers in attendance • Mareeba/Atherton (QLD), April 2018, 16 attendees • Bundaberg (QLD), April 2018, 13 attendees • Werribee (VIC), April 2018, 29 attendees • Lardner Park (VIC), May 2018, 34 attendees • Shepparton (VIC), May 2018 • Darwin (NT), February 2019, 35 attendees • E.E Muirs and Sons Agronomy and Sales Conference (Werribee, VIC), July 2019, 40-50 attendees • Mareeba/Atherton (QLD), July 2019, 30 attendees • Virginia (SA), August 2019 • Devonport (TAS), August 2019 • Ayr (QLD), September 2019, 26 attendees • Bowen (QLD), September 2019, 24 attendees • Bundaberg (QLD), February 2020, 10 attendees • Darwin (NT), February 2020, 20 attendees

Output	Target audience	Output location
		Grower visits undertaken in Darwin (NT), Katherine (NT), Bowen (QLD), Ayr (QLD), Mareeba (QLD), Atherton (QLD), Bundaberg (QLD), Lockyer Valley (QLD), Coffs Harbour (NSW), Greater Sydney, Bacchus Marsh (VIC), Werribee (VIC), Mornington Peninsula (VIC), Cranbourne (VIC), Clyde (VIC), Adelaide Plains (SA), Virginia (SA), and Perth (WA). Mock surveillance training and data collection activities undertaken in Tyabb (VIC) and Werribee (VIC) (led by Cesar Australia) and additional mock surveillance training activities undertaken Atherton (QLD) (led by AUSVEG).
External news articles		https://www.hortidaily.com/article/9175315/american-serpentine-leafminer-a-threat-to-all-of-horticulture/ https://www.hortidaily.com/article/9027246/australia-enhancing-future-preparedness-for-vegetable-leafminer/

* Output developed by Cesar Australia

Table 2. Cape York and Torres Strait Islands – communication and engagement

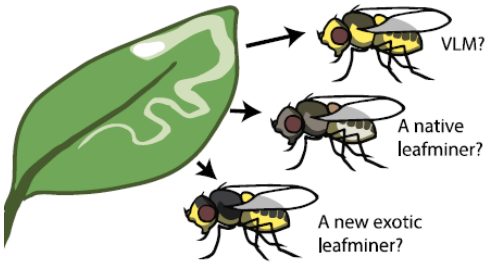
Output	Target audience	Description / output location
Cape York Peninsula scoping study and tourism survey	Industry	Scoping study developed https://www.horticulture.com.au/globalassets/hort-innovation/resource-assets/mt16004-cape-york-and-torres-strait-biosecurity-report.pdf The study includes the results of a tourism survey conducted during 2018 field surveys along the CYP
Community presentations	Community	2018 Meetings with schools (primary and secondary) in Weipa, NPA and TSI involving both AUSVEG and Cesar Australia researchers Involvement of Cesar Australia researchers in a career day held on Thursday Island, reaching about 300 students from across the Torres Strait Islands, years 7 to 12 2019 Meetings with schools (primary and secondary) in NPA and TSI involving both AUSVEG and Cesar Australia researchers Thursday Island Frog Gully Community Garden training activity/workshop involving AUSVEG and Cesar Australia researchers
Education tools for schools	Schools	Educational tools developed for schools and distributed during 2018 and 2019 school visits: PowerPoint presentations for secondary students, leafminer detection activity for primary students
Council meetings	Community	Meetings conducted in Lakeland, Cooktown, Weipa, Bamaga, and Thursday Island during 2018 field surveys
Research update letters*	Community	Summary of results (Figure 1 and Figure 2) obtained from TSI and NPA research activities distributed along with permission seeking letters to all relevant stakeholders before 2019 field trip
TSI and NPA parasitoid wasp guide	Community	Guides distributed during 2019 field activities, as well as via via Torres Shire Council, NPA regional council, Apudthama NPA rangers, Torres Strait Regional Authority, and the Northern Australia Quarantine Strategy networks https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#torres_strait_npa
TSI and NPA survey guide	Industry	Guides distributed during 2019 field activities, as well as via via Torres Shire Council, NPA

		regional council, Apudthama NPA rangers, Torres Strait Regional Authority, and the Northern Australia Quarantine Strategy networks https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/#torres_strait_npa
Surveillance training exercises for biosecurity officers and ranger groups	Industry	Mock surveillance training and data collection activities undertaken in Thursday Island (QLD) with Northern Australia Quarantine Strategy biosecurity officers, and in Cairns (QLD) with Torres Strait Regional Authority rangers, led by Cesar Australia researchers. Informational presentations delivered by AUSVEG with the PowerPoint file distributed to relevant groups.

Detecting exotic leafminer flies in the Torres Strait

The vegetable leafminer fly (VLM) is a garden pest found in the Torres Strait and NPA. They are very small black and yellow flies that create tunnels inside leaves (called leaf mines). The larval lifestage of the fly spends a week creating mines inside a leaf, then cuts an opening in the mine and crawls out to pupate in the soil.



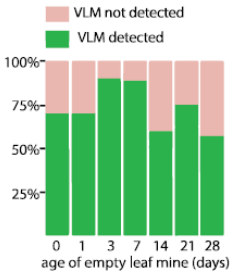


VLM populations grow quickly, so it is very important to quickly detect VLM if it spreads to a new area. Finding leaf mines is a good indicator that VLM is present, but most leaf mines we find are empty (the larva has already emerged), making it difficult to say what species of fly caused the damage - VLM, one of Australia's other native leafminer, or new exotic leafminer? We need a reliable way to tell the leafmines apart when we find them empty.

If we find an empty leafmine, can we tell if it was caused by VLM?

Leafminer larvae do leave something behind when they leave their leafmines: DNA, in the form of waste and skin. We wanted to know if this trace DNA could let us identify what species had created the leaf mine. In July 2018 we ran an experiment in the Frog Gully Community garden with the help of the North Australia Quarantine Strategy and the Torres Strait Regional Authority. We collected empty leaf mines between one day and one month after the larvae had emerged to see if we could still detect the DNA left behind.





We found that we can detect VLM, even in empty leafmines that were one month old, with a success rate of about 75%.

This is very exciting news for those who conduct surveillance for VLM as well as for other exotic leafminers (of which the Torres Strait is still free), since these exotic leafminer attack many of the same plants and create near identical leafmines. Using DNA to distinguish empty leaf mines can help us quickly determine if VLM has spread, or if a new exotic leafminer has arrived in the Strait.

This research was possible with support from the Northern Australia Quarantine Strategy, the Torres Strait Regional Authority, the Kaurareg Native Title Aboriginal Corporation, the Torres Shire Council, myPathways, the Apudthama Land Trust, Seisia Enterprises and the NPA Regional Council.

Thank you Harold Matthew and the TI NAQS team, and Ilana Foster and the TSRA team for your help running this experiment!



Hort Innovation

The strategic levy investment project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) is a part of the Hort Innovation Vegetable and Nursery Funds.

This project has been funded by Hort Innovation, using the vegetable and nursery research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

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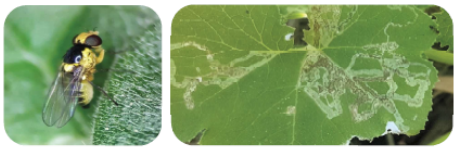
If you have any questions about our project, or about vegetable leafminer, please get in touch with:

Dr. Elia Pirtle, cesar
0414 143 456
epirtle@cesaraustralia.com

Figure 1. Summary of field work results relating to diagnostic technique experiments delivered to TSI and NPA stakeholders between 2018 and 2019 field trips

Searching for the veggie leafminer

The vegetable leafminer fly (VLM) is a garden pest found in the Torres Strait and NPA. They are very small black and yellow flies that create tunnels inside leaves (called leaf mines). The larval lifestage of the fly spends a week creating mines inside a leaf, then cuts an opening in the mine and crawls out to pupate in the soil.



It is very important to quickly detect this pest if it spreads to a new area. It can quickly grow to very large numbers if the wrong chemicals are sprayed. This means detecting the pest early is vital to making good management choices and preventing costly damage.

The Torres Strait community is helping us answer important questions about VLM, to build a surveillance plan based on science that the vegetable industry and the government can follow, to ensure we can catch the pest early if it spreads.

How long should you search an area, before you can be confident that there are no leaf miner flies present?

To answer this, we needed to know how good people usually are at spotting the leaf mines left behind by VLM. But because VLM is not present in growing areas, we couldn't have folks survey for real VLM leaf mines in crops. So instead, we have been hiding fake leaf mines in vegetable crops, and inviting growers and other vegetable industry workers to conduct quick surveys, to see how many of our fake leafmines are detected, and how long it takes to spot a leaf mine.



Searching for fake leaf mines in a spinach crop in Victoria

But we needed to make sure our fake mines are spotted at the same rate as the real thing. To test this, we conducted surveys on Thursday Island, where VLM is present. We invited biosecurity officers, rangers and community members on the island to survey for both real and fake leaf mines within the local Community Garden.



Searching for leaf mines in the Frog Gully Comunity garden

Thanks to these surveys, we discovered that most people will only notice about 30% of the leafmines that are hidden in a crop. **The data so far tells us that surveyors should spend about 1 minute searching each 1 meter of plants in a row, to have at least an 80% chance of spotting a hidden leaf mine.** We will continue to fine tune and test this recommendation, and then build them into a surveillance guide for VLM.

This research was possible with support from the Northern Australia Quarantine Strategy, the Torres Strait Regional Authority, the Kaurareg Native Title Aboriginal Corporation, the Torres Shire Council, myPathways, the Apudthama Land Trust, Seisia Enterprises and the NPA Regional Council.

Thank you to the TI NAQS team, the TI TSRA team, the myPathways garden team, the students of the Tagai State College and our other friends on Thursday Island who participated!

Hort Innovation

The strategic levy investment project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) is a part of the Hort Innovation Vegetable and Nursery Funds.

This project has been funded by Hort Innovation, using the vegetable and nursery research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

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If you have any questions about our project, or about vegetable leafminer, please get in touch with:

Dr. Elia Pirtle, cesar
0414 143 456
epirtle@cesaraustralia.com

Figure 2. Summary of field work results relating to surveillance technique experiments delivered to TSI and NPA stakeholders between 2018 and 2019 field trips

Appendix 9: Cape York & Torres Strait Biosecurity: a scoping study undertaken on behalf of the Australian vegetable and nursery & garden industries

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Dr. Jessica Lye [1]

[1] Cesar Australia

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Hort Innovation



MT16004: Cape York & Torres Strait Biosecurity

A scoping study undertaken on behalf of the Australian vegetable industry and nursery & garden industries.

Dr Jessica Lye, cesar

cesar

THE UNIVERSITY OF MELBOURNE

Plant Health AUSTRALIA

AUSVEG

Australian Government
Department of Agriculture and Water Resources

Overview

This report describes engagement and extension activities undertaken on the Cape York Peninsula and in the Torres Strait as a part of the Hort Innovation investment MT16004: *Preparedness RD&E for the vegetable leafminer*.

It includes assessment of emerging pest transmission pathway risks for Australian horticultural industries and suggests methods to support continuing engagement in the region for the purpose of reducing the risk of exotic pest spread to production regions.

Abbreviations

FNBZ 1 – Far Northern Biosecurity Zone 1

FNBZ 2 – Far Northern Biosecurity Zone 2

NAQS – Northern Australia Quarantine Strategy

NPA – Northern Peninsula Area

NPARC – Northern Peninsula Area Regional Council

TSRA – Torres Strait Regional Authority

TSIRC – Torres Strait Islands Regional Council

QDAFF – Queensland Department of Agriculture, Fisheries, and Forestry

VLM – Vegetable leafminer

Acknowledgements

This study was possible thanks to the time volunteered by individuals on the Cape York Peninsula and in the Torres Strait. Special thanks go to Mayor Eddie Newman, Councillor Joseph Elu, George Saveka, the Apadthama Rangers, Diana Russel, Eddie Sailor, Chris La Rosa, Aken Akee, Bobby Bimawel, Arthur Wong, Harrison Artu, Christo Lifu, Cook Shire Biosecurity officers, and Scott Templeton. Thanks also to Madeleine Quirk (AUSVEG) and Paul Umina (cesar) for input.

The strategic levy investment project *RD&E program for control, eradication and preparedness for Vegetable leafminer (MT16004)* is a part of the Hort Innovation Vegetable and Nursery Funds. This project has been funded by Hort Innovation, using the vegetable and nursery research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

EXECUTIVE SUMMARY 4

SCOPE AND ACTIVITY SUMMARY 5

REPORT SCOPE 5
ITINERARY 6
EXTENSION RESOURCES 8
EXTENSION AND ENGAGEMENT ACTIVITIES 9

REGIONAL ANALYSIS 15

ENTRY AND EXIT POINTS 15
CROP PRODUCTION 17
GOVERNMENTS AND AUTHORITIES 18
BIOSECURITY LEGISLATION 19
PLANT PEST THREATS 20
BIOSECURITY AUTHORITIES 21
REGIONAL BIOSECURITY STRATEGY 21
REGIONAL EXTENSION INITIATIVES 22
DEVELOPMENT GROUPS AND INITIATIVES 23
BIOSECURITY EDUCATION AND AWARENESS 26
FURTHER REGIONAL INSIGHTS 31
TRAVELLERS AND BIOSECURITY RISKS 32

TOURIST SURVEY 34

DEMOGRAPHICS 35
RESPONSE TO SURVEY QUESTIONS 36
SOURCES OF BIOSECURITY INFORMATION 40

DISCUSSION 42

RECOMMENDATIONS 44

REFERENCES 45

APPENDIX 1 47

Executive Summary

All Australian commercial production regions that grow vegetable leafminer (*Liriomyza sativae:* VLM) hosts, of which there have been over 300 identified, are at risk from this exotic pest and it is likely that growers in these regions will be required to actively manage it in the future. At the time of writing, VLM was contained to a small area on the mainland, at the town of Seisia on the Cape York Peninsula – a region that is well known as being at the frontline of biosecurity in Australia.

Cape York Peninsula and the Torres Strait region is a natural north-south pathway for exotic pests moving from South East Asia, with the vegetable leafminer being one example that has moved southbound along the Torres Strait islands and into the Cape. While the region is high risk for exotic pest incursions is also has high value as a buffer zone between Papua New Guinea and major horticultural regions in Australia.

The region is currently undergoing major changes. Tourism is a rapidly growing sector, an influx of funding is leading to major infrastructure initiatives, and regional biosecurity legislation has recently been updated.

This report details engagement and extension activities on the Cape York Peninsula and in the Torres Strait undertaken by AUSVEG, in collaboration with cesar, over 8 May-23 May 2018 and undertaken as a sub-component of MT16004.

This engagement visit allowed the project team to meet Cape York and Torres Strait locals, including community leaders, undertake initial discussions about the impact of exotic pests in the region, and learn about the biosecurity challenges and needs in these communities.

This report provides Hort Innovation, and associated levy payers with a snapshot of Cape York and Torres Strait biosecurity arrangements and provides insights into emerging biosecurity risks, such as increasing tourism in the region, that may have a future impact on horticultural production regions.

Finally, the report makes recommendations for investigating mutually beneficial collaborations between Australian horticultural industries and far northern communities and initiatives, for the purpose of supporting good biosecurity practices, and encouraging pest reporting in Cape York and the Torres Strait.

Scope and Activity Summary

Report scope

At a project level, MT16004 (Preparedness RD&E for the vegetable leafminer) aims to raise awareness and management knowledge of vegetable leafminer (VLM) among vegetable and nursery growers to aid smooth and fast adoption of best management practices for control of this new pest. Cape York Peninsula and Torres Strait communities are either already observing damage caused by VLM or are in close proximity to affected regions. To limit its impact on already affected communities and slow its spread further south the MT16004 project team was tasked with raising awareness and management knowledge of VLM among these communities.

This MT16004 project component involved conducting one engagement mission to Seisia (Cape York Peninsula) in year 2 of the project, as well as deliver training in VLM control based on project R&D outcomes in year 3 of the project. In order to ensure legacy of engagement mission findings, findings from the year 2 trip were included in this report to Hort Innovation for the benefit of the Nursery & Garden Industry, and the Vegetable Industry. Readers of the report will notice that it contains background on the region – this is information that the author deemed important to include, in order to appropriately contextualise findings and recommendations.

This report endeavours to answer the following questions:

1. What are the major pathways that may facilitate exotic plant pest transmission south into production regions?
2. What are the emerging risks that may impact on maintaining biosecurity in the region?
3. What are the current biosecurity initiatives and organisations in the region, and is there an opportunity for Australian horticultural industries to collaborate on biosecurity activities?
4. What are the current plant pest best management practices (BMPs) used in Torres Strait and Cape York communities? Is there an opportunity for Australian horticulture professionals to provide training in BMPs for plant pests?

To this end, engagement mission activities included:

1. Undertaking extension activities to raise awareness about VLM in the region with communities – particularly with councils, schools, and business owners in the tourism sector;
2. Meeting community leaders and horticultural personnel in Cape York and the Torres Strait to develop relationships, brief them on the risk posed by VLM, and learn about current biosecurity initiatives;
3. Undertaking extension activities to raise awareness about VLM in the region with tourists; and
4. Surveying tourists to better understand the biosecurity risk posed by travellers to the Torres Strait.

Engagement and extension work undertaken by the MT16004 project team has laid foundations for building collaborative biosecurity relationships between this important region and production industry stakeholders. This report aims to inform and guide future Nursery & Garden Industry, and Vegetable Industry engagement in Cape York Peninsula and the Torres Strait.

Itinerary

Between 8 May and 21 May 2018 Dr Jessica Lye (AUSVEG) and Dr Elia Pirtle (cesar) travelled from Cairns, QLD, to Thursday Island in the Torres Strait (Table 1). The journey was undertaken by car and ferry, to allow VLM host plants to be surveyed for the presence of the pest during the journey up Cape York Peninsula and on Torres Strait Islands. The region map, with the route and islands visited for VLM surveillance are displayed in Figure 1.

Table 1. Engagement mission itinerary

Dates	Locations
8–10 May	Cairns, Lakeland, Cooktown, Coen, Weipa, Bramwell Junction.
11–14 May	New Mapoon, Injinoo, Seisia, Roko Island, Albany Island, Zuna Island.
15–17 May	Thursday Island and Horn Island.
18–21 May	Seisia and Loyalty Beach (cesar researcher remained in the Torres Strait for further surveillance activities)

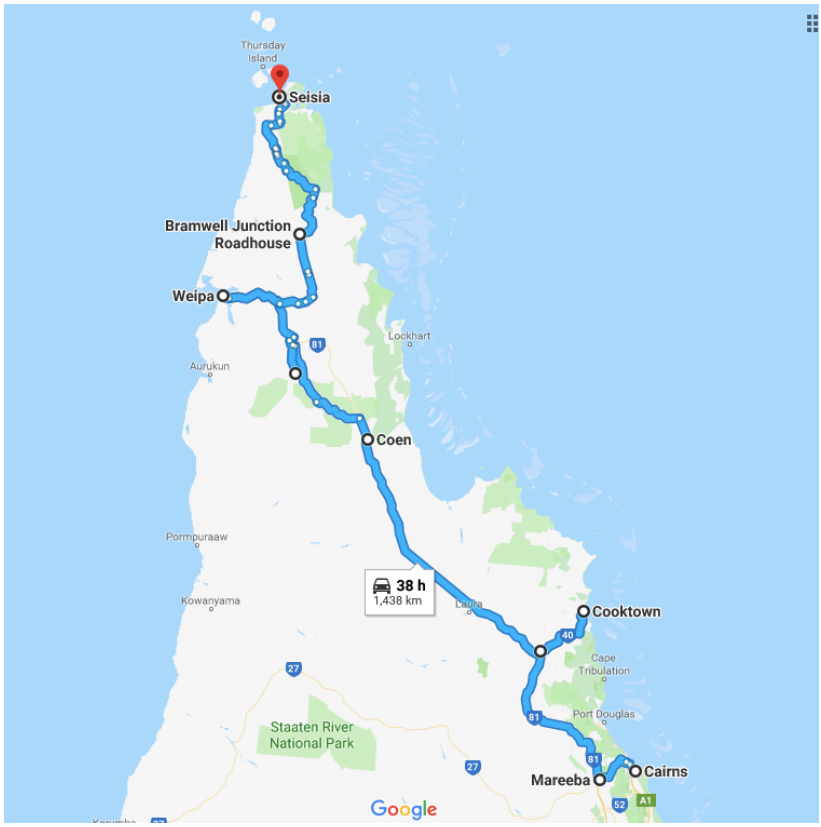




Figure 1. Route followed from Cairns to Seisia (above) and islands surveyed in the Torres Strait for VLM (below, yellow pins), and Thursday Island in the Prince of Wales island group (below, top left of map)

Extension resources

During the trip the project team conducted a number of VLM awareness raising activities with business owners, schools, councils, and tourists throughout Cape York Peninsula. Several resources had been developed for use during the trip to aid extension activities. They are described below.

- Laminated information stakes were staked into communal gardens at campsites, schools, and in shared spaces within the community in regions where VLM had not been detected. The information stake describes the symptoms of VLM infestation and encourages reporting for the purpose of tracking VLM spread (Figure 2).
- Project flyers and VLM awareness flyers were handed out during briefings, as well as laminated A3 awareness posters.
- A school colouring handout, which educates children about VLM and what it likes to eat was supplied to the Cape York Peninsula Biosecurity Centre, and schools in Weipa, Thursday Island, Horn Island, and Bamaga.
- A 'mock surveillance' activity, with guidelines for use, was handed out at schools visited in the Cape, and the activity was undertaken at one school (Figure 2).



Figure 2. Extension resources used during the trip – information stake (top left), tourist survey information sheet (top right), mock surveillance materials (bottom)

Extension and engagement activities

Table 2. Meetings, contacts made, insights, and extension activities (including follow up activities) undertaken during this engagement mission

Day & location	Activity	Connections	Insights	Follow up activities
Day 1: **Cairns & Lakeland**	○ VLM presentation to Lakeland Progress Association. ○ Supplied association with poster and fliers.	Lakeland Progress Association: Stephanie O'Kane (Executive Officer, Lakeland Progress Association), Martin Garadi (Grower), Martin Arnold (Grower), Shaun Jackson (Grower), John Lorenzo (Grower).	Approx. 10 commercial growers in the area, growing a mix of watermelon (often for export), eggplant, passionfruit, cucumber, and banana. The Lakeland washdown station was recently decommissioned due to costs to keep it running. There are plans to build a tourist information centre in Lakeland although there is no timeframe for this.	AUSVEG sent the association a project article for use in their newsletter.
Day 2: Cooktown and Lakeland	○ VLM briefing for Cooktown biosecurity officers. ○ VLM briefing for Cape York NRM extension officer. ○ Lakeland grower visits – surveillance and briefings. ○ Lakeland primary school visit and briefing for principal. ○ VLM briefing to owner of Lakeland Tourist Park.	Cook Shire Council: Cathy Johnson (Senior Biosecurity Officer) and Darryn Higgins (Biosecurity Officer). Prior connection had been made with Dean Schreike (Biosecurity Officer). Cape York NRM: Natasha Reid (Industry Development). Prior conversations had been	Cooktown area has many small hobbyist growers, mainly organic, and they sell each Saturday at the market. In future, if anyone from the project comes to visit again they should attend the market to speak to the growers. None of the growers or other community members we spoke to had heard of VLM. The Lakeland primary school principal was	AUSVEG sent the VLM activity pack in e-copy to the primary school. AUSVEG sent Cook Shire Council a VLM weed host list so they may keep an eye out for damage while controlling weeds.

	○ Supplied all visited with posters and fliers. ○ Supplied tourist park and primary school with an awareness stake for their gardens. ○ Supplied school with VLM activity pack.	conducted with Michael Goddard (Gully and Grazing Project Officer), and Will Higham (Program Manager). Lakeland Primary School: Lauren O'Malley (Principal)	highly supportive, since most children at the school have parents involved in growing.	AUSVEG sent Cape York NRM e-copies of the project flyers, and a VLM PPT for use during grower meetings.
Day 3: Coen and Weipa	○ VLM briefing for Cape York Peninsula Biosecurity Centre Manager. ○ VLM briefing for Outback Spirit tour operators in training. ○ VLM briefing for Weipa caravan park owners. ○ VLM briefing for Weipa Town Authority. ○ Supplied all new connections with poster and fliers.	Cape York Peninsula Biosecurity Centre: Scott Templeton (Biosecurity Compliance, QDAF) Weipa Town Authority: Judey Brown (Coordinator, Communities), Michael Rowland (Chair)	The Inspection Centre used to be at Laura, then it moved to Coen. Since that time the car counter has shown a significant increase in travellers along the Peninsula Developmental Road. Weipa Town Authority and the Weipa caravan park owners were unaware of VLM but were happy to help distribute awareness material. The project team was invited to submit an article for the regional newspaper via the Weipa Town Authority.	AUSVEG sent the VLM activity pack in e-copy to the primary school. AUSVEG sent e-copies of flyers to WTA and an article for publication. AUSVEG sent a VLM article to the Cape York news for publication.
Day 4: Weipa	○ VLM briefing for Rio Tinto Land Rehabilitation team. ○ Canvassing of tourists at the Weipa Caravan Park. ○ VLM activity pack supplied to St Joseph's primary school. ○ Visit to grade 2 class at Western Cape College primary campus.	Rio Tinto: Harry Nevard (Land Rehabilitation Coordinator) Western Cape Primary: Samantha Blocksidge (Head of Curriculum)	Rio Tinto have a weed survey team and they keep a local weed list. Home owners had not heard of VLM. Pest control in Weipa home gardens is heavily organic and includes soap, detergent, neem oil, and milk.	*Western Cape College Primary subsequently wrote an article in the school newsletter about VLM and this visit.

	○ Students taken through the VLM colouring activity and mock surveillance activity. ○ VLM briefing for homeowners during surveillance visits at Weipa port.		Running a leafminer activity session at Western Cape College Primary School integrated well with the second-year course syllabus, in which they had been learning about insect lifecycles.	
Day 5: Weipa and Bramwell Junction	○ Canvassing of tourists at the Weipa Caravan Park. ○ Project fliers and an information stake were supplied to the owners of Weipa caravan park. ○ VLM briefing for manager of Moreton Telegraph Station. ○ VLM briefing for manager at Bramwell Station. ○ Each station was supplied with poster, fliers and information stakes.	Bramwell Station and Tourist Park: Ken Godfrey (Manager)	Most tourists canvassed at Weipa Caravan Park had not heard of vegetable leafminer but were interested to learn about it. All tourists canvassed were unaware of the updated Cape York Peninsula biosecurity zoning. Station employees were unaware of the vegetable leafminer but were willing to put awareness collateral on display.	
Day 6: Bramwell Junction and Seisia	○ Canvassing of tourists at Bramwell Station. ○ Bramwell Roadhouse supplied with poster, fliers and information stakes.		On the road to the Jardine Ferry crossing there was one biosecurity sign, approximately 1 km south of the crossing. It showed the old quarantine zone, denoting the zone as one single zone from Coen north to the tip. The river crossing personnel are currently not trained in biosecurity and have no powers under the act. There is a popular tourist destination, Fruit Bat Falls, which may be used as a good tourist information location in future.	

Day 7: Seisia, Bamaga and Injinoo	○ Meeting with Mayor Eddie Newman to discuss biosecurity challenges in the region. ○ Visit to NPA College where a presentation was made to the Year 7 agri-science students. ○ VLM briefing for Land and Sea Rangers (Injinoo). ○ Canvassing of tourists at Seisia Tourist Park. ○ All new connections supplied with posters fliers, and information stakes.	NPA State College: Harrison Artu (Farm Technical Officer) Seisia Enterprises: Arthur Wong (Owner) NPARC-Apudthama Rangers: Erra Bond (Operations Co-Coordinator) NPARC-Apudthama Rangers: Christo Lifu (Operations Co-Coordinator)	The NPARC is becoming very aware of biosecurity and the role the Jardine Ferry crossing may play in controlling traffic. The NPA has experienced incursions from the south, such as the expansion of the cane toad range. Gamba grass, which has spread north into the NPA, is also a problem pest and increases fire risk (see 'Further insights' section for more information). The NPA is home to a high number of free roaming horses, which could play a role in pest spread around the NPA. Many residents in the NPA keep chillies (a VLM host) in their gardens. Pest management practices in home gardens are largely organic. Roads leading north into the NPA from Coen are largely unsealed at the time of writing (see 'Entry and exit points' section for more information). NPA State College were very happy to receive a visit and it was suggested that the project team visit the college in the future to update the students and staff on project findings. The college NPA State College farm would be an ideal location for future VLM management workshops.
Day 8: Roko, Albany and Zuna Islands	○ VLM surveillance on islands. ○ Briefed island inhabitants. about identification of VLM ○ Island inhabitants given. contact information for the project team in case they observed any signs of VLM.		There was only one residence on each island surveyed. Inhabitants on these islands were interested to learn about VLM. One couple on Zuna island (where a putative VLM detection had been made on that visit) mentioning that their tomato plants had died out the previous year due to whitefly and leafminer.

			The couple on Zuna island was asked to refrain from transporting host material to the mainland.	
Day 9: Horn Island	○ VLM briefing for acting Principal at Tagai College Horn Island Primary Campus. ○ Supplied the campus with a pest UTE guide for northern pests, project poster and flier.	Tagai State College, Horn Island: Matthew Deady (Principle).	Horn Island Primary School Campus has a 'Harmony Garden', in which a diversity of VLM hosts had been planted: pumpkin, cucumber, tomato, marigolds. The garden displayed heavy VLM infestation and the Principal, who had not heard of VLM, was informed of this. This garden would be an ideal location for future VLM management workshops.	AUSVEG sent the VLM activity pack in e-copy to the primary school.
Day 10: Thursday Island	○ Visit to Tagai College, secondary campus and presentation to the students on exotic pests, biosecurity, and VLM. ○ VLM briefing for Torres Strait Island Regional Council (TSIRC) rangers.	TSIRC: Aken Akee (Environment Officer), Diana Russell (Town Planner), Bobby Bimawel (Pest Officer), Steven Sailor (Pest Officer) Tagai State College: Tim Hillier (Head of Department Land and Sea Management), Chris La Rosa (Secondary Biology Teacher)	The cane toad has become a major problem on Thursday Island, with a toad collecting competition being held every Friday by TSIRC. Several local connections made during the visit to Thursday Island mentioned that tomato and chilli plants in their gardens had been severely impacted by VLM (VLM damage was confirmed in at least one reported case). TSIRC officers were shown minute parasitoid wasps under the microscope to allay concerns about 'wasps' being advised for use in VLM control.	

Day 11: Thursday Island	○ VLM briefing for Torres Strait Regional Authority (TSRA)	TSRA: Mark Blair (Senior NRM Officer), George Savioka (Industry Development Officer)	TSRA runs a Sustainable Horticulture project throughout the Torres Strait (see section 'Regional extension initiatives' section for more information).	AUSVEG sent a VLM article to the Torres Strait newspaper for publication.
Day 12 and 13: Seisia and Loyalty Beach	○ Canvassing of tourists at Seisia Holiday Park and Loyalty Beach Campground and Fishing Lodge. ○ Briefing given to the managers of both tourist parks. ○ Fliers and information stakes were supplied at both parks.	Loyalty Beach Campground and Fishing Lodge: Pat Lennox (Owner)	Tourists and park managers were unaware of VLM but were very interested to learn about it. New VLM infestation found at the Seisia Tourists Park, in close proximity to where tourists were 'sunning' their basil plants (see 'Tourist survey' section for more information).	

Regional Analysis

Cape York Peninsula is the northern-most part of Australia, with a land area of approximately about 220,000 square km. It is made up of a number of regions, including Western Cape York (Weipa, Mapoon, Napranum & Aurukun), Eastern Cape York (Iron Range), Central Cape York (Hann River Roadhouse, Musgrave Roadhouse, Coen, Archer River, Moreton Telegraph Station, Bramwell Tourist Park, Bramwell Junction), and Lower Cape York (Bloomfield Track, Bloomfield, Wujal Wujal, Cooktown, Hopevale, Laura, Lakeland and Palmer River).

The peninsula is transected by the Jardine River, with the area north of the Jardine being surrounded by the Coral Sea to the east, the Gulf of Carpentaria and Arafura Sea to the west, and the Torres Strait (and islands) to the north.

The regional climate is tropical, monsoonal with a wet and dry season. Most rainfall can be expected between November and April, with average rainfall being 1700mm, and falling as heavy thunderstorms, monsoonal lows or tropical cyclones. Temperatures remain warm (18-30°C in the dry and 24-36°C during the wet).

Cape York Peninsula and the Torres Strait are perceived as being remote wildernesses with a high cultural and environmental value. The region is sparsely populated, with 7,513 people in Cape York and 4,514 people in the Torres Strait islands. However, this number increases substantially each dry season with influxes of tourists.

Rio Tinto operates the largest bauxite mine in the world out of Weipa. Therefore, a large portion of the land around Weipa is leased by Rio Tinto. Cattle stations are another major industry on the Cape, with the largest station being located at Bramwell.

Entry and exit points

The transport infrastructure servicing the region's remote communities is served by a primary north-south road access route, local access roads and air and marine facilities of various scales. There is a significant reliance on air and sea transport, particularly during the wet season when road closures are common.

There is a daily plane service (Qantas Link) between Cairns to Weipa, as well as a Qantas Link service between Horn Island to Cairns, with Horn Island being the only island within the Prince of Wales Island group to be flat enough to host an airstrip. There is also an airport serviced by SkyTrans in Coen, into which the Royal Flying Doctors service arrives each week, and an airport at Bamaga from which the Regional Express runs a regular passenger flight to Cairns.

While those listed above are the main methods of accessing or leaving the region by air, additional airports and landing strips are located at Aurukun Airport, Cooktown Airport, Pormpuraaw Airport, Kowanyama Airport, Lizard Island Airport, Lockhart River Airport, Northern Peninsula Airport, Weipa Airport, and RAAF Base Scherger.

After flying to Horn Island arrivees can then catch the MacDonalds water taxi to Thursday Island, the main tourism destination in the Torres Strait. Peddles Ferry is then the main method

of travelling from Thursday Island to Seisia on Cape York Peninsula. Many inhabited islands in Torres Strait also have airstrips suitable for light aircraft which can be accessed through regular passenger transport or charter flights.

The major point of entry by land is via the Mulligan Highway (228 km), which runs from Mareeba to Lakeland, and the Peninsula Developmental Road (524 km), which runs from Lakeland to Weipa.

Roads are sealed until Lakeland, after which there are sections of sealed and unsealed road until Coen. West and North of Coen is largely unsealed road that becomes rutted from increased road traffic over the course of the dry season and can be unusable during the wet season due to flooding.

There are several plans that outline intentions for improving road infrastructure on the Cape York Peninsula, which will in turn support economic growth, more tourism, and transport of produce. These are:

- Far North Queensland Regional Action Plan
- Cape York and Torres Strait Transport Infrastructure Plan
- Peninsula Developmental Road Investment Plan

Maintenance of highways on Cape York Peninsula is split between several local councils, which means that plans to seal the entire Peninsula-Development Road may progress disparately over the next few years.

Entry into Cape York Peninsula can also be gained by sea. There are five ports in the region, four of which are managed by the Far North Queensland Ports Corporation. The fifth, the Port of Weipa, is operated by North Queensland Bulk Ports Corporation. The Sea Swift barge is a major sea transporter of fresh produce from Cairns to the Cape York Peninsula and Torres Strait.

With a number tourism and infrastructure plans having been developed for the region in recent years, it is clear that increasing accessibility to the Cape is a focus for the state government and local governments.

Surveillance at entry points (Sea Swift Docks)

This trip involved VLM surveillance around the Sea Swift docks at Seisia, Weipa, Thursday Island, and Horn Island. Siratro (a VLM host plant) grows prolifically on fences surrounding each dock, which may aid establishment of the fly should it be transported on Sea Swift produce and blown to shore.

Crop production

The Lakeland region is the most northern commercial horticultural region in Australia and is classified as a priority agricultural area by the Queensland Government. Major crops are banana, sorghum, peanuts, with some pineapple, watermelon, and mixed solanaceous crops at a smaller scale also grown.

As a cropping region the Lakeland area is relatively free of many plant diseases found elsewhere in the state and is seen as a low risk option for growing high quality produce. Further, due to its placement on the intersection of the Mulligan Highway and the beginning of the Peninsula Developmental Road, Lakeland is the 'tourism gateway' to the Cape for those visitors travelling by road. The town is also experiencing a growing seasonal population of transient farm workers (mainly backpackers).

In 2018, the Lakeland Progress Association reported that there are ten horticultural producers in the area in close proximity to the town (Stephanie O'Kane, pers comms). The Lakeland Local Area Plan (2017) suggests that there is strong potential for the expansion of the local agricultural industry if further water for irrigation can be accessed, with investigations into water resource options underway.

Therefore, a likely future scenario is an increase in pest transmission risk (from north or south) due to an expansion of mixed horticultural cropping in Lakeland, larger influxes of transient farm workers, and greater numbers of tourists passing through the town on the way to or from Weipa or the Northern Peninsula Area (NPA) and Torres Strait.

Other horticultural production elsewhere on Cape York Peninsula and in the Torres Strait is small scale hobby farm, community garden, or backyard garden production with minimal pest control measures practiced - particularly pesticide based control. The preference towards organic methods of growing was noted several times during meetings with locals over the course of the trip.

According to senior biosecurity staff at Cook Shire Council, the Cooktown area has many small hobbyist growers, mainly organic, and they sell each Saturday at the market. The method of growing is predominantly organic. Feedback from several backyard gardeners in Weipa, and consultation with the Mayor of the Northern Peninsula Area Regional Council (NPARC) also confirmed that horticultural growing practices in much of the Cape was organic, with control methods involving milk, detergent mixes, or spraying of chili or garlic mixtures.

Governments and authorities

Local government areas cover Aurukun, Cook Shire, Hope Vale, Kowanyama, Lockhart River, Mapoon, Napranum, Northern Peninsula Area, Pormpuraaw, Wujal Wujal and Weipa Town Area. The local governments and authorities of Cape York Peninsula and Torres Strait identified during this trip are listed below and shown in Figure 3.

Cook Shire Regional Council

Aurukun Aboriginal Shire Council
Hope Vale Aboriginal Shire Council
Kowanyama Aboriginal Shire Council
Lockhart River Aboriginal Shire Council
Mapoon Aboriginal Shire Council
Napranum Aboriginal Shire Council
Pormpuraaw Aboriginal Shire Council
Wujal Wujal Aboriginal Shire Council

Northern Peninsula Area Regional Council
Torres Strait Island Regional Council
Torres Shire Council
Weipa Town Authority
Torres Strait Regional Authority
Torres Cape Indigenous Council Alliance

While this list covers the majority of authorities in the region it must be noted here that there are multiple and overlapping Aboriginal entities for the traditional ownership and management of lands and waters, such as the Cape York Land Council. Many parts of Cape York Peninsula and the Torres Strait are allocated Native Title, which can include pastoral leases, statutory Aboriginal freeholds and trustee arrangements (Memmott and Blackwood, 2008).

Liaising with governing bodies of native titles

The governing bodies of native titles should be considered, contact made, and permissions sought in the case of any biosecurity activities launched on the part of the Australian horticulture industry.

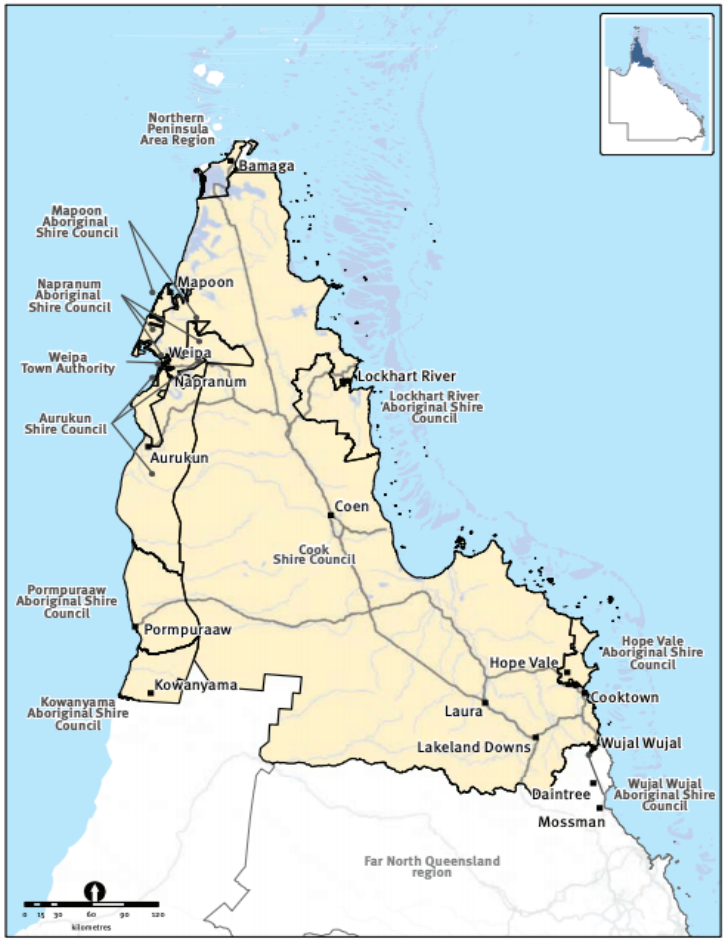


Figure 3. Local governments and authorities in Cape York Peninsula (Excludes Torres Strait authorities) (Source: Cape York Regional Plan, 2014).

Biosecurity legislation

Under the *Plant Protection Act 1989* and *Plant Protection Regulation 2001*, the area of Cape York North of Coen was gazetted as the Far Northern Pest Quarantine Area and the Cape York Peninsula Targeted Pest Quarantine Area. These legislative boundaries were repealed in 2016 and were replaced by new legislation under the *Biosecurity Act 2014* and *Biosecurity Regulation 2016*.

Under the Queensland *Biosecurity Act 2014* the region is now split into Far North Biosecurity Zone 1 (FNBZ 1), which begins at latitude 11 degrees 49 minutes south near Heathlands and the Jardine ferry crossing, and Far North Biosecurity Zone 2 (FNBZ 2), which begins at latitude 13 degrees 45 minutes south, which is north of Coen at the Cape York Peninsula Biosecurity Centre. FNBZ 1 spans the five townships of the Northern Peninsula Area as well as the Torres Strait.

The Torres Strait is further sectioned into the Torres Strait Protected Zone, which encompasses the region north of Mawai Island to Papua New Guinea, and the Torres Strait Permanent

Biosecurity Monitoring Zone, which encompasses islands between Cape York Peninsula and Mawai Island. These additional zones are legislated under the federal *Biosecurity Act 2015.* A treaty allows movement of people and traditional vessels between 13 Papua New Guinea townships and islands within the Torres Strait Protected Zone without the need to undergo entry requirements, however biosecurity compliance checks of vessels and goods are undertaken.

According to the Queensland Biosecurity Regulation 2016 plant material cannot move out of FNBZ 1 or south out of FNBZ 2 without a permit. Further, federal legislation specifies that plant material, soil and other risk materials (such as animals, dairy products, fresh vegetables, and untreated hides) cannot move from the Torres Strait Protected Zone to the Torres Strait Permanent Biosecurity Monitoring Zone, or from either of these zones into mainland Australia without a permit. There are currently no specific legislative instruments in place to restrict movement of plant material north into Cape York Peninsula from mainland Australia.

By necessity, the regulations that dictate movement of biosecurity risk materials between those zones are fairly complex. Even with the aid of handouts and flyers developed by the Australian Department of Agriculture and Water Resources and the Queensland Department of Agriculture and Fisheries, which can be picked up biosecurity offices and tourist hubs throughout the Cape and the Torres Strait, it is easy to understand why these regulations may become confusing for tourists.

Plant pest threats

Some high-profile horticultural Emergency Plant Pests that have been identified as a risk of spreading south through the Torres Strait and NPA include the Asian honey bee, banana skipper butterfly, exotic fruit flies, mango pulp weevil, red banded mango caterpillar, sugarcane borers, coffee mealy bug, and vegetable leafminer. A variety of exotic weeds, plant diseases and animal diseases are also described in regional awareness materials (see '*biosecurity education and awareness*' section for more information).

The vegetable leafminer had been surveyed for extensively at the time of writing, by the Queensland Department of Agriculture and Fisheries, the Northern Australia Quarantine Strategy, and more recently by the MT16004 project team. Based on surveillance results throughout the Cape and Torres Strait, the leafminer remains restricted to Seisia on the mainland, although a new detection of VLM was made at the Seisia Tourist Park during this trip, and a suspect detection was made on Zuna Island.

Additional *Liriomyza* sp. threats from the North

There are now two more exotic leafminers that have been detected within close proximity of the Torres Strait. These are the pea leafminer and serpentine leafminer.

Biosecurity authorities

The Department of Agriculture and Fisheries (Queensland State Government) and the Department of Agriculture and Water Resources (Federal Government) have a strong biosecurity footprint in the region, as regulators, educators, and surveyors of exotic pests. Inspection of risk materials moving southward through the Cape is carried out at the Cape York Peninsula Biosecurity Centre (formerly the Coen Information and Inspection Centre), which is managed by the Queensland State Government.

The Northern Australia Quarantine Strategy (NAQS) is a biosecurity program managed by the Federal Government. It was established in 1989 and is responsible for surveillance of exotic pests across northern Australia. It also manages biosecurity arrangements for traditional visitors from Papua New Guinea to the Torres Strait islands, and southward movement of people and products between the special Torres Strait biosecurity zones. It also plays a strong role in raising public awareness (see the 'Regional extension initiatives' section for more information).

A biosecurity working group comprised of representatives from DAF, the Australian Department of Agriculture and Water Resources (DAWR), Queensland Health, Australian Department of Environment and Energy, Torres Shire Council, Northern Peninsula Area Regional Council, Torres Strait Regional Authority, and Torres Strait Island Regional Council, has recently developed the 'Torres Strait and Northern Peninsula Area Regional Biosecurity Strategy'. The Strategy provides a framework and an ethos for how all regional stakeholders, including three levels of government, traditional owners, local residents and visitors to the region can better manage biosecurity threats.

Following development of the strategy, the Queensland State Government announced additional funding for a Far Northern Biosecurity Initiative of $1.7 million over three years which will help it deliver on its commitment to the strategy and bolster biosecurity capacity in the region. The intention of the additional funds is to support a range of new biosecurity measures in the region, including indigenous traineeships, extended operating hours for the Cape York Peninsula Biosecurity Centre, development of new awareness materials, and new engagement initiatives with local communities.

Regional biosecurity strategy

Developed by the Torres Shire Council, NPARC, TSRA and TSIRC, and the Queensland and Federal governments, this strategy was formally endorsed on 25 September 2018.

It was developed under the auspices of the 'Northern Australian Agricultural Competitiveness White Paper' in recognition of the "*unique challenges and opportunities in the management of biosecurity risks in Torres Strait and the Northern Peninsula Area of Queensland*" (TS & NPA Strategy, 2018).

Vision of the strategy

"We will support sustainable communities, protect the unique and pristine environments within our region, and play our part in preventing the movement of damaging pests and diseases, through practical, integrated and culturally appropriate biosecurity risk management."

As detailed by the strategy, its vision will be achieved by focussing on six priority areas:

1. Collaborative partnerships (encouraging community participation, regional coordination of biosecurity management, and use of local knowledge)
2. Understanding threats (pathway analyses, early reporting, data capture, pest assessments)
3. Making biosecurity more meaningful to communities (who need to see benefits from biosecurity and be engaged in addressing local biosecurity problems)
4. Communication and awareness (updating biosecurity materials in the region, use of biosecurity ambassadors and other communication channels)
5. Improved response (local partnerships, and tailored response plans for the region)
6. Improved capability and capacity (training, contingency planning, arrangements with local groups, such as the rangers)

Therefore, any collaborative biosecurity activities undertaken by Australian horticultural industries should keep this regional strategy, and its priority areas, in mind.

Regional extension initiatives

Major biosecurity awareness work in the Cape and Torres Strait is carried out by the Queensland Department of Agriculture and Fisheries, notably through the Cape York Peninsula Biosecurity Centre (described in the 'Biosecurity education and awareness' section of this report) and the Indigenous Land and Sea Ranger Program, and the Federal Department of Agriculture and Water Resources through the NAQS community engagement program 'Top Watch', which was established in 1992.

The Federal Government undertook an internal review of NAQS awareness activities in 2010, during which it developed the program's first communication strategy. Top Watch activities are directed at residents in the Torres Strait special biosecurity zones and the program encourages these stakeholders to keep a 'top watch' and report suspect pests. A key feature of this awareness program is the employment of local ranger staff to promote the Top Watch messages. On an annual basis funding of NAQS outreach is small compared with funding for surveillance, and awareness activities are often integrated into other NAQS activities.

According to a 2014 Federal Government brochure that featured NAQS initiatives and employees: "*It provides a vehicle for establishing strong working relationships with local communities, facilitating surveillance activity on privately owned land and improving*

compliance with legislation on the movement of prohibited items through the Torres Strait risk pathway."

Those engaged through NAQS Top Watch outreach activities have been predominantly locals rather than tourists (although NAQS does supply information materials to travellers, particularly during community events).

Recent outreach has also been conducted by Costa the Garden Gnome and Dirt Girl. They were enlisted Biosecurity Champions for Cape York Peninsula and the Torres Strait by the Department of Agriculture and Water Resources in 2017 with the intent of raising biosecurity awareness among travellers. Costa and Dirt Girl took part in developing a biosecurity awareness video 'Biosecurity in the Torres Strait is Everyone's Business'. The video has received 474 views on YouTube since its publication on 9 April 2018.

Development groups and initiatives

Community and natural resource development groups that actively participate in, or run activities that would align with, or support, further biosecurity awareness include:

- The Cape York Natural Resource Management group
- The Lakeland Progress Association
- The Junior Ranger Program (run through the Indigenous Land and Sea Ranger Program)
- The TSRA 'Healthy Communities' and 'Environmental' programs
- Landcare Australia
- My Pathways
- The North Australian Indigenous Land and Sea Management Alliance

Several initiatives that are currently run by these groups are expanded on below.

Mekem Garden Sustainable Horticulture Project

The TSRA Environmental Program, with support from its Healthy Communities program, has a component that aims to promote market garden activities through the 'Mekem Garden Sustainable Horticulture Project'. This project delivers food production training throughout the Torres Strait and encourages communities to access TSRA grant funding to aid them in developing and maintaining their own community gardens. This initiative is also coordinated in conjunction with the Regional Landcare Facilitator role and receives funding from the Australian Government's 'Caring for our Country' Program.

This trip included a meeting with George Saveka, the Regional Landcare Facilitator with the TSRA Land and Sea Management Unit, and main officer working on the Sustainable Horticulture project. The project aims to achieve the following:

- Facilitate workshops on outer islands to provide practical and technical demonstrations on food gardening.
- Provide guidance and assistance in the establishment of community gardens in two communities.

- Provide guidance and assistance in the development and maintenance of plant nurseries.
- Increase awareness of sustainable horticulture throughout the region through a monthly newspaper garden column and island visits.

Mr Saveka travels regularly between the inhabited islands of the outer Torres Strait, training communities in growing practices. It was noted by Mr Saveka that Torres Strait island soils are quite diverse and providing advice for growing on these diverse soils can be difficult.

According to the TSRIC the islands can be divided into four main groups: an eastern group of high volcanic islands, a central group of low sandy islands, a western group of high islands composed of volcanic and granitic rocks, and a northern group of low islands composed of mangrove muds and peats. During the meeting with Mayor Newman of NPARC it was also pointed out that there are soil issues in the Cape. It is very acidic, porous and there have been reports of nematode issues. Combined with the climate this makes it difficult to grow vegetables.

A land of complex growing conditions

The soil profile of the Cape and Torres Strait is complex. The western region of the cape, which is a large river plain, is composed of laterite, a mineralised soil of low fertility, typically overlain by hard ferrous deposits. Beneath them the pale coloured, soft material often contains bauxite. While visiting Seisia it was mentioned that the soil was quite acidic.

Importantly, this project provides better access to fresh and nutritious fruits and vegetables. As noted by TSRA Chairperson, Mr Toshie Kris in one information flyer about the sustainable horticulture project:

> *"Our Elders taught us how to grow and use different plant species, and now it's our children's turn. Except today they have the added advantage of combining traditional knowledge with contemporary horticultural practices."*

It was also noted by Mr Saveka that there is a need to educate communities about the nutritional content of fresh produce, particularly vegetables, and the health benefits associated with growing this produce. The cost of fresh produce is higher than average (Appendix 1), and it is possible that more NPA and Torres Strait communities will endeavour to start their own market and community gardens. Therefore, there is potential for additional surveillance and best practice pest management should a partnership be formed with the TSRA sustainable horticulture project.

Fresh produce can be costly in the NPA and Torres Strait

The cost of fresh produce was recorded at one Cape York retail outlet during the trip, and it was observed that the cost of many vegetables was above average, with the costlier items being broccoli at $5.99 a head and snow peas at $33.00/kg.

Thoughtful travelling Cape York campaign

The Cape York Natural Resource Management group coordinates the 'Thoughtful Travelling Cape York' campaign, which seeks to encourage visitors to leave Cape York campsites better than they find it. Their Cape York Keep it Clean guide contains information on camping best practices, location of rubbish bins on the Cape, managing fire risks, cultural considerations, and notably, reducing the risk of transferring weed seeds around and out of the Cape. Multiple collaborating organisations are involved in this initiative and there may be opportunity to supply further information on biosecurity best practices and on key horticultural pest threats.

Gateway to the Cape information centre

The Cook Shire Council has developed plans for a new information centre to be built at Lakeland, adjacent the Mulligan Highway and Peninsula Developmental Road intersection at the current location of the wash down station (which is now decommissioned with plans for full removal). This 'Gateway to the Cape' Tourist Information Centre is intended to become a regular stopping point for travellers and will provide a useful opportunity for educating visitors before they enter the region.

Therefore, this future education centre provides an opportunity for awareness to be raised about key horticultural pests as most tourists on the Cape access and leave the region by road. Further information about information centre plans can be found in the Lakeland Local Area Plan Background Report (2017) or sourced directly from the Lakeland Progress Association.

Lakeland washdown station a point of interest for travellers

The Lakeland washdown station was noticed by many tourists on the drive north into the Cape, with some even driving onto it and attempting to get the wash down station working (see section 3). The future of the wash down station is undetermined.

Outreach to schools

Several schools were visited during this trip. They were: Western Cape Primary School, Lakeland Primary School, St Joseph's Parish School (Weipa), Northern Peninsula Area State College, and Tagai State College (tertiary campus on Thursday Island and primary campus on Horn Island).

Primary schools were supplied with a vegetable leafminer colouring activity, and instructions for conducting a 'mock surveillance' competition. Secondary school biology students at Tagai State College and agriculture students at Northern Peninsula Area State College were shown parasitoid wasps under the microscope and photos of vegetable leafminer damage.

The feedback from school staff was positive and the two secondary schools expressed an interest in future visits from the project researchers. Speaking about career paths into research is highly beneficial for local students as the region does not have a large number of scientists. In addition, the students are actively engaged in field work at the school farm in the case of Northern Peninsula Area State College. This means that students can notify local rangers if vegetable leafminer damage is observed at the farm as long as they are aware of what indicators to look out for (the Northern Peninsula Area State College is in Bamaga, an area in which vegetable leafminer has not yet been detected).

Biosecurity education and awareness

Biosecurity radio alerts

As you travel throughout Cape York Peninsula, you will see a number of signs asking you to help protect Australia by tuning to FM 88 on your car radio. This radio station is dedicated to biosecurity updates for travellers and is accessible near urban areas on the Cape. However, during this trip, which was undertaken in the first week of the tourist season, radio updates could not be accessed. This may have been due to a lack of alerts at that point in time or due to the radio frequency and area signal.

The biosecurity alerts are broadcast on low powered FM radio transmitters that reach within a limited distance around the sites where they are installed. The 'transmitters are run by the Department of Agriculture and Water Resources. According to the Australian Commonwealth publication 'Quarantine Protects Cape York' this broadcast should be accessible at the Hann River, the Cape York Peninsula Biosecurity Centre, Archer River, Moreton Telegraph Station, the Jardine ferry crossing and at Seisia. It would be useful to know how many travellers try to access the updates on this radio station, and how this station can be further used for further awareness.

Biosecurity signage

Signage for the NAQS public awareness campaign, 'Quarantine Top Watch', is prevalent. This signage encourages the public to report exotic pest species, and to comply with biosecurity regulations. Additional signage observed included an aged sign for controlling banana

movement near Bamaga, and an information sign describing the red banded mango caterpillar at Pajinka (the tip of Cape York). Examples of signs are shown in Figure 4.

Surveying of visitors to the Cape during this trip revealed that most understood that the Cape has special biosecurity status based on signs they had seen while driving north on the Peninsula Developmental Road. However, based on discussions with tourists during the survey component of this study, this signage could have greater impact if placed earlier, perhaps on the Mulligan Highway and in the southern section of the Peninsula Developmental Road.

On the road to the Jardine Ferry crossing there was one biosecurity sign, approximately 1 km south of the crossing, which showed the old quarantine zone, denoting the zone as one single zone from Coen north to the tip. Outdated signs are soon to be replaced with updated versions as a result of new funding, as described in 'Biosecurity authorities' section of this report.



Figure 4. Examples of roadside signs observed during this trip.

Biosecurity Inspection Centre

The Cape York Biosecurity Centre (formerly the Coen Information and Inspection Centre) was visited during this trip where we met with Scott Templeton, Biosecurity Compliance Officer, who briefed us on the role of the inspection centre and the region of Cape York Peninsula that is under QDAF jurisdiction for pest surveillance (FMBZ 1 and FNBZ 2).

The Inspection Centre was once based at Laura before moving to Coen. The car counter positioned at Musgrave during this period (the 1990's) counted 7000 cars travelling north. In 2017 the Cape York Biosecurity Centre car counter counted 27,000 cars travelling North. The centre managed to inspect 18,000 of those cars with two full time staff members and two casual employees (pers comms, Cape York Biosecurity Centre staff).

Tourists frequently stop at the Cape York Peninsula Biosecurity Centre, where they can pick up a variety of information materials and receive a briefing from staff about what the centre does, and what the major biosecurity risks are in the region (Figure 5). Feedback in the visitor guide highlighted the appreciation of visitors after receiving face to face biosecurity information.

Some feedback included:

- *'Love the info!!!'*
- *'Great video!!!'*
- *'Fantastic info! Thank you!'*
- *'Great info & very interesting!'*

During this visit a tourism company (Outback Spirit) arrived and requested a briefing at the centre. This tourism company was training its newest group of tourism operators and on this particular visit they had brought approximately 15 tourist-guide trainees for education from the centre. This represented a perfect opportunity to brief the trainees about VLM, as well as supply the company with awareness flyers. Under the recent biosecurity funding boost the Cape York Biosecurity Centre will soon be updated with new resources. This funding boost will also support extending operational hours of the centre (information packs are currently placed at the inspection point on the weekend when the centre is closed).



Figure 5. Awareness material and biosecurity displays at the Cape York Peninsula Biosecurity Centre, and the inspection point (bottom left).

Flyers and brochures

There were several information flyers and brochures available for travellers along the route (Figure 6). Apart from the selection of information guides made available at the Cape York Peninsula Inspection Centre, the 'Help Manage Biosecurity Risks in Australia' (Top Watch) brochure was available to the public at the Northern Australia Quarantine Strategy office on Thursday Island, as well as a flyer instructing visitors about what cannot be moved out of the Torres Strait without a permit.

The 'Travelling in Cape York and Torres Strait' guide, obtained at Coen, gives tourists an overview of biosecurity zoning and regulations in the region. There is also useful information in this guide in relation to pests (particularly weeds) that may be carried between regions if tourists are not careful. It is split into two major sections - considerations for those travelling north, and considerations for those travelling south. An important aspect to consider is when travellers are most likely to pick up this guide, and therefore, locations (and times during the year) along the Mulligan Hwy and Peninsula Developmental Road where they may be made available. For example, one page in this guide provides information on imikania vine, which has been found in Speewah near Mareeba. Therefore, having that information readily available for travellers before they travel from, or pass through, the Mareeba region would aid in targeted risk reduction.

The Peddles Ferry also carries biosecurity material that raises awareness about Torres Strait biosecurity zones. Laminated biosecurity handouts are supplied on each seat, and the ferry plays a video during its southerly transit that educates passengers about the Torres Strait Permanent Biosecurity Monitoring Zone legislation.

Other potential locations for awareness material include a popular tourist destination, Fruit Bat Falls, which is located slightly south of the Jardine Ferry Crossing, as well as the Jardine Ferry Crossing itself (Figure 7).



Figure 6. Information material picked up at the NAQS office on Thursday Island (top three images), and a laminated flyer supplied on the Peddles Ferry (bottom).



Figure 7. Tourists stop for a rare swim on the Cape at Fruit Bat Falls (left), and the view on the approach towards the Jardine Ferry crossing – the beginning of FNBZ1 (right).

Further regional insights

This trip involved meetings with several authorities in the region, and insights from these meetings have helped to contextualise the findings in this report. One meeting was held with Mr Eddie Newman, Mayor of the NPARC. Mayor Newman was able to highlight the risks faced by the NPA from plant pests and weeds moving from south to north, two examples of which were the cane toad (which has recently extended its range to Thursday Island) and gamba grass, which had been brought to QLD as a fodder crop several decades previously. Key insights about the region gained during this meeting are included below.

Pest transmission and pest management

- Weeds and seeds coming from the south is perceived to be a big issue.
- There are many plant pests spreading from the north – it is a constant issue.
- Chemical control of insects in the NPA would mostly be for mosquito and growing of produce is largely done organically.

Jardine Ferry Crossing

- There are no biosecurity check points for people moving north into the Cape. The Jardine Ferry crossing is a potential location for such a checkpoint.
- The Jardine Ferry crossing hours are likely to be extended from 6am-7pm although this is still to be confirmed. This would allow locals working south or north of the ferry to travel back home.
- The ferry crossing should ideally have a biosecurity checkpoint on both sides for travellers.
- There has been discussion about bringing the wash bay north from Lakeland to the Jardine Ferry Crossing.
- The Mayer suggested a practical investment – dongas could be built at the Jardine Ferry crossing, which would enable the Ferryman to work extended hours.
- Investment in biosecurity training for the ferry workers was also suggested, which would enable the ferry personnel to educate travellers.
- Biosecurity bins and new signage is needed, especially at the Jardine crossing, but potentially at other key locations on the drive north.

Tourists

- Tourist traffic has increased over the years but is also 'bigger' (e.g. there are caravans, quad bikes and buses travelling on the Peninsula Developmental Road now).
- Plans to completely seal the Peninsula Developmental road in the next decade was also noted. This will mean more tourist traffic moving north and south down the NPA, and between the Torres Strait and the mainland. This pest transmission pathway will come under increasing pressure.

Biosecurity inspections

- Not enough biosecurity inspections between the mainland and the Torres Strait islands are being carried out – the entry of the cane toad to Thursday Island highlights this.

- The Cape York Peninsula Biosecurity Centre is not active 24/7 and locals mainly travel at night and on weekends to beat the traffic, which means they would miss any inspection at Coen as they drove north.
- Dinghies routinely travel between islands and to the mainland and would avoid any biosecurity checks.*

*On advice from biosecurity authorities – *DAWR do intercept and inspect dinghies which may have crossed a quarantine zone – not all of these vessels can be checked by local officers who perform patrols at boat ramps. Quarantine signage is located at all boat ramps in the NPA.*

Travellers and biosecurity risks

Tourism on Cape York and in the Torres Strait is heavily seasonal, with touring being restricted to May and November due to monsoonal weather and road closures. A Cape York Tourism Action Plan was developed by Cape York Sustainable Futures (2015-2016) to guide infrastructure and development activities from 2016 to 2021. The action plan makes note of the expected changes to Cape York tourist type in the near future. It is predicted that, apart from the 4WD and fishing enthusiasts, visitation will increase to include shorter term travellers with caravans or motorhomes. It also predicts that visits from tourists flying directly in and directly out will increase.

Over recent years sealing of the Peninsula Developmental Road has begun. It is expected that further sealing of the road will lead to a change in the type of tourist visiting the Cape, as well as supporting an extension of the tourist season due to a reduced number of wet season road closures. At the time the plan had been developed the bulk of visitors were described to be mature aged, traveling in 4WDs and preferring to camp.

Infrastructure priorities included in the plan are:

- Continued improvements to the Peninsula Developmental Road;
- Communication and increased mobile phone coverage;
- Improved facilities at airports; and
- Dredging of the Endeavour River to enable large passenger ships to land.

The Cape York Region Package, which was announced in 2014 by the Queensland Government, allocated $260.5 million to improving infrastructure in the Cape. The funds, contributed between the federal government and the Queensland Government, have supported further sealing of the Peninsula Developmental Road between Laura and Weipa.

Tourism in the Cape York Peninsula Area has one major activity hub in the lower Cape based around Cooktown. The other major tourism hub is Pajinka (the Tip of Cape York) and Weipa?

According to the Cape York Tourism Action Plan the target tourist market is self-drive domestic and international visitors, who enjoy camping or modest accommodation (for example, 4WD adventure seekers, or those interested in fishing). This plan also highlights a secondary target market of those visitors who are interested in a short holiday and transit via flying, cruising, or driving. Periphery target markets include people seeking authentic indigenous experiences,

those seeking an Australian outback area adventure, birdwatchers, and ecotourists. According to this plan there is also the intent to build a bridge over the Jardine river.

Short-term visitors predicted to increase

A number of regional infrastructure and tourism planning initiatives in recent years have led to predictions that the number of short-term visitors will increase as the Cape becomes more accessible.

Tourist volume

It is difficult to determine exact annual Cape and Torres Strait tourist numbers. However, it is clear that the number is increasing with the rise of ecotourism and increased accessibility. There is a need to better understand tourist numbers, their purpose in visiting the Cape and Torres Strait, the method of travel, and origins, in order to better determine the risk of travellers as a pathway for exotic plant pests.

Sourcing data from the following companies may aid this risk assessment:

- Coen Centre (car counter).
- Seisia Caravan Park.
- Weipa Caravan Park.
- Peddles Ferry.
- Umagico Caravan Park.
- Loyalty Beach Camp Park.

Visitors through Bramwell Station Tourist Park

Bramwell Station is a major tourist stop during peak season. In the month of July it holds the Bramwell Music Muster that attracts thousands of tourists, as well as the Bramwell Cup Bush Carnival. On the night we arrived Bramwell Station had 12 tourists staying in the campsite. According to the station manager, the following week the number was expected to swell to 80 people each night. There are plans to develop an 'eco-tourism' route near the station, which would encourage visitors to stay longer at the station. In regard to the type of tourist that is visiting the station, the manager mentioned that caravans were very uncommon five years ago. During our stay we noted two of the six campsites pitched were caravans, the owners of which noted that they were pleasantly surprised by how easy it had been to transport the caravans up and down the Cape.

Visitor biosecurity awareness

Tourists represent a growing risk for transfer of plant materials and plant pests both north into the Cape and Torres Strait, and south into production regions and communities. With this risk pathway in mind, part of this engagement tour focussed on interviewing locals about their observations concerning growing numbers of tourists, verifying the type of tourist visiting the region, and interviewing tourists themselves to gauge their level of biosecurity awareness.

Tourist Survey

This trip was conducted in the first week of the tourist season, when visitors had started to arrive at the Cape and make their way north, mainly to camping grounds or fishing spots. Since awareness material was being supplied to each tourist park encountered during the trip it was decided that a biosecurity awareness survey would be undertaken with short term visitors at these campsites. Before surveying tourists, permission was received from the manager of each tourist park. Surveyed tourist parks were: Weipa Tourist Park, Bramwell Station, Seisia Holiday Park, and Loyalty Beach Campground.

The aims of this survey were to gauge the level of biosecurity awareness of short-term visitors to Cape York Peninsula, find out from where they source biosecurity information before travelling, and to determine the origins of these tourists. Interactions with tourists during each survey served as an opportunity to educate newly arrived tourists about biosecurity regulations on the Cape and the Torres Strait, as well as the risk posed by VLM. Visual aids (laminated maps and pictures of VLM damage) were used to supplement the briefing and those surveyed were supplied with the project flyer and vegetable leafminer awareness flyer.

After supplying an information sheet about the survey and gaining agreement from the surveyee, background information was collected on the following topics: Age range, occupation, origin, purpose of visit, method of travel, towage, and accommodation type. After collecting this information the following questions were asked:

1. In your view, how important are biosecurity policies & regulations for Australian communities and industries (1 = low, 5 = high)
2. Can you name a past or present exotic pest outbreak in Australia?
3. Have you ever heard of the Vegetable leafminer? (If yes, where did you hear about it?) (Yes/No/Unsure)

At this point the surveyee was supplied with a briefing on vegetable leafminer, the risk posed by the pest, where it was currently located on the Cape, and information about the project. They were also advised on how leafminer damage manifests and how they should report suspect detections. The surveyee was then asked the following questions:

1. Have you visited Cape York Peninsula / Torres Strait previously? (Yes/No/Unsure)
2. If yes, during your previous visit did you transfer plant products out of the Cape or Torres Strait? (Yes/No/Unsure)
3. Are you aware that Cape York and the Torres Strait are designated biosecurity zones? (Yes/No/Unsure)

At this point the surveyee was supplied with a briefing on the new biosecurity zoning in the Cape. The surveyee was then asked the final question:

1. Has your new knowledge raised your awareness about the importance of biosecurity on CYP and in the TS? (Yes/No/Unsure)

The surveyee was then invited to make further comment on the subject on the survey, biosecurity, or the vegetable leafminer and this discussion was documented. The total number of tourists surveyed was 62.

Demographics

The most common age range of those surveyed was between 50-60 at 34%, followed by 60-70 at 27%. Other age brackets were surveyed at the following frequencies: 20-30 at 11%, 30-40 at 16%, and 40-50 at 11%. Of those surveyed 27% were female and 73% male. Occupations were varied, however the most commonly supplied single occupation was 'retiree' at 23%. Other common occupations included trades, health industry work, and truck drivers (Table 3).

Table 3. Summary of occupations supplied by surveyees (n = 62)

Occupations	**% of surveyed**
Retiree	23%
Trades (Carpenter, fitter & turner, builder, tree lopper, arborist, electrician, rigger, maintenance supervisor)	23%
Health (Carer, nurse, nursing assistant, physio, disability support worker, mental health worker, health manager)	15%
Community services (Police, firefighter)	6%
Truck driver	6%
Producer (Blueberry grower, wheat grower, sheep farmer)	5%
Education	3%
Other (Vet, financial planner, financial manager, picker and packer, administration, student, court registrar, plant operator, security guard, hospitality)	19%

Response to survey questions

Purpose of visit, method of travel and origin

The Cape York Tourism Action Plan states that Around 90% of visitors are domestic with the balance being international travellers. This complies with the results from this survey whereby of those surveyed, the majority (95%) had driven up the Cape through Coen on the Peninsula Developmental Road (rather than having flown into a more northerly town and driven south). Five percent of the surveyed flew into Bamaga and rented a car, however, at the time of the survey these respondents had no intention of driving south.

The origin of those surveyed was captured in 42 of the 62 interviews. Based on the data recorded, 90% of surveyees had travelled up from the east coast of Australia, with three of those surveyed having flown directly into Bamaga and the remainder having driven north from their points of origin. The summary of tourist origins are included in Table 4.

All surveyees supplied 'holiday' as the reason for the visit, with 32% specifically stating 'fishing holiday'.

While 55% of those surveyed did not have towage, of those that did have towage, boats were most common at 43%, followed by campervans at 29%, caravans at 14%, and trailers at 14%. Of the surveyed that were towing boats, trailers or had no towage, 74% were staying in tents, and 26% had booked cabins.

Surveys were conducted on an individual by individual basis, however since tourists commonly travelled in groups (on average 2-6 people per group) the actual number of boats, caravans, and trailers is lower than these percentages suggest. For instance, due to sharing of boats within groups, the total number of boats was 12, and the total number of campervans was eight. One surveyee admitted he was nervous about bringing the caravan but had been surprised by how easy it had been to tow it on the unsealed roads to Seisia. At least two of the surveyees noted they would have brought a caravan but were unsure they could be towed on the roads in the Cape.

Of the tourists surveyed, 45% reported to have visited Cape York Peninsula or the Torres Strait previously. Of those return visitors, 61% had visited 1-2 times previously, 7% had visited 3-5 times previously, and 32% had visited more than 5 times previously.

Table 4. Origins of surveyees (n = 42)

State or territory (regions)	**% of surveyed**
Queensland (rural QLD, Cairns, Burdekin, Bowen, Gold Coast, and unspecified)	50%
New South Wales (Coffs Harbour, Sydney, Wollongong, Byron Bay, Brawley Point)	21%
Victoria (Shepparton, Mario, Melbourne)	19%
Western Australia	5%
Northern Territory	5%

Importance of biosecurity

In response to the following question '*In your view, how important are biosecurity policies & regulations for Australian communities and industries (1=low, 5=high)'* 81% of those surveyed answered with a 5 (high), 13% answered with a 4 (moderately high), and 6% answered with a 3 (moderately). In response to this question one surveyee felt that "they were not educated enough to give a confident answer". Further comments from surveyees in response to this question were: *"5 x 2!"*

Awareness of biosecurity pests

In response to the following question '*Can you name a past or present exotic pest outbreak in Australia?'* the following pests were supplied: Cane toad, citrus canker, fireweed, locusts, warehouse beetle, carp, pythenium, foot-in-mouth, prawn whitespot, "weed in Kakadu infesting waterways", "flying fish disease in New Zealand", fire ants, fruit fly, sugarcane rust, prickly pear, rabbit, lantana, blackberries, Paterson's curse, foxes, cats, wild dogs, panama disease, papaya fruit fly, spiralling whitefly, "SE QLD African fish (like a brim)", talarpia, miner bird, morning glory, bitterweed, crown of thorns, banana freckle, cucumber green mottle mosaic virus, red imported fire ant, red tulip, red banded mango caterpillar, wheat rust, bees, cane beetle, Hendra virus, mad cow disease, mother of millions, *Aedes agypti*, varroa mite, Indian miner, and pigs.

The most commonly mentioned exotic pest was the cane toad (mentioned 11 times). Some answers provided were very context dependent. For instance, a blueberry grower from Coffs

Harbour had supplied cucumber green mottle mosaic virus as an answer. This grower also produces cucumber and had attended an information event about the virus the previous year. In addition, one surveyee supplied red banded mango caterpillar and noted he had visited Punjinka the previous day and had read the biosecurity sign about the pest. Interestingly, many answers related to endemic pests, rather than exotic pests under eradication or containment. This question was useful in further assessing the level of knowledge these tourists had in relation to biosecurity risks, as well as placing these surveyees in a 'biosecurity' mindset before answering the remaining questions.

Knowledge of vegetable leafminer

In response to the question '*Have you ever heard of the vegetable leafminer? (If yes, where did you hear about it?) (Yes/No/Unsure)*' 15% of surveyees answered 'yes' and 85% answered 'no'. Of those who answered yes, sources of their knowledge were Landline (two surveyees), TAFE, TV, online, and from a previous employer (this surveyee had worked for the Queensland Department of Agriculture and Fisheries before retirement).

There was some confusion regarding vegetable leafminer. Most of those surveyed who replied 'yes' were unsure of the source of their knowledge. In addition, when surveyees who had responded 'yes' were questioned further about their knowledge it became apparent that in most cases they were confusing vegetable leafminer with an endemic leafminer species, most likely brassica leafminer. One surveyee also noted that they may be getting confused with citrus leafminer.

Knowledge of biosecurity zoning

In response to the question '*Are you aware that Cape York and the Torres Strait are designated biosecurity zones? (Yes/No/Unsure)*' 81% of surveyees responded with 'yes', 18% responded with 'no', and 2% (1 person) responded with 'unsure' (n = 62). When the source of this knowledge was queried, several tourists noted that they had seen the signs outside of Coen (with four surveyees reporting they had picked up information packs from the centre), they had been briefed by the biosecurity officer at the Cape York Peninsula Biosecurity Centre, or they had seen road signs in other locations along the Peninsula Development Road. Those that had picked up information packs from the Cape York Peninsula Biosecurity Centre mentioned reading the Travellers Guide to Interstate Quarantine supplied with the pack. Several surveyees also noted that they had noticed the truck washdown bay at Lakeland, and one had tried to drive onto it "*because I wanted a clean car*" but found that the bay had been decommissioned. Several surveyees noted that they had driven north through Coen on the weekend and questioned why the inspection centre was not open.

Only four respondents specifically mentioned that they had conducted pre-holiday research on the topic online and had taken note of biosecurity instructions in their hardcopy road maps. In some instances, those surveyed were aware that the Cape was a biosecurity zone (many used the term 'quarantine zone') but were surprised when informed that the Cape is split into biosecurity two zones. One surveyee responded that he "*thought it was just banana that couldn't be taken south*". In some instances where a surveyee was aware of the biosecurity zoning arrangements, there was confusion about the regulations, with one surveyee noting "*I wasn't sure if it applied to travelling both north and south*" and another noted that "*the*

biosecurity manual in the [information] pack was not clear about what could be brought in and taken out."

One major theme arose during discussion of the biosecurity zoning on the Cape – tourists were clearly taking notice of the biosecurity road signs during the drive north, with signs being mentioned by 23% (14) surveyees as a source of awareness.

Further comments from surveyees in regard to signage included: *"Better education is needed, and more signs"; "Why there are not more biosecurity signs on the Cape?"; "There should be more signage on the drive up"; "Pictures of what to look out for would be useful"; "We heard on the ferry to Thursday Island that the Torres Strait is a quarantine zone. I had also seen the signage at the ferry crossing"; "I noticed a quarantine sign in Somerset which was very old"; "I noticed a lot of signs about the red banded mango caterpillar, which was near the Seisia wharf.*"

This survey also sought to gauge how much plant material was transferred out of the Cape and Torres Strait by tourists. Of those surveyed who had visited the region previously (n = 28) 21% noted that they had transferred plant products out of the Cape or Torres Strait and 7% were unsure. Of those that had carried plant material out of the region previously on respondent mentioned that the plant material taken south was a coconut, while another noted they had taken plant material *"into the cape, not into Torres Strait".* Another who had replied 'no' qualified the answer with "*maybe burs on clothing*".

Knowledge change

In response to the following question '*Has your new knowledge raised your awareness about the importance of biosecurity on CYP and in the TS? (Yes/No/Unsure)'* 97% of respondents answered 'yes' and 3% answered 'no' (n = 62). Of those that answered 'no' they added that their biosecurity awareness was *"already high"* and they had picked up information packs from the Cape York Peninsula Biosecurity Centre during the drive north. Further comments from surveyees in response to this question were: "Absolutely, thanks for the information"; "It certainly has"; "Definitely"; "Definitely raised it"; "It was high anyway"; "Absolutely!"; "Already high"; "Already high and very mindful"; "Sure has".

Further comments from surveyees

Those surveyed were invited to make further comments following completion of the questioning. Those comments not covered in the previous sections are included below.

"This survey is the most I had ever heard about biosecurity".

"We were not aware Coen had an information centre and we'll stop in when driving south"

"We don't want that [VLM] going south".

It was noted by one of the surveyed that quarantine regulations "need to be strict"

"There was nowhere to dump plants or plant products at the Jardine Ferry Crossing"

"We had to park on the other side of the road from the centre to take an information pack, and there was no designated place to park. This made it confusing"

"Travellers could sign a document at the Jardine Ferry Crossing to ensure they understand the regulations"

Sources of biosecurity information

Several sources of information were mentioned by those surveyed, with signage, followed by the Cape York Peninsula Biosecurity Centre being the most commonly cited reason for having some knowledge of biosecurity regulations and risks in the region. One surveyee mentioned that they were aware that the Cape is a biosecurity zone because it was listed a road map he was using to navigate the region, but this map was outdated and did not show the two new biosecurity zones on the Cape.

This surveyee commented that he and his partner had gained a lot of information about the biosecurity status of the cape from WikiCamp Australia. This is a camping application that claims on its website to be "*the largest and most up-to-date database of campgrounds, caravan parks, backpacker hostels, day use area, points of interest, information centres and public dump points*". This surveyee made the suggestion that biosecurity authorities could place information and updates on that app.

When the search term 'biosecurity' is input on the WikiCamps chat forum only three chat threads appear. However, using 'quarantine' as the search term results in a plethora of threads. In many of these threads campers are requesting information about biosecurity requirements when crossing state borders and asking confirmation from other forum users about checkpoint locations (Figure 8).

Another pre-holiday source of information about biosecurity requirements was online 'grey nomad' forums, although none were specifically mentioned, although the main reason for checking these forums seemed to be for information on road status pre-arrival.

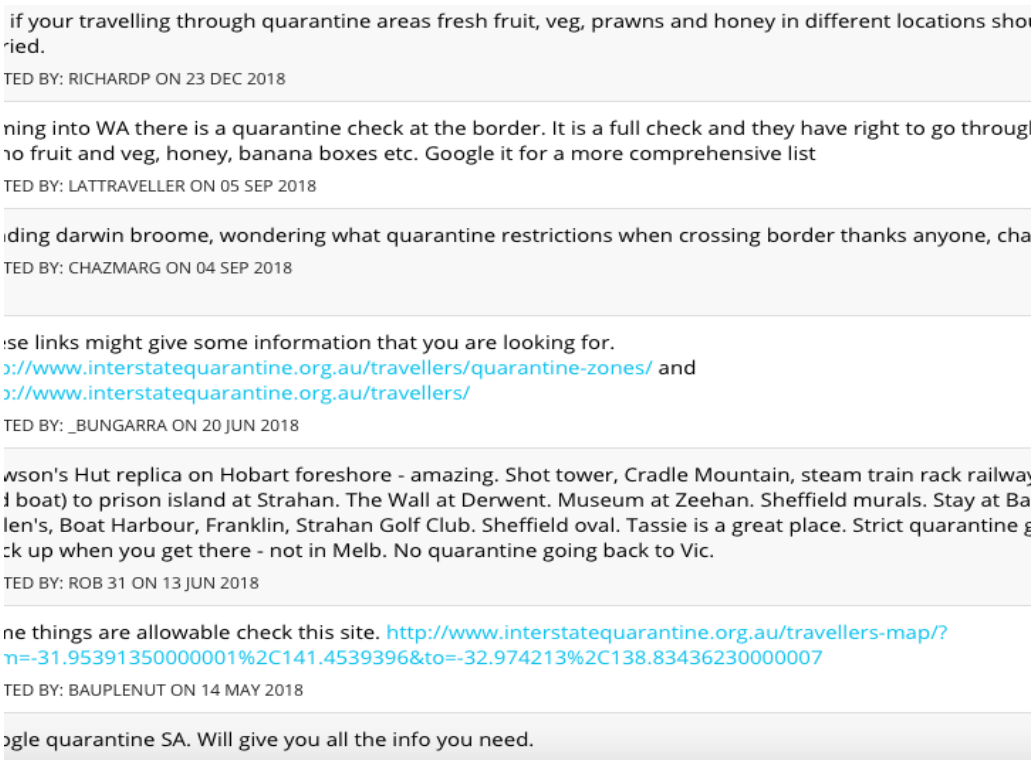


Figure 8. Excerpt from the WikiCamps Australia forum following input of the search term 'quarantine'.

Discussion

This was the first industry led and industry driven biosecurity scoping study to be carried out in the Cape York Peninsula and the Torres Strait. This initiative represents a concerted effort by the Vegetable industry and Nursery & Gardens Industry to develop networks in the region, learn about the operational components of the region, understand the biosecurity legislation that governs the region, and investigate the role that tourists may play as a risk pathway into southern growing regions.

One common question from tourists that were surveyed was in regard to how long it would take for VLM to spread south. At the Seisia Holiday Park there was a good example of the short time it could take an exotic pest to spread south when assisted by human movement.

Two surveyees travelling together were carrying two sweet basil plants (hosts for VLM) in their caravan, which had been brought up with them from south of Coen. They had placed the basil plants outdoors on the caravan step to get some sunlight. However, unbeknownst to these visitors their caravan was situated approximately 50m from a VLM infestation. They had intended to carry the basil plants south after leaving Seisia, however these caravaners were asked to leave the plants in Seisia and they quickly agreed.

The tourist survey highlighted that the region attracts a high amount of return visitors. This suggests that the region offers more than a new place to visit and adventure, and that short-term visitors to the region feel a bond to the place, which could support adoption of better biosecurity best practices. The fact that many tourists to the region may return should be kept in mind when developing biosecurity awareness and educational material, as well as when governments and other organisations decide where to present this information. However, it should also be noted that this survey was conducted in the first two weeks of peak season and it is possible that return visitors preferentially visit the Cape early in the season to avoid crowds both at the comp sites and on the fishing grounds.

It is clear that the numbers of tourists visiting the Cape and Torres Strait is increasing and will likely increase at a greater rate as infrastructure continues to improve. However, the tourist camping experience has also changed. One surveyee described a previous visit in 1988 when she had been "*camping rough*" as the cape was much different back then. During that trip she had survived on tinned food. However, good roads, car fridges, and proximity to goods means that tourists are now able to carry fresh produce fresh with them when moving up and down the Cape.

While many of those surveyed noted that they were aware of the Cape York Peninsula and the Torres Strait special biosecurity status, the source of their information was often from road signs noticed while driving through Coen or further north from Coen. A few travellers had also noticed the decommissioned wash bay at Lakeland. Few of those surveyed listed information sources accessed before the holiday. Therefore, according to the results of this survey many tourists travelling to the region by road would become aware of biosecurity regulations only once they had entered the Cape, which places far northern communities at risk from spread of southern pests.

While there are no regulations that restrict plant material from moving north, there are good examples of exotic pests moving north and disrupting the ecology of the region (e.g. gamba grass now contributes to bush fire risk on the Cape, and the cane toad is now found in abundance on Thursday Island). In consulting with the Mayor of NPARC during this trip it is clear that locals are concerned about what else may move north and impact on their way of life.

Based on advice from the Queensland Department of Agriculture and Fisheries, tourists are encouraged to ensure their equipment, vehicles and clothing are clean from plant material before entering Cape York and the Torres Strait (DAFF, 2018). However, prominent signage displayed on the Mulligan Hwy prior to Lakeland, or in the southern section of the Peninsula Development Road may contribute to further biosecurity precautions being taken by tourists.

Apart from the number of tourists entering the region each year, the type of traveller is also likely to change. One surveyee admitted he was nervous about bringing the caravan but had been surprised by how easy it had been to tow it on the unsealed roads to Seisia. Another surveyee noted he would have brought the caravan but was unsure it could be towed on the roads. Once tourists realise that the road is navigable with towage caravans and campervans will likely become commonplace.

As the Cape attracts more and more short-term visitors, recent funding government funding will support several important initiatives being undertaken, such as extension of operating hours at the Cape York Peninsula Biosecurity Centre, and updated signage. However, horticultural industry activities could further boost biosecurity in the region by leveraging existing initiatives.

As noted in this study, there are several existing biosecurity initiatives and plans for future initiatives with which Australian horticultural industries may partner. These include the Thoughtful Travelling campaign, information displayed at the Cape York Peninsula Biosecurity Centre, and the future information centre to be developed at Lakeland. In particular, the TSRA Sustainable Horticulture project may be approached to collaborate on pest management, pest identification, and soil management training in the Torres Strait, which would support the region in being an effective pest 'buffer' zone, support better nutrition in the region, and aid community-based pest surveillance. Councils in the NPA could be approached to partner on similar initiatives on the Cape.

There are also businesses in the region that have a stake in keeping it pristine and pest free. These include tour guide companies (such as Outback Spirit), holiday parks, and regional flight companies. Information about high priority horticultural pests and biosecurity best practices for tourists may be applied to these companies for upload to websites, and to aid briefing of tourists.

At the outset of the scoping study a series of questions were developed that the project had sought to answer. Findings from this scoping study are intended to be considered if further vegetable industry and nursery industry activities in the region are planned.

Recommendations

1. Investigate an opportunity to partner with the TSRA Sustainable Horticulture Project and provide support that would increase knowledge of crop production, crop protection, and pest surveillance at market gardens.

   (This may be in the form of sending industry representatives to hold pest management, biosecurity, or soil health workshops in these communities. Information provided by the vegetable industry about the nutritional value of different crops would also be beneficial for Torres Strait communities and would aid development of a partnership approach.)

2. Further investigate opportunities to supply awareness collateral for use at the future Lakeland Information Centre and to the Thoughtful Travelling Cape York Campaign.

3. Based on the location of each council and their proximity to major north-south transport routes, biosecurity partnerships with these authorities for the purpose of raising awareness among communities and tourists should be considered.

4. Investigate developing a biosecurity project for exotic horticultural pests that may be integrated into the regional school syllabus and completed by college students. Such a project may involve a pest surveillance element.

5. Investigate developing a biosecurity awareness training module and biosecurity awareness material for specific use by tourism and travel companies that cater to the region.

6. Authorities involved in planning locations of biosecurity signage and access to awareness material should consider the findings of the tourist survey in this report and place awareness collateral on the Mulligan Highway and southern section of the Peninsula Developmental Road.

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The State of Queensland, Department of Environment and Science, Soil survey of the Cape York Peninsula, Far North Queensland - CYP, licensed under Creative Commons Attribution 4.0

The State of Queensland, Department of Agriculture, Fisheries and Forestry (2018) Travelling to the Cape? Help protect Queensland from exotic pests and diseases information brochure

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Appendix 1

Price of fresh produce in Seisia - May 2018

Commodity	**Price**
Sugar snap peas	$29.99/kg
Garlic	$21.99/kg
Broccoli	$5.99
Celery	$5.99
Celery half	$4.00
Silverbeet	$5.49
Pumpkin (Jap whole)	$2.49/kg
Potato (washed)	$3.59/kg
Carrots	$3.59/kg
Sweetcorn	$1.89/kg
Half cabbage (sugarloaf)	$4.00 each
Zucchini	$8.49/kg
Leeks (bunch)	$7.49/kg
Cabbage (whole green)	$7.99/kg
Cabbage (whole red)	$8.99 each
Snow peas	$33.00/kg
Eggplant	$8.49 each
Cucumber	$3.29 each
Iceberg lettuce	$2.99 each
Baby cos lettuce twin pack	$7.49 each
Capsicum (green)	$7.49/kg
Capsicum (red)	$10.99/kg
Capsicum (gold)	$15.99/kg
Tomato	$5.00/kg

Appendix 10: *Liriomyza huidobrensis* Continency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Rohan Burgess [1], Dr. Peter Ridland [2], Dr. Elia Pirtle [3]

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December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Liriomyza huidobrensis Contingency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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December 7, 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

CONTINGENCY PLAN

SERPENTINE LEAFMINER (*LIRIOMYZA HUIDOBRENSIS*)



Merle Shepard, Gerald R.Carner, and P.A.C Ooi, Insects and their Natural Enemies Associated with Vegetables and Soybean in Southeast Asia, Bugwood.org



Central Science Laboratory, Harpenden, British Crown, Bugwood.org

December 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

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1. Purpose and background of this contingency plan 6
2. Pest details 7
2.1 Background and impact of pest 7
2.2 Life cycle 7
2.3 Host range 8
2.4 Signs and symptoms 8
2.5 Dispersal 10
2.6 Current geographic distribution 10
2.7 Risk of establishment within Australia 13
2.8 Risk pathways within Australia 14
2.8.1 Risk pathways – international and domestic 14
2.8.2 Predicting spread potential 19
2.9 Risk of economic impact within Australia 20
3. Diagnostic information 23
3.1 Diagnostic considerations 23
4. Surveillance and collection of samples 25
4.1 Surveillance 25
4.1.1 Technical information for planning surveys 25
4.1.2 Delimiting surveys in the event of an incursion into new areas 28
4.1.3 Activities for general surveillance immediately following a detection 30
4.2 Collection and treatment of samples 30
4.2.1 If collecting for morphological diagnosis 30
4.2.2 If collecting for molecular diagnosis 31
5. Immediate response to detection 34
5.1 Tracing 34
5.2 Quarantine and movement controls 34
5.3 Destruction strategy 34
5.3.1 Priorities and considerations 34
5.3.2 Decontamination of tools and equipment 35
5.3.3 Chemical control 35
5.3.4 Physical control options 40
5.3.5 Biological control 40
5.4 Decision support for eradication 41
6. Emergency Plant Pest Response Deed and PLANTPLAN 47
6.1 Emergency Plant Pest Response Deed 47
6.2 PLANTPLAN 48
7. References 49
8. Appendices 55
Appendix 1: Known hosts of serpentine leafminer 55
Number of host records for *L. huidobrensis* within plant families 55
Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*) has been provided within the scientific literature 57

Commercial, ornamental, and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*) has been provided within the scientific literature 72
Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*) 89
References 92
Appendix 2: Leaf mining damage caused by established species 97
References 109
Appendix 3: Permits for the control of serpentine leafminer and other *Liriomyza* spp. on different host crops 110
Appendix 4: Additional impact prediction outputs 127

1. PURPOSE AND BACKGROUND OF THIS CONTINGENCY PLAN

The serpentine leafminer (*Liriomyza huidobrensis*) is an exotic leafminer that affects a wide range of horticultural industries, including the vegetable and melon industries. It is currently widespread overseas and is present in nearby countries such as Indonesia.

This Contingency Plan provides background information on serpentine leafminer to assist in determining the requirements for the initial response to a detection of this species in Australia. Only key information to support an immediate response is provided in this document.

Additional information can be found in the following supporting material:

- Awareness material such as the fact sheets from Plant Health Australia (PHA), AUSVEG, and commonwealth, state and territory jurisdictions.
    - www.planthealthaustralia.com.au/pests/pea-leaf-miner/
    - www.agriculture.gov.au/pests-diseases-weeds/plant/leaf-miner
    - www.dpi.nsw.gov.au/biosecurity/plant/insect-pests-and-plant-diseases/exotic-leaf-miners
    - https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/
- Overseas websites with additional information
    - https://gd.eppo.int/taxon/LIRIHU
    - www.cabi.org/isc/datasheet/30956
    - https://cipotato.org/riskatlasforafrica/liriomyza-huidobrensis/
    - https://ufdc.ufl.edu/IR00004169/00001
    - http://ipm.ucanr.edu/PMG/r280300911.html

2. PEST DETAILS

Preferred common name:	Serpentine leafminer
Other common names	Pea Leafminer; South American leafminer
Scientific name:	*Liriomyza huidobrensis* (Blanchard, 1926)
Synonyms:	*Agromyza huidobrensis* *Liriomyza cucumifoliae* *Liriomyza decora* *Liriomyza dianthi*
Taxonomic position:	Class: Insecta Order: Diptera Family: Agromyzidae Genus: *Liriomyza* Species: *Liriomyza huidobrensis*

2.1 Background and impact of pest

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. Nearly all species are very host-specific, but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world.

The serpentine leafminer is one such species and is of concern as it has a wide host range including many horticulture crops in which it can cause significant yield losses and quality reductions (see Appendix 1). The serpentine leafminer is widespread overseas being present in North and South America, Asia, Africa, and Europe.

2.2 Life cycle

The lifecycle of the serpentine leafminer consists of an egg inserted by a female just under the surface of the leaf of a host plant. The egg hatches in 2-5 days. The larvae then begin to feed within the leaf creating tunnels or mines that get larger as the larvae moult and mature. After passing through three larval stages in 4-7 days the larva leaves the plant to form a puparium in the soil underneath the host plant. After 7-14 days (at 20°C and 30°C) an adult emerges and begins to reproduce (CABI and EPPO date unknown). The time taken to complete each life stage vary depending on host and temperature. With cooler temperatures resulting in longer development times. For example, Lanzoni et al., (2002) studied the effect of temperature on serpentine leafminer on common bean at 15, 20, 25 and 30°C, their results found total development times ranged from 43.6 days at 15°C to 16.1 days at 25°C, no adults emerged at 30°C. Similarly, Mujica et al., (2017) investigated serpentine leafminer development on faba bean and found development took between 65.62 days at 10°C to 13.48 days at 30°C, and that the lifecycle was not completed at 32 or 35°C.

The serpentine leafminer is reported to be able to survive winter temperatures as low as -11.5°C as pupae and can complete its lifecycle at temperatures between 5.7 and 30°C (EFSA Panel on Plant Health 2012).

2.3 Host range

Serpentine leafminer is a polyphagous pest of many agricultural and ornamental hosts (see Appendix 1) It has been recorded from over 250 plant species representing 50 plant families.

Important agricultural hosts include pulses, oilseeds, vegetables, herbs, cereals, and a wide range of ornamental species such as chrysanthemum as well as weed and pasture species.

A detailed host list is included in Appendix 1.

2.4 Signs and symptoms

Adult flies are small (1.3-2.3 mm long with a wing length of 1.5-2.2 mm (Okoth et al., 2014)) with a shiny black mesonotum and yellow markings on the head and (Figure 1).

Female flies use their ovipositor to puncture the leaves of the host plants causing wounds which serve as sites for feeding (by both males and females) or oviposition. Feeding punctures appear as white speckles between 0.13 and 0.15 mm in diameter. Oviposition punctures are usually smaller (0.05 mm) and are more uniformly round. However, the appearance of the punctures does not differ between *Liriomyza* species so cannot used to separate species.

Leaf mines are pale coloured with black and brown areas. Mines are created by the feeding larvae and are the most obvious symptom of infestations of *Liriomyza* spp. (Figure 2). Mines can be linear or serpentine and increase in width along their length as the larvae matures.

Factsheets with more information on what to look for in the field can be found at the following websites:

- www.planthealthaustralia.com.au/pests/pea-leaf-miner/
- https://ufdc.ufl.edu/IR00004169/00001
- http://ipm.ucanr.edu/PMG/r280300911.html
- https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/



Figure 1 Adult serpentine leafminer. Source: Central Science Laboratory, Harpenden, British Crown, Bugwood.org



Figure 2 Leaf mines on chrysanthemum. Source: National Plant Protection Organization, the Netherlands, Bugwood.org

2.5 Dispersal

Typically, *Liriomyza* leafminers are considered to have invaded countries via movement of infested plants (generally on ornamentals, but also some horticultural crops such as pea pods). While fully formed mines should be readily visible, signs of early infestations are much less obvious and are easily overlooked.

Agromyzid flies are considered as "moderate fliers" (Yoshimoto and Gressitt 1964) and in agricultural situations, the flies tend to remain close to their target crops, often only moving very short distances between host plants (Fenoglio et al., 2019). They also have the potential to be spread over large distances through wind dispersal. Flight mill experiments on the related *L. sativae* have shown that leafminers can fly up to 8.22 km but that the average flight distances was < 1.0 km at 18-36°C (Lei et al., 2002). Similar flight ability is expected for serpentine leafminer.

Serpentine leafminer adults have been shown to disperse on average 46 m within 24 hours via a release recapture experiment (Fenoglio et al 2019). Similarly, *Liriomyza trifolli* individuals within glasshouses have been shown to disperse on average 26 m but up to 100 m from original infestation points within 24 hours (Jones and Parella 1986) and in a field study were generally not found more than 50 m away from infested glasshouses (Ozawa et al 1999). *Agromyza frontella*, a related agromyzid leafminer pest of lucerne with similar flight dynamics, was observed to spread at a rate of about 100 km per year (or averaging about 0.3 kms per day) during an incursion into the Midwestern United States (Venette et al., 1980). There is also some evidence of long distance dispersal of Agromyzids via wind, with Agromyzid species reportedly trapped as high as 900 meters in the air column (Hardy and Milne 1938; Glick 1960; White 1970), and over oceans (Cheng 1976, Yoshimoto and Gressit 1964).

However, windborne dispersal is likely to be a minor pathway of leafminer dispersal in Australia, and human-mediated pathways should be prioritised, particularly given the accumulation of research pointing to this as a major contributor to the movement of invasive species (Capinha et al. 2015; Wichmann et al. 2009). Such pathways include transport of contaminated produce, or "hitch-hiking" adults associated with the large volume of human movement.

Since the 1980s the serpentine leafminer has spread from South America into Central America, North America, Europe, Africa and Asia through the movement of plant material (cuttings, cut flowers) (CABI and EPPO date unknown). This suggests that there is the potential for it to spread rapidly to new regions. Plant material should be considered as a potential pathway for the introduction/spread of this pest. The introduction of plant material from overseas is regulated, however illegal movement of plant material poses a potential risk for the entry of the pest into the country.

2.6 Current geographic distribution

The serpentine leafminer is thought to have originated in South America and since spread to North and Central America, Asia, Africa and Europe.

The current distribution of this pest is presented in Table 1. Note, serpentine leafminer has also been found at various times in a number of European countries including Bulgaria, Denmark, Finland, Hungary, Ireland, Lithuania, Slovenia, United Kingdom, but has since been eradicated are no longer present in those countries (EFSA Panel on Plant Health 2012).

Table 1. Countries where serpentine leafminer is known to occur

COUNTRY/REGION	REFERENCE
Argentina	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Scheffer and Lewis (2001); Spencer (1989); Weintraub et al., (2017)
Austria	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014)
Belgium	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Milla and Reitz (2005); Weintraub et al., (2017)
Belize	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005)
Brazil	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Spencer (1973); Weintraub et al., (2017)
Canada	Marin et al., (2005); Milla and Reitz (2005); Scheffer et al., (2001); Weintraub et al., (2017)
Chile	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Weintraub et al., (2017); Spencer (1973)
China	He et al., (2002); Maharjan et al., (2014); Milla and Reitz (2005); Weintraub et al., (2017)
Colombia	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Spencer (1973); Weintraub et al., (2017)
Comoros	Maharjan et al., (2014)
Costa Rica	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Scheffer and Lewis (2001); Spencer (1983); Weintraub et al., (2017)
Croatia	Maharjan et al., (2014)
Cyprus	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014)
Czech Republic	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014)
Dominican Republic	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Spencer (1989)
Ecuador	Maharjan et al., (2014); Scheffer et al., (2001); Weintraub et al., (2017)
El Salvador	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005)
France	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Trouvé et al., (1991); Weintraub et al., (2017)
French Guiana	Maharjan et al., (2014)
Germany	EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Weintraub et al., (2017)
Greece	EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Weintraub et al., (2017)
Guadeloupe	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz

COUNTRY/REGION	REFERENCE
	(2005); Spencer et al., (1992)
Guatemala	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Scheffer et al., (2001); Weintraub et al., (2017)
Honduras	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005)
India	CABI and EPPO (date unknown); Maharjan et al., (2014); Singh and Singh (2013)
Indonesia	Maharjan et al., (2014); Milla and Reitz (2005); Scheffer et al., (2001); Shepard and Braun (1998); Weintraub et al., (2017)
Israel	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Scheffer et al., (2001); Weintraub et al., (2017); Weintraub and Horowitz (1995)
Italy	CABI and EPPO (date unknown); Suss (1991); EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Weintraub et al., (2017)
Japan	Weintraub et al., (2017)
Jordon	Maharjan et al., (2014)
Kenya	Guantai et al., (2015); Maharjan et al., (2014); Milla and Reitz (2005); Weintraub et al., (2017);
Lebanon	Maharjan et al., (2014); Weintraub et al., (2017)
Malaysia	Maharjan et al., (2014); Weintraub et al., (2017)
Malta	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014)
Mauritius	CABI and EPPO (date unknown); Maharjan et al., (2014); Weintraub et al., (2017)
Mexico	Maharjan et al., (2014)
Morocco	Hanafi (2005); Maharjan et al., (2014); Weintraub et al., (2017)
Nepal	Maharjan et al., (2014)
Netherlands	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Milla and Reitz (2005); Weintraub et al., (2017)
Nicaragua	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005)
North Korea	Maharjan et al., (2014)
Norway	Maharjan et al., (2014); Weintraub et al., (2017)
Panama	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005)
Peru	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Spencer (1973); Weintraub et al., (2017)
Philippines	Weintraub et al., (2017)
Poland	EFSA Panel on Plant Health (2012); Maharjan et al., (2014)
Portugal	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012);

COUNTRY/REGION	REFERENCE
	Maharjan et al., (2014); Weintraub et al., (2017)
Réunion	CABI and EPPO (date unknown); Maharjan et al., (2014); Vercambre and De Crozals (1993); Weintraub et al., (2017)
Saudi Arabia	Dawah and Deeming (2002)
Serbia	Dobrosavljevic et al., (2017)
Seychelles	Maharjan et al., (2014)
Singapore	Maharjan et al., (2014)
South Africa	Maharjan et al., (2014); Milla and Reitz (2005); Scheffer et al., (2001); Weintraub et al., (2017)
South Korea	Maharjan et al., (2014); Weintraub et al., (2017)
Spain	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012); Maharjan et al., (2014); Weintraub et al., (2017)
Sri Lanka	Maharjan et al., (2014); Milla and Reitz (2005); Scheffer et al., (2001); Weintraub et al., (2017)
Sweden	Maharjan et al., (2014)
Switzerland	Maharjan et al., (2014)
Syria	Maharjan et al., (2014)
Taiwan	Milla and Reitz (2005); Shiao and Wu (2000); Weintraub et al., (2017)
Thailand	CABI and EPPO (date unknown); Maharjan et al., (2014); Weintraub et al., (2017)
Turkey	Maharjan et al., (2014); Weintraub et al., (2017)
United States of America (including Hawaii)	CABI and EPPO (date unknown); Maharjan et al., (2014); Scheffer and Lewis (2001); Spencer (1973); Spencer (1989); Weintraub et al., (2017)
Uruguay	Maharjan et al., (2014)
Venezuela	CABI and EPPO (date unknown); Maharjan et al., (2014); Milla and Reitz (2005); Spencer (1973);
Vietnam	Maharjan et al., (2014); Weintraub et al., (2017)
Zimbabwe	Weintraub et al., (2017)

2.7 Risk of establishment within Australia

CLIMEX models are provided in Jovicich (2009). These found that the serpentine leafminer is most likely to establish along the eastern, southern and southwestern coastline of Australia and Tasmania.

Refer to: https://ausveg.com.au/app/data/technical-insights/docs/VG06113.pdf for further details.

Similarly, Weintraub et al., (2017) suggest serpentine leafminer may likely establish in parts of Australia that have mild winters (i.e. do not receive snow) and have summer highs of 30-35°C.

Cesar Australia has created a model of the potential geographic distribution of the serpentine leafminer in Australia, the results of which are summarised below (Figure 3). A copy of the relevant

reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

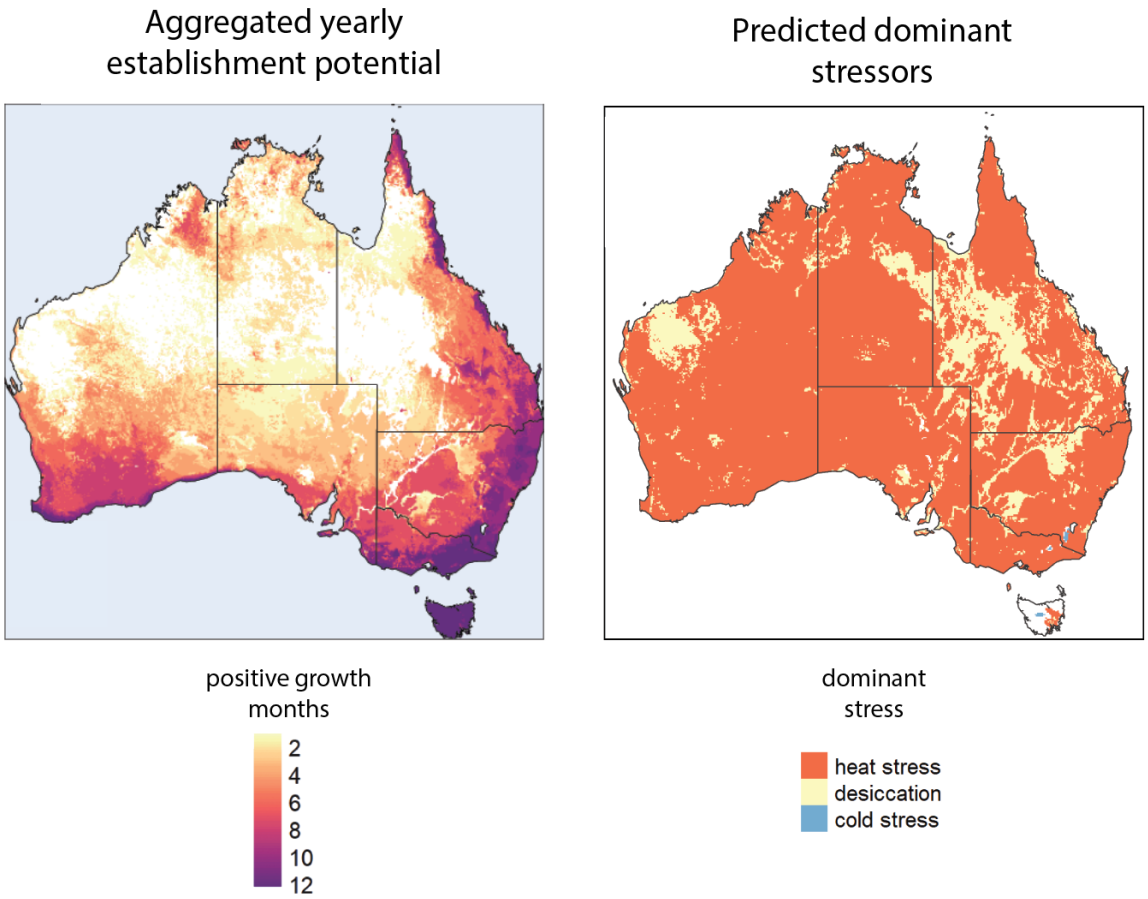


Figure 3 Aggregated yearly establishment potential of L. huidobrensis as the number of months across the year with increasing population sizes) based on temperature and moisture constraints (left) and predicted dominant stressors based on the highest mortality rate from desiccation, cold, and heat stress across the year (right).

2.8 Risk pathways within Australia

2.8.1 Risk pathways – international and domestic

This species could potentially enter new areas by hitchhiking on goods, aircraft, vehicles, or via the movement of plant material (DAWR 2016). Table 2 lists some of the potential international risk pathways and control measures in place. These are also be broadly applicable to other *Liriomyza* species.

Table 2. International risk pathways for the spread of the pest and control measures

RISK PATHWAY	DESCRIPTION	CONTROL MEASURES
Cut flowers on regulated imports	Infested cut flowers moved to residential dwellings that contain suitable hosts and the insect transfers to a host.	Border inspections
Infected plant material on passenger or mail	Infested plant material moved to residential dwellings that contain	Border inspections

RISK PATHWAY	DESCRIPTION	CONTROL MEASURES
pathways	suitable hosts and the insect transfers to a host.	
Natural dispersal from Papua New Guinea (PNG)	Ongoing spread from PNG to Torres Strait and/or Far North Queensland via traditional movements, winds or flight.	Ongoing surveillance via the Northern Australia Quarantine Strategy Domestic quarantine
Infested plant material	Plant material or pupae in soil spread to new areas in baggage, vehicles or camping equipment.	Domestic quarantine

Additionally, as the larvae of *Liriomyza* spp. leave the plant to pupate, pupae may also be spread with crop debris or soil associated with infested areas. Table 3 lists some of the potential risk pathways and potential domestic control measures to reduce the spread of serpentine leafminer. These will also be broadly applicable to other *Liriomyza* species.

Table 3. Risk pathways for the spread of the pest and potential domestic control measures

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Cut flowers (host plants)	Cut flowers that are a host of serpentine leafminer (see Appendix 1) pose a significant risk. The life of cut flowers is long enough to allow the completion of the pest's lifecycle (CABI and EPPO, date unknown). Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Hitchhiking adults and pupae may be transported on cut flowers.	The application of systemic insecticides may be required before allowing cut flowers grown in an infested area to be moved outside the infested area. Further information on chemical control is provided in Section 5.3. 3. All cut flowers leaving infested areas will also need to be visually inspected for signs of leafminers before leaving the area.
Cut flowers (non-host plants)	Cut flowers that are not a host of serpentine leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host cut flowers are transported with or spent time in close proximity with host cut flowers. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	To reduce the risk of spread, cut flowers that are not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with serpentine leafminer.
Nursery Stock (containerized host plants)	Nursery stock that is a host plant for serpentine leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Containerized nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae may also be present in the soil of containerized stock. Larvae emerge from the leaf and pupate in the soil under host plants meaning potting mix/soil can also potentially spread the pest. Larvae very occasionally attach to leaves or surrounding non-soil surfaces to pupate. Adults may potentially spread with containerized nursery stock as a hitchhiker.	Visual inspection of containerised nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae in the soil (see Figure 2, however pupae will be very hard to detect as they are mixed into the substrate) or attached to leaves and container surfaces. To reduce the risk of spread, containerised nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) should also be considered. Further information on chemical control is provided in Section 5.3.3.
Nursery Stock	Nursery stock that is a host plant for	Visual inspection of bare rooted

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
(bare rooted host plants)	serpentine leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Bare rooted nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae are less likely to be present in bare rooted stock than containerized stock. However, upon emergence, if larvae are unable to find a substrate to bury into, they will attach to any surface available (including tray walls or plant leaves) meaning pupae can potentially still spread via bare rooted stock. Adults may potentially spread with bare rooted nursery stock as a hitchhiker.	nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae attached to leaves or surfaces surrounding the roots (see Figure 2). To reduce the risk of spread, bare rooted nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Further information on chemical control is provided in Section 5.3.3.
Nursery Stock (non-host plants)	Nursery stock that is not a known host plant of serpentine leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host stock spent time in close proximity with host stock. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	Visual inspection of nursery stock for the presence of adults resting on leaves. To reduce the risk of spread, nursery stock that is not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with serpentine leafminer. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered as a precaution to limit the potential for spread.
Non-	People and small businesses	Should an incursion occur effort will

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
commercially grown seedlings and plants	selling/trading seedlings at a local level (e.g. at farmers markets, school fetes, roadside stalls, etc.) pose similar threats to those posed by nursery stock, (i.e. plants can spread eggs, larvae, pupae and potentially adults via host plants to new areas). However, being less regulated this is a difficult pathway to control and therefore potentially poses a higher risk than commercial nursery businesses.	be needed to identify those involved in the non-commercial trade of seedlings to reduce the risks that they pose. Similar to commercial nursery stock, host plants should not be moved from infested to non-infested areas without being inspected and treated with an appropriate systemic insecticide. Likewise, non-host crops grown in proximity to host plants should be inspected and/or treated as a precaution if grown in an area known to be infested with serpentine leafminer, as there is a small risk that non-host crops could potentially spread adult leafminers as hitchhikers. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered. Further information on chemical control is provided in Section 5.3.3.
Plant material including hay, plant debris and crop waste	Hay, plant debris and crop waste could potentially spread pupae and adults to new areas. Larvae are not expected to be able to survive long after plant debris and begins to wilt and rot (once it cannot feed on the plant, the larvae must vacate the leaf to begin pupation or die), however pupae will survive plant debris and crop waste. Adults may also hitchhike on hay, plant debris and crop waste.	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without being treated with an appropriate insecticide. Further information on chemical control is provided in Section 5.3.3.
Soil	Larvae emerge from the leaf and pupate in the soil under host plants (Capinera 2014). Soil therefore poses a potential risk for moving the pest to new areas.	Minimise potential spread by reducing the spread of soil off infested sites (e.g. by cleaning down machinery between properties).
Tools, equipment, and machinery	Tools, equipment, and machinery used on farm in the vicinity of host plants pose a low risk of spreading the pest, however, may transport pupae via soil deposits, or may transport adults.	Clean tools, equipment and machinery before moving off infested sites.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Transport vehicles	Vehicles in the vicinity of host plants could allow the pest to hitchhike (in cabins etc.) to new areas (DAWR 2016).	Clean vehicles before moving off infested sites.
Conveyances (includes crates, boxes, bins, pallets)	Crates, bins, pallets, etc. that are clean of soil are unlikely to spread leafminers.	As a precaution, clean conveyances before moving between sites to reduce any risk of spread.
Seed	There are no expected pathways for leafminer spread by the movement of seed.	Not applicable.
Fruit (without leaves)	There are no expected pathways for leafminer spread by the movement of fruit (without leaves). It is expected the process of harvesting and cleaning fruit will remove leaf and soil material that could spread larvae and pupae and would cause adults to disperse rather than hitchhike with fruit to new areas.	Not applicable.
Fresh leafy or green vegetables (including peas)	Mines can occur on leaves of a wide range of green vegetables. There is therefore a risk that leafminer eggs or larvae (and potentially pupae and hitchhiking adults) could be spread to a new area via the movement of infested leafy vegetables. Mines do not occur on fruiting structures, with the exception that *Liriomyza huidobrensis* can form mines in the pods of snow peas and disrupt market access (Gitonga et al. 2010). Thus, movement of snow pea pods may also pose some risk.	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without undertaking suitable treatments to reduce the risk of pest spread.

2.8.2 Predicting spread potential

Cesar Australia has created a model of the potential rate of area invasion by the serpentine leafminer at entry points across Australia, the results of which are summarised below (Figure 4). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

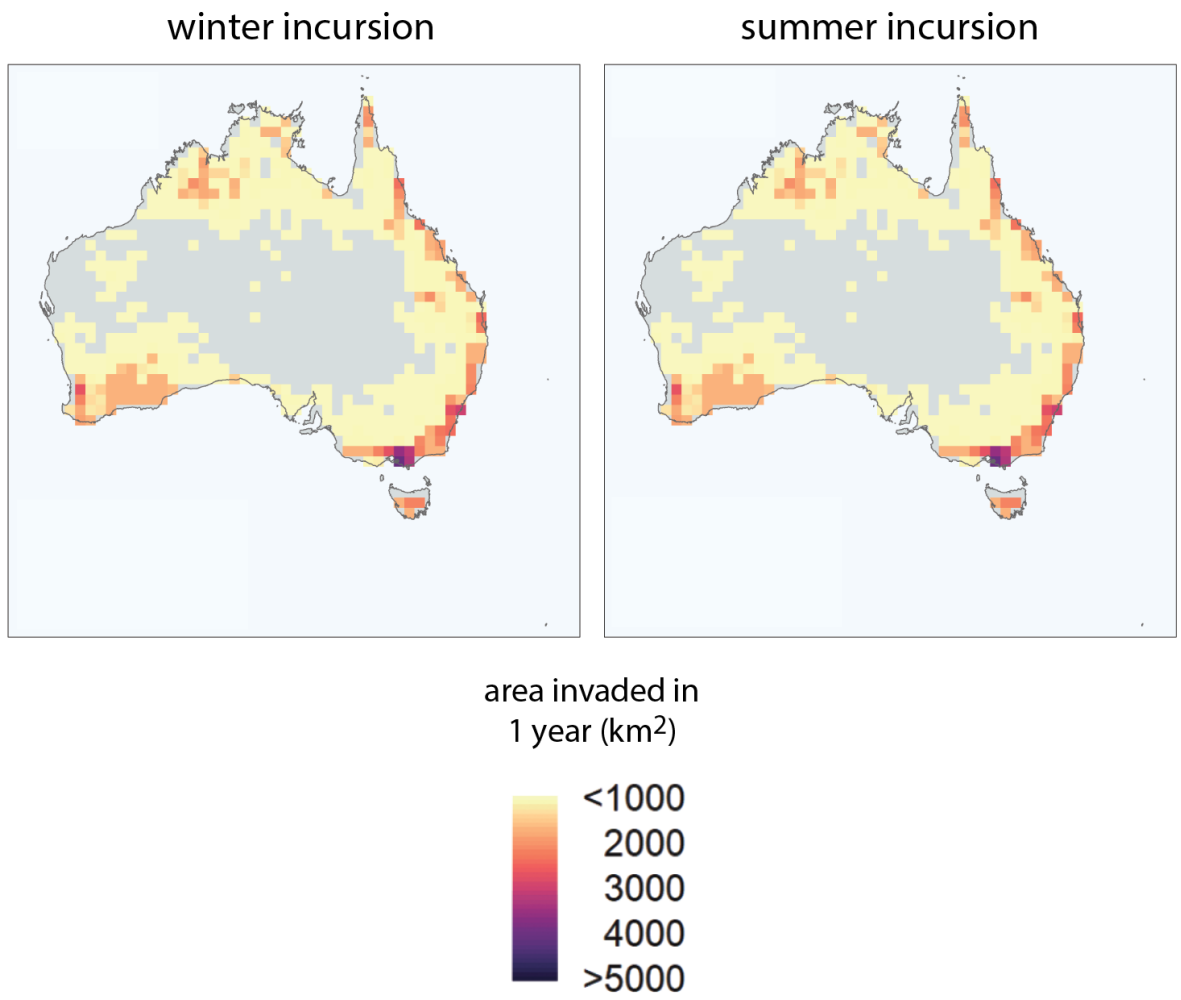


Figure 4 Rate of spread from locations across Australia. For each grid cell an incursion is simulated with the mean predicted area invaded by L. sativae in one year shown by the colour gradient. The mean predicted area is estimated from 10 replicated simulations. Incursions were commenced in July for a winter incursion and January for a summer incursion.

2.9 Risk of economic impact within Australia

Cesar Australia has created a model of the potential economic impacts of the serpentine leafminer in Australia, the results of which are summarised below (Table 4, Table 5, Figure 5). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

Table 4 Impact potential of serpentine leafminer on affected commodities in terms of international collated reports of proportions crop value lost due to direct feeding damage.

Host crop	proportion crop value lost			Sources
	Min	Max	Mean	
Beans	0.70	1.00	0.70	Rauf et al. 2000
Broccoli	0.10	1.00	0.10	Shepard et al. 1998
Capsicum	0.50	1.00	0.52	Spencer 1973; Elmore and Ranney 1954
Celery	0.15	1.00	0.46	Rauf et al. 2000; Weintraub et al 1998; Rauf et al. 2002
Cucumber	0.70	1.00	0.70	Rauf et al. 2000
Cut flowers	0.43	1.00	0.56	Rauf et al. 2000; Torres et al 1995

Potatoes	0.12	1.00	0.56	Rauf et al. 2000; Kwon et al. 2019; Waterhouse and Norris 1987; Chavez 1987; Chiluwal et al. 2012; Rauf et al. 2001; Shepard et al. 1998
Spinach	0.50	1.00	0.68	Rauf et al. 2000; Spencer 1973; Lange et al 1957
Tomatoes	0.00	1.00	0.15	Alves et al. 2017; Lopes et al 2019; Rauf et al. 2003

Table 5 Australia wide accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of serpentine leafminer at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preferences hosts may be overestimated while impacts of high preference hosts may be underestimated. Predictions were made using ESIM framework and are the culmination of the establishment, spread and impact modules (with each level of risk building upon the previous). For impacts calculated at state levels, see Appendix 4, Table 20, for additional information. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.

	Incursion Location					
Crop	***Bundaberg***	***Darwin***	***Devonport***	***Melbourne***	***Perth***	***Sydney***
Beans	1.50 (0.00)	0.00 (0.00)	1.24 (0.17)	0.05 (0.02)	0.00 (0.00)	0.00 (0.00)
Broccoli	0.00 (0.00)	0.00 (0.00)	0.50 (0.21)	3.49 (1.63)	0.01 (0.01)	0.01 (0.03)
Brussels	0.00 (0.00)	0.00 (0.00)	0.33 (0.04)	0.03 (0.08)	0.00 (0.00)	0.00 (0.00)
Cabbages	0.00 (0.00)	0.00 (0.00)	0.12 (0.00)	0.34 (0.16)	0.00 (0.00)	0.02 (0.03)
Capsicum	1.69 (0.00)	0.00 (0.00)	1.50 (0.03)	0.01 (0.02)	0.00 (0.00)	0.00 (0.00)
Carrots	0.00 (0.01)	0.00 (0.00)	3.73 (0.03)	0.03 (0.03)	0.02 (0.03)	0.00 (0.01)
Cauliflowers	0.00 (0.00)	0.00 (0.00)	0.97 (0.00)	2.00 (0.96)	0.00 (0.01)	0.01 (0.01)
Flowers	0.89 (0.07)	0.00 (0.00)	1.40 (0.85)	0.47 (0.51)	0.01 (0.00)	0.45 (0.47)
Lettuces	0.07 (0.00)	0.00 (0.00)	0.27 (0.81)	2.00 (0.91)	0.02 (0.02)	0.05 (0.05)
Melons	3.98 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Nurseries	0.99 (0.18)	0.00 (0.00)	1.60 (0.66)	1.95 (1.31)	0.03 (0.01)	0.76 (0.56)
Onions	0.00 (0.00)	0.00 (0.00)	3.72 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Peas	0.29 (0.00)	0.00 (0.00)	0.68 (0.14)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Potatoes	2.40 (0.00)	0.00 (0.00)	6.02 (0.08)	0.30 (0.37)	0.00 (0.01)	0.01 (0.02)
Pumpkins	0.35 (0.00)	0.00 (0.00)	0.14 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Tomatoes	5.50 (0.00)	0.00 (0.00)	0.35 (0.02)	0.00 (0.01)	0.00 (0.00)	0.02 (0.02)
Total	**17.68 (0.25)**	**0.00 (0.00)**	**22.57 (1.81)**	**10.67 (4.44)**	**0.09 (0.08)**	**1.35 (1.00)**

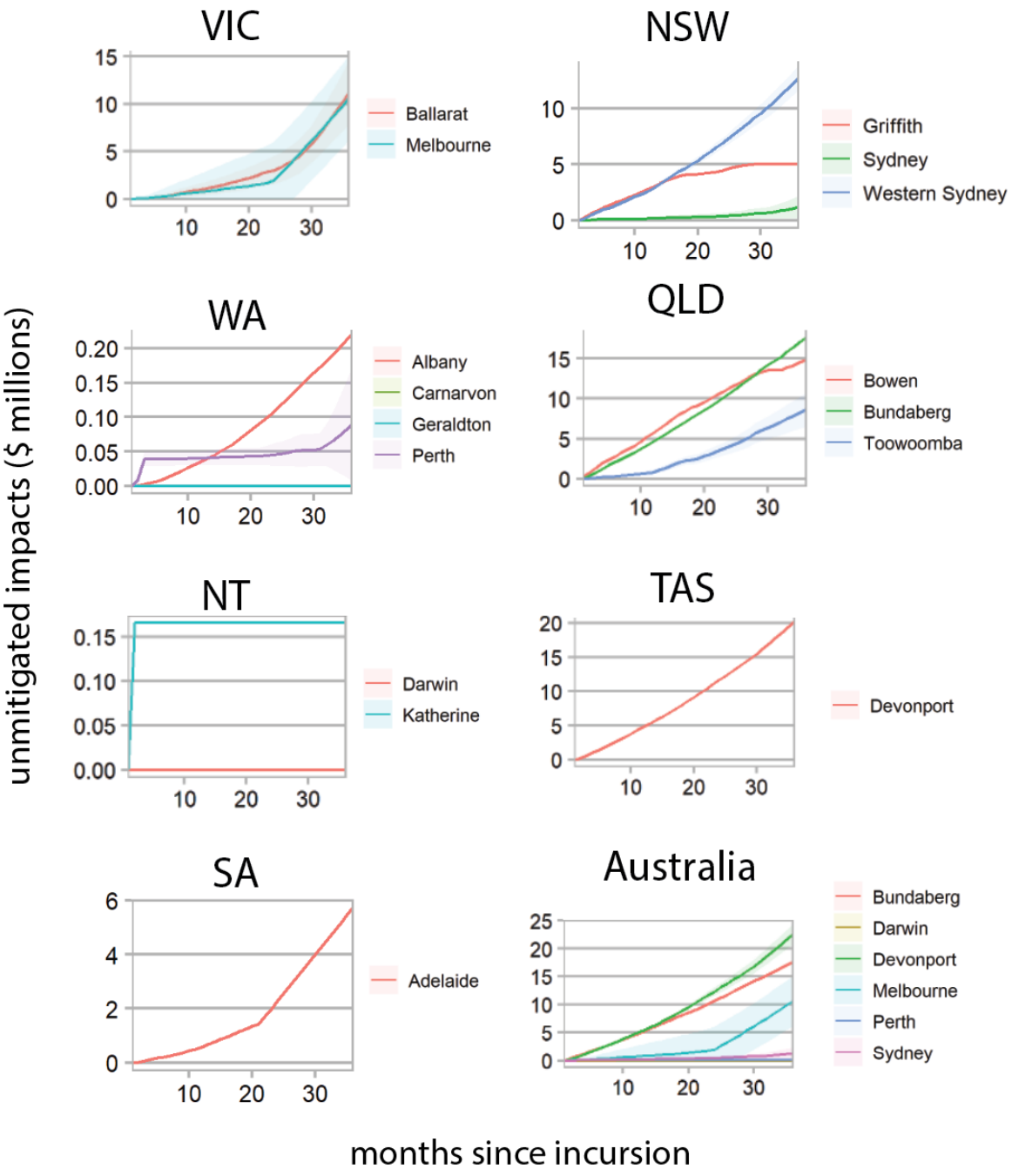


Figure 5 State level accumulating unmitigated impacts in millions of dollars across all host crops after 3 years resulting from a spring (September) incursion of serpentine leafminer at key entry and establishment points. For state level impact accumulation by crop type, see Appendix 4, Figure 9 for additional information

3. DIAGNOSTIC INFORMATION

Morphological diagnosis to a species level requires adult male flies. These can be collected by sweep netting, yellow sticky traps or collecting leaves with larvae in them and allowing the larvae to develop into adults. Alternatively, molecular tools can be used to identify larvae and adult flies.

A National Diagnostic Protocol for serpentine leafminer does not currently exist. However, an International Plant Protection Convention (IPPC) Diagnostic Protocol (IPPC 2016) and a European and Mediterranean Plant Protection Organization (EPPO) diagnostic protocol for *Liriomyza* spp. (EPPO 2005) are available. These diagnostic protocols should be referred to for the diagnosis of suspected *Liriomyza* spp. It should be noted that these protocols do not include information on environmental DNA (eDNA). Additional information on the use of eDNA for *Liriomyza* spp. leafminer diagnosis can be found at Pirtle et al., *in press*.

An information portal for polyphagous agromyzid leafminer identification is available at https://keys.lucidcentral.org/keys/v3/leafminers/index.htm, which includes species-specific fact sheets, and a pictorial Lucid3 key that allows users to make a preliminary identification of pest agromyzids and to be able to distinguish these species from non-pest endemic agromyzids. A factsheet for *Liriomyza huidobrensis* is accessible here: https://keys.lucidcentral.org/keys/v3/leafminers/key/Polyphagous%20Agromyzid%20Leafminers/Media/Html/Liriomyza_huidobrensis.htm

3.1 Diagnostic considerations

Considerations when triaging samples of suspected serpentine leafminer for diagnostics include:

- One of the greatest challenges in detection and subsequent management of *Liriomyza* species overseas has been the difficulty in correctly identifying the species (Reitz et al., 2013), which can have considerable overlap in adult morphology, host range and damage symptoms left on a plant. In the presence of indigenous leafminer, it may be difficult to detect a new incursion, or determine the limits of an infestation (Powell 1981).
- There are also several difficult to distinguish Agromyzid species present within Australia that are morphologically indistinguishable from serpentine leafminer to a non-expert (including *L. brassicae* and *L. chenopodii*). This is also true for some Agromyzids that are exotic to Australia. In many cases, specimens must be distinguished by expert taxonomists, or via molecular means.
- Within Australia, there at least 50 leafminer species (family Agromyzidae) (and presumably many more that have not yet been described) that create leaf mines that look similar to serpentine leafminer and may appear on the same host plants. Some of the most common include *Liriomyza brassicae* (which shares several brassica hosts with serpentine leafminer), *Liriomyza chenopodii* (which shares cultivated beets with serpentine leafminer), and *Phytomyza syngenesiae (*which shares cultivated asters with serpentine leafminer, but the adult form is readily distinguishable from serpentine leafminer) (Appendix 2). Moreover, there are an unknown number of additional Dipteran and Lepidopteran (Figure 8) species that share the leaf mining habit.
- Of these species that create similar leaf mining damage, some can be distinguished from serpentine leafminer via their mine characteristics, and their pupal or adult forms. (see Appendix 2, IPCC 2016, Salvo and Valladares 2004). However, the high degree of individual variation between each leaf mine means there can always be exceptions to these guidelines. For example, while serpentine leafminer pupate outside the leaf, there are rare instances where a larvae will fail to emerge and complete pupation within the leaf mine.
- Due to the high degree of morphological overlap of adults and visual overlap of damage, sample collection (of flies of any life stage and/or leaf mines) will be paramount for identifying and delimiting potential serpentine leafminer incursions.

- Surveyors should be familiar with the serpentine leafminer host plant list (Appendix 1), as well as with commonly observed signs of indigenous leafminer damage and the extent of known overlap with indigenous Agromyzids for common host genera (Appendix 2) before conducting serpentine leafminer surveillance. For example, damage caused by the closely related *L. brassicae* is essentially indistinguishable from serpentine leafminer and is very common on brassica crops (Appendix 2). Crops such as tomato, cucumbers and melons, on the other hand, have little to no reported overlap with indigenous Agromyzids (Appendix 2). Mines detected on these crops during surveillance activities should be considered highly suspicious.

4. SURVEILLANCE AND COLLECTION OF SAMPLES

4.1 Surveillance

4.1.1 Technical information for planning surveys

Detection and delimiting surveys are required to determine the extent of outbreaks, ensure areas free of the pest retain market access, and ensure appropriate quarantine zones are established to contain outbreaks.

Initial surveillance priorities for serpentine leafminer should consider the following:

- Visual surveillance for the leaf mines created by serpentine leafminer is expected to be the most reliable way to detect new incursions. This is because the mines created by leafminer larvae persist permanently on a leaf, extending the detection window beyond the lifespan of the fly itself. In comparison, sweep netting requires active populations be found, as do yellow sticky traps. While sticky traps can be left in place for extended periods to increase the chance of catching an adult, in open field settings, relying solely on sticky traps would be impractical due to the very large number of traps that would be needed to be placed and processed, to reach a statistically acceptable detection likelihood for small or isolated populations.
- In closed cropping systems, sticky traps become much more practical, but should still be accompanied by visual surveillance.
- Sticky traps will have a high level of bycatch (i.e. other insects will be caught in the trap), which can increase processing time for manual identifications. However, molecular diagnostic methods are also available for bulk invertebrate samples collected via a sticky trap (see Section 4.2.2.2).
- When undertaking visual surveillance in plant crops, the leaf mines are the most conspicuous symptoms to look for, though stippling from oviposition and feeding may also be apparent.
- Mines are expected to be more common on the edges of fields and in the upper portions of the plant (Weintraub 2001).
- The serpentine leafminer has an extensive list of weed hosts (see Section 2.3) and therefore surveillance in broadleaf weeds as well as cultivated crops should be conducted.
- Larvae, pupae and adults can be identified using molecular methods (refer to Section 3).
- Only adult male leafminers can be identified using morphological methods.
    - Adults can be captured using yellow sticky traps, sweep netting (IPPC 2016), or rearing from infested material. The latter is the most practical and reliable method for collecting adult flies. Infested leaves can be placed into an insectarium to develop into adults (IPPC 2016). Techniques for rearing agromyzids are described in Griffiths (1962) and Fisher et al. (2005).
- Collectors should be made aware of common signs of leafminers and note symptomatic plants when conducting surveillance for serpentine leafminer.
- Australia is already home to several leaf miner species, both native and naturalized, which can be confused for serpentine leafminer. Surveyors should be made aware of the native and

naturalized species they might expect to see in the particular region and crops they are surveying.

- Early detection surveillance will be most effective if timed when serpentine leafminer population growth potential is greatest, such as during warm (around 20-30°C) conditions. The seasonality of serpentine leafminer activity at major centers is provided in Table 6.

Table 6 Major Australian growing regions showing risk seasonality for serpentine leafminer activity across the year. Green means highest establishment risk (where the population growth potential was greater than 50% of the maximum predicted growth rate) yellow means lower establishment risk (where the population growth potential was between 1 and 50% of the maximum), and red means lowest establishment risk (where predicted growth rates are negative, meaning populations are predicted to decline).

State	Incursion point	Seasonality
		Jan Feb Mar Apr May Jun Jul Aug Sep Oct Nov Dec
Western Australia	Perth	
	Geraldton	
	Carnarvon	
	Bunbury	
	Albany	
Victoria	Thorpdale	
	Melbourne	
	Ballarat	
Tasmania	Smithton	
	Devonport	
South Australia	Mt. Gambier	
	Loxton	
	Adelaide	
Queensland	Toowoomba	
	Bundaberg	
	Bowen	
	Atherton	
Northern Territory	Katherine	
	Darwin	
New South Wales	West Sydney	
	Sydney	
	Griffith	

4.1.1.1 Surveillance tools

Yellow sticky traps and sweep netting can be used as supplementary methods to visual searches during delimiting surveillance activities, particularly when suspicious damage has been observed, or for monitoring existing outbreaks.

- Sweep netting is expected to be most effective when employed near recently active leaf mines, and is less effective near old leaf mines (which tend to be darker coloured than fresh mines, see Figure 6). Windy conditions can also reduce the effectiveness of sweep netting as adult leafminers tend to remain low in foliage during windy conditions (Foster 1986).



Figure 6 Younger mines (on the left) are more white-coloured with more distinct black trails of frass. Frass becomes less distinct as mines age, and mines develop a brown colour (right). Younger mines are expected to give the best sweep netting success. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

- Agromyzid flies are attracted to the colour yellow (Chavez et al., 1987), and can therefore be captured on yellow sticky traps. Yellow sticky traps are more effective for *Liriomyza* adults than other types of traps, such as funnel traps and yellow water pans (Chavez and Raman 1987). Vacuum sampling has also been shown to be a less effective means of sampling adult leafminer (Weintraub 2001).
- A great deal of effort overseas has been dedicated to improving the effectiveness of yellow sticky traps for *Liriomyza* adults (Heinz et al 1992) including the modifications of size, shape, adhesives, lures, height and orientation. For example, several studies report a strong effect of trap height on the number and species trapped (Trumble and Nakakihara 1983, Zehnder and Trumble 1984, Zoebisch and Schuster 1990), however these results do not always appear consistent and may be difficult to extrapolate across different crop types. Moreover, optimal height may vary considerably between *Liriomyza* species (Zehnder and Trumble 1984).
- Some considerations when using yellow sticky traps for *Liriomyza* surveillance are provided below:
    - Traps should be placed (vertically oriented) at the edges of paddocks.
    - Optimal trap height may vary with crop type. Studies on potato have found that traps placed at the crop's canopy height captured more flies than traps placed near the ground or above the crop's canopy (Weintraub and Horowitz 1996).
    - Traps should be left for a minimum of 24 hours, to ensure traps are in place during the peak flight time for serpentine leafminer, which occurs just after sunrise (Weintraub and Horowitz 1996).
    - Traps may be left in place for up to one month, at which point they should be collected, to prevent excessive degradation of DNA and allow for molecular confirmation of any leafminers caught on the trap. If traps are placed outdoors and exposed to the elements and/or high levels of bycatch they should be collected sooner than one month.

- Experimental lures developed from the extracted volatiles of known plant hosts have been shown to be attractive to *Liriomyza*. For example, lures made from spruce, basil, juniper or clove oil have been shown to attract serpentine leafminer (Gorski 2005).

4.1.2 Delimiting surveys in the event of an incursion into new areas

Following a new detection of serpentine leafminer, the following guidelines are recommended for delimiting surveillance:

- Surveillance to delimit a detection of serpentine leafminer should take into account tracing information as outlined in Section 5.1 to determine potential pathways for movement of material from the site of the initial detection.
- Surveillance should be a combination of the following:
    - Visual inspection in high risk areas (e.g. areas with suitable hosts).
    - Sweep nets to collect adults around infested host plants.
    - Yellow sticky traps, particularly in closed cropping systems and around infested plants/crops.
- Surveillance should be accompanied with awareness material, signs and personal visits to households and businesses within the surveillance zone and any buffer zones.

The suggested surveillance procedure for serpentine leafminer is as follows:

1. Delimiting surveillance should be conducted immediately upon detection of any incursions within a radius within 1 km of the incursion site, based on natural spread dynamics of agromyzid leafminer. The Surveillance zone may be increased as deemed necessary for the individual response.
2. At each site, chose a crop that is a preferred host plant of serpentine leafminer (refer to Section 2.3).
3. Choose a survey path that preferences the following areas if possible:
    - Paddocks and weeds near transport routes and unloading areas. These are preferred, because a major long-distance pathway of leafminer spread is human assisted movement.
    - Paddock edges should be included in the survey path, with the 'incoming wind side' preferred, since short distance movements of leafminer can be driven by wind.
    - Broadleaf weeds along paddocks should be included in the survey path.
4. Leafminer damage may be patchy at low densities.
5. At low densities, leaf mines have a relatively low detectability, despite their characteristic appearance. Slower search speeds will improve detection likelihood, however, when time is limited, search speed trades off with the total area covered (reducing the number of possible encounters). This trade-off is important in realistic paddock monitoring scenarios.
    - When time is not limited: To achieve a detection probability of 20%, 50%, and 80% respectively, when leaf mine density is about 1% of plants affected, the mean (and 95% CI) search time required per metre is 11.2 (4.8, 20.5), 34.8 (15.0, 63.8), and 80.8 (34.9, 148.2) seconds spend 1 minute searching per 1 m^2 of area (based on data collected during vegetable leafminer survey effort trials).

- When time is limited: The optimal search speed, balancing area coverage with sensitivity, when leaf mine density is about 1% of plants, is about 10 seconds per 1 m^2 of area. Further benefits are not achieved by increasing search speed beyond this level to achieve greater area coverage.(These recommendations are based on data collected during Vegetable leafminer (*L. sativae*) survey effort trials – other *Liriomyza* leafminers are expected to require similar effort)).

6. A survey plan should take into consideration any variables regarding the environment or surveyor that might reduce detectability and increase the effort spent accordingly. These variables might include:
    - *Plant type*
        i. *Leaf density:* leafminer damage may be harder to spot in plants that are very dense and leafy, than in plants with larger, sparser or flatter leaves.
        ii. *Plant age:* it may be easier to spot leaf mines in young plants with few leaves and more open space between plants than in larger mature, overlapping plants.
        iii. *Colour:* leafminer damage may be harder to spot against light coloured leaves than against darker leaves.
        iv. *Height*: Leafminer damage in plants that are at eye level may be easier to spot than crops that are low to the ground.
        v. *Time of day/direction of survey relative to the sun*: Survey success may be higher when surveyors are walking with their back to the sun, rather than walking into the sun.
    - *Residues on plants:* Chemical residue on plants may reduce the contrast between the mine and the leaf, or obscure mine patterns with background residue patterns.
    - *Damage from other pests/disease on plants:* damage caused by other pests or disease may obscure leaf mines, or lead to false positive detections.
    - *Surveyor experience:* Inexperienced surveyors may have a learning curve.
    - *Weather conditions:* Surveys during poor weather should be avoided when possible.
7. If suspicious damage is detected, samples should be collected (see Section 4.2), and yellow sticky traps should be placed around the affected area, in an attempt to capture adults and diagnose the fly responsible for the damage.
    a. If serpentine leafminer are confirmed, visual surveillance along with sticky traps should be used to monitor the edges of the outbreak area.
8. If no suspicious damage is detected, the initial round of delimiting surveillance should be followed with a second round occurring after one week.
    a. This second round is necessary as leafminer eggs are undetectable in the leaf, and hatch after about 2-5 days. A crop that appears clean during the initial survey may still develop symptoms within a week.
    b. After the first revisit, any further revisits should occur on a monthly basis. One lifecycle of the serpentine leafminer is estimated to take approximately 2 to 4 weeks, dependent on host and temperature.

4.1.3 Activities for general surveillance immediately following a detection

Given the wide host range of this pest and its noticeable impact it may be a suitable target for general surveillance programs that request submission of images and/or samples from the public.

To establish a general surveillance program, the following will be required:

- Use of online or app reporting tools such as MyPestGuide to promote the submission of reports of suspected detections.
- Factsheets to provide information on the pest, symptoms, impacts and reporting mechanisms.
- Media releases to describe the impact of the pest, surveillance programs and activities within the response program.
- Information for industry communication channels including articles for industry newsletters, magazines and websites, information for Twitter feeds and Facebook, and presentations for industry talks.
- A website to provide information for the public and for commercial businesses and links to other relevant sites.
- Release and promotion of information on details for physical sample collection and submission, as well as information on how to take and submit images of flies/leaf mines.
- Broader awareness campaigns should consider literature (brochures and factsheets) in several languages.
- A non-expert friendly surveillance and sample collection guide for vegetable leafminer, which also has relevance for the serpentine leafminer, as well as other useful information, can be found here.

4.2 Collection and treatment of samples

4.2.1 If collecting for morphological diagnosis

Of the four life stages (egg, larva, pupae and adult) only adult males are identifiable to a species level using morphological features.

4.2.1.1 Collection of adult male specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the foliage. However, the most practical and reliable method is the collection of leaves with mines containing mature larvae or pupae which can then be reared until adult flies emerge (refer to Section 4.2.1.3).

4.2.1.2 How to collect sticky traps for identification

Sticky traps should be left in the field for up to one month (but preferably 2 weeks or less), to allow morphological identifications. Upon collection, sticky traps should be folded lightly in half sticky side inwards (not pressed shut), and placed in a sealed, padded plastic bag. Traps should be sent within a week of collection (and kept in cool, dry place in the meantime, but not frozen).

4.2.1.3 How to collect plant samples for rearing

For rearing of adult flies, mined leaves containing pupae or mature larvae can be collected into an

insectarium and kept in a constant temperature room for regular checking. Simple insectariums include sealed partially inflated ziplock bags, or sealed rigid plastic containers or jars. A piece of paper towel should be placed into the insectarium along with the infested leaves to regulate humidity. Once pupae emerge from the leaves, they should be transferred carefully with a wet paintbrush to a new rigid container, which contains a layer of damp sand but no leaf material. Pupae can be placed directly onto the damp sand. Techniques for rearing agromyzids are described in the IPCC diagnostic protocol for *Liriomyza* (IPPC 2016, available from here).

4.2.1.4 How to preserve plant samples for identification

Identifying the plant species affected by suspected serpentine leafminer damage can aid diagnoses, particularly when surveying in weeds. To preserve a plant for identification, leaves can be stored between sheets of dry newspaper. However, if plant material with suspected leaf mines is pressed and dried, there will be no chance of rearing out adult flies from those leaf mines. Therefore, whenever plant identity is in question, infested material should be stored in an insectarium for rearing, and an accompanying pressed sample of uninfested leaves should be taken to facilitate for identification of the host.

4.2.1.5 How to preserve leafminers

Adults caught in sweep nets or reared in insectariums can be placed in 70% ethanol and stored indefinitely for morphological purposes. For adults reared in insectariums, it is advised to collect these adults in the afternoon, as emergences tend to occur in the morning, and enough time should be given for wings to fully expand before collection.

4.2.1.6 How to transport leafminers

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.1.7 How to transport plant samples

Plant samples may be collected if the host needs to be identified. Queensland Herbarium (2016) provides information on how to collect and preserve herbarium specimens. Samples should then be taken to state herbaria for identification.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.2 If collecting for molecular diagnosis

All life stages can potentially be diagnosed using molecular methods, including empty leaf mines.

4.2.2.1 Collection of specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the

foliage. However, as other life stages can be diagnosed using molecular methods leaves with mines containing pupae or mature larvae should also be collected for analysis. Rearing these life stages in an insectarium is not necessary.

4.2.2.2 How to collect sticky traps

Upon collection, sticky traps should be either:

- Placed inside a plastic sleeve that has been cut along one long side and along the bottom, the trap and sleeve can then be sealed in a plastic zip lock bag.
- Folded in half, sticky side inwards, and placed in a sealed plastic bag.

Traps should be sent within a week of collection (and kept in a cool, dry place in the meantime, but never frozen).

4.2.2.3 How to collect plant samples

Plant samples with suspected serpentine leafminer damage should be collected for molecular confirmation. If it is possible to discern a live larvae inside a leaf mine (which can be spotted through the leaf mine as a small yellow maggot, either by eye or with a hand lens, these samples are always preferable (which can be spotted through the leaf mine as a small yellow maggot, either by eye or with a hand lens, see Figure 7),. However, mined leaves that appear empty can also be collected. Empty mine samples that look youngest should be preferred, as they have the greatest chance of containing a live larva (see Figure 6).



Figure 7 A larva inside a leaf mine. Larvae are visible to the eye in later stages of development. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

4.2.2.4 How to preserve plant samples

Mined leaves containing pupae or mature larvae, or mined leaves that appear empty can all be placed straight into 100% Ethanol. Small mined leaves should be rolled and placed into the tube of absolute ethanol. For larger leaves, the mine should be cut out, with care taken to avoid cutting across the mine, and the cut out should be rolled and placed into the tube of absolute ethanol. These samples should be frozen and mailed within a week of collection for molecular diagnosis.

Due to the very small amounts of DNA within leaf mines, the odds of sample contamination is much higher than when working with whole flies. Careful hygiene must be employed when collecting and

preserving suspicious leaf mine samples. Mines from different host plants or different locations should never be stored in the same container. Samples should either be immediately preserved into on site, or should be placed into unused, clean, sealed plastic bags on site and preserved within the day. If samples from multiple hosts or sites are being processed together at a field lab location, all equipment must be sterilised between each host/location (i.e. cleaning counter tops, sterilizing tweezers and other tools).

Leaf mines may also be preserved onto FTA cards, however DNA quality is somewhat lower than for preservation into 100% ethanol (Pirtle et al *in press*). To preserve leaf mines onto FTA cards, remove the mined leaf from plant and push the leaf mine face down onto the FTA card. Smear the mine thoroughly on the FTA card, for about 30 seconds, and then close the card and place in small sealed plastic bag. Do not rub mines from different host plants/sites onto the same FTA card, and do not store FTA cards from different host plants/sites in the same sealed bag. Store the FTA cards in a cool, dark, dry place and send for analysis within a week of collection

4.2.2.5 How to preserve leafminers

Specimens required for molecular diagnostic work, including larvae, pupae or adults, should be killed and preserved in absolute ethanol, frozen at -80°C, or preserved on an FTA card.

4.2.2.6 How to transport leafminers and plant samples

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

5. IMMEDIATE RESPONSE TO DETECTION

5.1 Tracing

Detection and delimiting surveys are required to delimit the extent of the outbreak, support areas free of the pest to retain market access, and ensure that appropriate quarantine zones are established.

Extensive tracing (trace forward and trace back) may be feasible as *Liriomyza* spp. can be readily dispersed by the movement of infested plant material, soil and crop debris. The focus should be on high risk linkages including premises linked directly with the initial detection, particularly where movements of plant material (such as nursery plants and seedlings), or soil have occurred as these pathways may move serpentine leafminers over long distances.

Further information on possible risk pathways are presented in Section 2.8 and in Table 2.

5.2 Quarantine and movement controls

If Restricted or Quarantine Areas are practical, no plant material should be moved from the infested to unaffected areas without first being inspected and appropriately treated. The size of the Restricted or Quarantine Area will be dependent on the type and scale of the incursion. However, a zone 1 km in radius should be considered for initial delimiting surveillance activities (Section 4.3). This zone can then be expanded as needed depending on the specific situation.

Voluntary movement control should be considered for urban/residential detections. Voluntary controls would involve negotiation with residents to undertake inspection and treatment of goods prior to movement from infested areas. Residents should be advised on measures to minimise the inadvertent transport of the pest from the infested area to unaffected areas. Voluntary compliance is likely to be implemented for urban areas using awareness campaigns to highlight high risk goods/situations and appropriate treatments.

5.3 Destruction strategy

5.3.1 Priorities and considerations

If eradication of an outbreak is considered feasible, the priority will involve the destruction of the pest. Adult leafminers may be present around host plants; eggs, larvae and pupae may be present in the leaves of host plants, and pupae may be present within the first 5 cm of soil under host plants.

Considerations include:

- The infested plants may need to be sprayed with a suitable insecticide (refer to Section 5.3.3), then bagged and then buried (or tilled/ploughed directly into the soil if a large area is affected).
- Other host plants in the immediate area should be inspected and treated as needed.
- Soil and plant media in greenhouses should be treated (heat or chemically) to destroy any pupae that may be present in the soil.

- Machinery and equipment used on site may need to be washed down to minimise the potential risk of spreading the pest between sites.

5.3.2 Decontamination of tools and equipment

Machinery, equipment, tools and clothes should be cleaned and disinfected between sites to minimise the potential to spread leafminers, and other pests and diseases that may be present on the site, to new areas.

For points to consider when decontaminating, refer to the PLANTPLAN Guideline document: Disinfection and Decontamination (PHA, 2015). which includes guidelines for the destruction and decontamination of tools used when working with emergency plant pests.

Several state agriculture departments have also produced publications on cleaning down machinery and vehicles and should be referred to if decontamination is required, examples include:

- www.dpi.nsw.gov.au/__data/assets/pdf_file/0010/545554/procedure-decontamination-vehicles-and-equipment.pdf.
- www.daf.qld.gov.au/__data/assets/pdf_file/0011/58178/cleandown-procedures.pdf.
- http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/weeds/weedstop-vehicle-hygiene-program/machinery-hygiene.

5.3.3 Chemical control

Overseas *Liriomyza* leafminers are most damaging when parasitoids are removed due to the use of non-selective insecticides (Cesar Australia 2018; Murphy and LaSalle 1999). For the ongoing management of leafminers it will therefore be important that chemicals are selected that are reasonably selective and have low toxicity towards key parasitoids.

The European and Mediterranean Plant Protection Organization (EPPO) provides guidance (EPPO 2009) on how to manage and eradicate Vegetable leafminer infestations in Europe, which are broadly applicable to other *Liriomyza* spp., such as the serpentine leafminer. For Vegetable leafminer, the EPPO recommends that foliar sprays are applied at weekly intervals until control has been achieved. Importantly the EPPO suggests rotating chemicals and modes of action (where possible) each treatment (Table 7). In greenhouse environments the use of a suitable space treatment is also suggested (EPPO 2009). Based on this it would be ideal if pesticides from different mode of action groups are available for each host crop, as this would allow pesticides to be rotated rather than relying on a single product being continuously used to manage/treat the pest.

Table 7 An ideal chemical rotation schedule used for eradication of Liriomyza leafminers in Europe (refer to EPPO 2009)

WEEK	PESTICIDE USED TO TREAT INFESTED AREA
Week 1	Chemical A
Week 2	Chemical B
Week 3	Chemical C
Week 4 onwards	Repeat above sequence

Pesticides registered for the control of serpentine leafminer overseas include the following actives:

- Abamectin
- Azadirachtin
- Chlorantraniliprole
- Cyantraniliprole
- Cyromazine
- Diazinon
- Emamectin benzoate
- Indoxacarb
- Permethrin
- Spinetoram
- Spinosad
- Spirotetramat

It should be noted that Australian Pesticides and Veterinary Medicines Authority (APVMA) permits will need to be put in place before these pesticides can be used for the control of serpentine leafminers. Permits are being applied for as part of Horticulture Innovation Australia Project MT16004 (RD&E program for control, eradication and preparedness for Vegetable leafminer), and these are noted in Table 8.

As the serpentine leafminer can affect many plant species (including agricultural and ornamental species) (refer to Section 2.3), it is important that all potential hosts are covered by at least one, but preferably multiple, pesticide permits. Appendix 3 provides a summary of the pesticides that can be used on each crop type and host and should be used as a guide when treating new serpentine leafminer outbreaks, or if eradication is unsuccessful, managing the pest. As new permits are approved, they will be made publicly available on the APVMA permit database: https://portal.apvma.gov.au/permits.

Table 8 Insecticide permits that have been applied for or granted by the APVMA for the control of serpentine leafminer in Australia (as of July 2020)

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[1]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Cyromazine (MOA 17)	Contact and translaminar activity	Not effective against adults but being an insect growth regulator is very effective against larval stages (Cesar Australia 2018b). Experiments on the related *L. trifolii* found that Cyromazine was very effective against first and second instar but less effective against late instar larvae (Yathom et al., 1986).	Nursery stock (non-food)	333 g in 20 L water/10 tonne potting mix	PER83506	31 October 2022
			Fruit trees (non-bearing)	950 g/ha or 95 g/100 L applied in 1,000 L/ha.		
			Broccoli	1 kg/ha or 150g/100L water	PER81867	30 November 2023
			Fruiting vegetables – cucurbits			
			Fruiting vegetables – other than cucurbits (excluding mushrooms and corn)			
			Head lettuce			
			Legume vegetables			
			Root and tuber vegetables			
			Stalk and stem vegetables			
			Brassicas (excluding broccoli) **Note: this crop must be destroyed if treated and must not be made available for human consumption**			
			Leafy vegetables (excluding head lettuce) **Note: this crop must be destroyed if**			

[1] Refer to Appendix 3 for detailed information on chemical control options.

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[1]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
			treated and must not be made available for human consumption			
Dimethoate (MOA 1B)	Contact and systemic activity	Dimethoate is reported to control *Liriomyza* spp. adults and larvae however resistance is known overseas (Tran and Takagi 2005)	Pulse crop (grains)	75ml (400 g a.i/L products)/100L water	PER89184	31/3/2025
Emamectin benzoate (MOA 6)	Contact and translaminar activity	Same mode of action as abamectin. Reported efficacy against larvae, adults and eggs (Cesar Australia 2018b)	Brassica vegetables (including broccoli, Brussels sprouts, cabbage, cauliflower)	250 to 300 g/ha (44 g a.i/kg products); or 650 to 780 mL/h (17 g a.i/L products) (suppression only)	PER87563	30 June 2024
Spinetoram (MOA 5)	Contact, systemic and translaminar activity	Spinetoram has shown efficacy against *L. sativae* adults and larvae (Shimokawatoko et al., 2012)	Vegetable legumes (peas and beans)	400 ml (48 g a.i)/ha	PER87878	28/2/2023
Spirotetramat (MOA 23)	Contact, systemic and translaminar activity	Affects the development of larvae and has little impact on adults (Ring 2019)	Capsicum	400ml (96 g a.i)/ha	PER88640	31/5/2023
			Celery			
			Chili			
			Eggplant			
			Green beans			
			Lettuce (head and leafy)			
			Parsley			
			Rhubarb			
			Snow & sugar snap peas			

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[1]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
			Tomato			

Research undertaken as part of the Hort Innovation project MT16004 has identified the suitability of the following as potential control options for *Liriomyza* spp.:

- Azadirachtin (a systemic insecticide that acts as an insect growth regulator, MOA unknown) on ornamental and floriculture plants.
- Abamectin (an insecticide with contact and translaminar activity, MOA 6) on fruiting vegetables, cucurbits, leafy vegetables, legume vegetables, bulb vegetables, cabbage, celery, and rhubarb.
- Cyantraniliprole (a systemic insecticide, MOA 28) on bulb vegetables, fruiting vegetables, cucurbits, melons, and potatoes (note residue data will be required to support this use pattern).
- Diazinon (a contact insecticide, MOA 1B) on beans, melons, beetroot, lettuce, parsnips, tomatoes, and potatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Dimethoate (a systemic insecticide, MOA 1B) on green beans, peas, tomatoes, capsicums, and potatoes. Note can have significant impact on beneficial insects and not fit well with IPM systems.
- Emamectin Benzoate (an insecticide with contact and translaminar activity, MOA 6) on brassicas, leafy vegetables, fruiting vegetables, cucurbits, and melons.
- Indoxocarb (an insecticide with contact and some translaminar activity, MOA 22A) on tomatoes.
- Permethrin (an insecticide with contact activity, MOA 3A) on tomatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Pyrethrins (an insecticide with contact activity but very limited residual activity, MOA 3A) on brassicas, leafy vegetables, lettuce, fruiting vegetables, and legume vegetables.
- Spinetoram (an insecticide with contact, systemic and translaminar activity, MOA 5) on cotton.

Permits for these use patterns have not been applied to date. However, in the event of an incursion, emergency permits for these use patterns could be developed if required.

5.3.4 Physical control options

Destruction of host plants and deep ploughing of crop residues can assist with leafminer control as adult leafminers have difficulty emerging from pupae buried deeply in soil.

Removal of weed hosts of the leafminer and planting susceptible crops away from weed hosts can also assist with the ongoing management of the pest by removing/avoiding potential leafminer reservoirs.

5.3.5 Biological control

Although not specifically for eradication, biological controls will assist in lowering leafminer populations and will be an important part of the management of serpentine leafminers should eradication not be possible in the event of an outbreak.

Parasitoids often provide effective suppression of leafminers in the field when non-selective insecticides are not used (Cesar Australia 2018). History has also shown that invading *Liriomyza* populations are rapidly exploited by endemic agromyzid parasitoids (Cesar Australia 2018). For these reasons it will be important that any pesticides used for the ongoing management of leafminers are compatible with Integrated Pest Management (IPM) systems.

Weintraub et al., (2017) identified more than 100 species of parasitoids that are reported to affect serpentine leafminer (Table 9) these represent 7 families of organisms. Agromyzid parasitoids tend not to be very host-specific and Australia already has several endemic species that would likely be effective against the serpentine leafminer. *Diglyphus isaea* is an example of a parasitoid that occurs in Australia and is mass reared overseas for the biological control of *Liriomyza* species including serpentine leafminers.

The importation of biological control agents is not warranted at this time as the adventive parasitoids such as *D. isaea*, already occur in Australia and would likely be suitable for the management of leafminers assuming pesticides are selected appropriately and are compatible with IPM systems.

Table 9 parasites and parasitoids of serpentine leafminer noted in Weintraub et al., (2017)

PARASITE NAME	PARASITE NAME	PARASITE NAME	PARASITE NAME
Agrostocynips clavatus	*Dacnusa sibirica*	*Halticoptera helioponi*	*Opius* sp.
Alloxysta sp.	*Dacnusa* sp.	*Halticoptera patellana*	*Pediobius metallicus*
Asecodes delucchii	*Diaulinopsis arenaria*	*Halticoptera peviana*	*Phaedrotoma luteoclypealis*
Asecodes sp.	*Diaulinopsis callichroma*	*Halticoptera* sp.	*Phaedrotoma mesoclypealis*
Bracon intercessor	*Diaulinopsis* sp.	*Hemiptarsenus fulvicollis*	*Phaedrotoma scabriventris*
Chrysocharis ainsliei	*Diglyphus albiscapus*	*Hemiptarsenus ornatus*	*Phaedrotoma* sp.
Chrysocharis bedius	*Diglyphus begini*	*Hemiptarsenus* sp.	*Platynocheilus cuprifrons*

PARASITE NAME	PARASITE NAME	PARASITE NAME	PARASITE NAME
Chrysocharis brethesi	*Diglyphus crassinervis*	*Hemiptarsenus unguicellus*	*Pnigalio incompletus*
Chrysocharis c.f. *aluta*	*Diglyphus intermedius*	*Hemiptarsenus varicornis*	*Pnigalio katonis*
Chrysocharis caribea	*Diglyphus isaea*	*Hemiptarsenus zilahisebessi*	*Pnigalio soemius*
Chrysocharis flacilla	*Diglyphus minoeus*	*Heteroschema* sp.	*Pnigalio* sp.
Chrysocharis ignota	*Diglyphus pachyneurus*	*Moneucoela* sp.	*Proacrias* sp.
Chrysocharis orbicularis	*Diglyphus pedicellus*	*Neochrysocharis beasleyi*	*Proacrias thysanoides*
Chrysocharis oscinidis	*Diglyphus pedicellus*	*Neochrysocharis diastatae*	*Proacrias xenodice*
Chrysocharis pentheus	*Diglyphus poppoea*	*Neochrysocharis formosa*	*Quadrastichus liriomyzae*
Chrysocharis pubicornis	*Diglyphus pulchripes*	*Neochrysocharis okazakii*	*Quadrastichus* sp.
Chrysocharis sp.	*Diglyphus* sp.	*Neochrysocharis* sp.	*Sphegigaster* sp.
Chrysocharis tristis	*Diglyphus* sp. (near *intermedius*)	*Notoglyptus tzeltales*	*Thinodytes cyzicus*
Chrysocharis vonones	*Diglyphus websteri*	*Oenanogastra sp.*	*Thinodytes* sp.
Chrysonotomyia sp.	*Disorygma pacifica*	*Opius caricivorae*	*Tribliographa* sp.
Cirrospilus ambiguus	*Epiclerus* sp.	*Opius chromatomyiae*	*Trichomalopsis* sp.
Cirrospilus vittatus	*Ganaspidium* sp.	*Opius dimidiatus*	*Trichopria* sp.
Closterocerus cinctipennis	*Gronotoma adachiae*	*Opius dissitus*	*Zaeucolia* sp.
Closterocerus okazakii	*Gronotoma micromorpha*	*Opius mandibularis*	*Zagrammosoma latilineatum*
Closterocerus pulcher	*Gronotoma* sp.	*Opius meracus*	*Zagrammosoma multileneatum*
Closterocerus sp.	*Halticoptera arduine*	*Opius pallipes*	*Zagrammosoma* sp.
Dacnusa sasakawai	*Halticoptera circulus*	*Opius scabriventris*	

5.4 Decision support for eradication

A summary of key activities associated with different detection scenarios of serpentine leafminer is provided in Table 10. Differing physical detection scenarios have been chosen to highlight operational differences in urban compared with agricultural and horticultural environments.

A summary of factors to be considered for eradication or alternative action is provided in Table 11.

Table 10 Recommended responses and considerations for differing scenarios of detection of serpentine leafminer

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Commercial glasshouse/ protected cropping setting	➢ Destruction of hosts within glasshouses could be considered to limit food sources or refuges (see Section 5.3.4). ➢ Chemical sprays to eradicate larvae and adults (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and in glasshouses managed by the same business as the infested glasshouse (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around the Infected Premises (IP). ➢ Focus on known hosts. ➢ Surveillance at linked properties. ➢ See Section 4 for points to consider when establishing a surveillance program. Key points for glasshouses are: • Undertake visual surveillance of all host plants. • Consider using visual surveys, sweep nets and yellow sticky traps.
Open agricultural/ horticultural setting	➢ Spray infested area using appropriate insecticide (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and survey jointly managed properties (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around each IP. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance awareness campaigns may be beneficial. ➢ Surveillance at linked properties should occur.

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Urban environment	➢ Chemical sprays to eradicate larvae and adults. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls in infested areas could be undertaken by permit or as voluntary movement controls (coupled with awareness campaign for urban areas).	➢ Surveillance on host plants within 1 km zone around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Surveillance to be undertaken in conjunction with awareness campaigns for households providing information on the pest and symptoms. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ Surveillance at linked properties.
Open natural environment (e.g. detection on roadsides or national park)	➢ Treatment of detections in a 1 km zone pending delimiting surveillance. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls in infested areas could be undertaken by permit or as voluntary movement controls. ➢ Movement restrictions may be considered pending outcomes of delimiting surveillance.	➢ Surveillance on host plants within 1 km zone around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance and awareness campaigns should be considered. ➢ Surveillance should be undertaken at linked properties.

Table 11 Summary of factors to be considered in determining whether eradication or alternative action will be taken for an incursion of serpentine leafminer

FACTORS TO CONSIDER REGARDING THE TECHNICAL FEASIBILITY OF ERADICATION
• **The population size and population structure associated with the initial detection.** • **The cost effectiveness of recommended control technique options.** - Multiple applications of several different pesticides over a period of time (weeks/months) may be required to successfully eradicate serpentine leafminer populations. • **The ability to remove or destroy all serpentine leafminers by the recommended control techniques.** - Permission to enter private premises for surveillance and treatment must be considered. - Determination of whether treatments can effectively eradicate populations within premises or environments. • **The recommended control techniques are publicly acceptable.** - Chemical treatments in residences or backyards may not be acceptable or will require negotiation with residents. • **Whether emergency containment measures can effectively be put in place to contain the outbreak.** • **Whether there are control methods, commonly employed for endemic pests, that may prevent the establishment of, or be impacted by the treatment, of serpentine leafminer.** • **Legislative impediments to undertaking an eradication response.** - If not a prescribed pest, confirmatory diagnosis will be required under legislation in some jurisdictions before powers to enter premises and undertake treatments can be enacted. - Delimiting surveillance in urban environments may be restricted to front yards and environs rather than entry into premises (unless permission from the resident can be negotiated). - Emergency permits will be required for chemical treatments. Amendments to chemical labels will be required for longer term management of the pest if eradication is not possible. • **The ability to delimit the known area of infestation.** - Determination of linked properties in an urban detection may be unfeasible given the ability of adult leafminers to disperse at a local level. High risk linkages such as the recent purchase of seedlings, cut flowers or other commodities that could vector the pest should be investigated. • **The ability to identify and close the pathway for entry of the pest into Australia.** - Pathways into Australia such as on nursery plants from overseas are managed via Post Entry Quarantine. - Natural dispersal is a possible pathway from neighbouring countries into northern Australia. Northern Australia Quarantine Strategy (NAQS) surveys offer an early detection mechanism to detect natural introductions from the north. If an outbreak is detected the pathway will need to be identified and closed. • **The dispersal ability of the pest.** - Serpentine leafminer has the ability to naturally spread over short distances through flight or wind assisted dispersal. - Nursery material that is a host of serpentine leafminer (including containerized plants, seedlings, bare rooted plants, etc.; see Appendix 1 for host list) and cut flowers that are host plants are the most likely means of spread to new areas. They may carry all life stages of the pest and will need to be managed to reduce the spread of the pest to new areas. Nursery material and cut flowers that are not a host of serpentine leafminer are at significantly lower risk (but may still transport adults across short periods if they have been in close proximity with host plants). - Plant material (potentially including fresh hay and crop debris) and soil could also facilitate the spread of the pest (via adults and pupae) to new areas. Tools, equipment, machinery and vehicles are lower risk but may still transport hitchhiking adults or pupae via soil deposits.

- Serpentine leafminer is considered unlikely to travel with harvested fruit (without leaves) as the process of harvesting or moving produce is likely to cause adults to disperse and provided soil and leaf material is absent it is unlikely to spread larvae and pupae. Leafminer are also not expected to travel via the movement of seed.
- Fresh green or leafy vegetables or fresh legume vegetables (e.g. snow peas) may allow the spread of eggs and larvae and potentially adults and pupae. Therefore, movement restrictions or mandatory treatments may be needed for some vegetable crops.
- See Table 3 for further details on possible dispersal pathways

- **The capability to detect *Liriomyza* leafminers at very low densities for the purpose of declaring freedom, and that all sites affected by the pest have or can be found.**
  - Surveillance typically relies on the use of visual inspection of host plants for mining activity as well as the use of yellow sticky traps and sweep netting.
- **The ability to put into place surveillance to confirm proof of freedom.**
  - Surveillance options are available for serpentine leafminer and include visual inspection of host plants for mines, yellow sticky traps and sweep netting.
- **Whether community consultation activities have or will be undertaken.**
  - In an urban environment, community consultation will be critical to assist in securing public support for delimiting surveillance and an eradication program.
  - Given the symptoms caused by this pest on a wide range of plants, general surveillance activities should be implemented comprising of awareness material, media releases, web-based reporting tools and the Exotic Plant Pest Hotline (1800 084 881).

6. EMERGENCY PLANT PEST RESPONSE DEED AND PLANTPLAN

The Emergency Plant Pest Response Deed (EPPRD) and PLANTPLAN should be referred to, in conjunction with this contingency plan when developing a Response Plan in the event of a pea leafminer detection in Australia.

6.1 Emergency Plant Pest Response Deed

PHA is the custodian of the EPPRD. This is a formal legally binding agreement between PHA, the Australian Government, all state and territory governments and national plant industry body signatories.

It covers the management and funding of responses to Emergency Plant Pest (EPP) incidents, including the potential for Owner Reimbursement Costs (ORCs) for growers. It also formalises the role of plant industries' participation in decision making, as well as their contribution towards the costs related to approved responses.

Under the EPPRD an EPP is defined as a pest that meets one or more of the following criteria:

a) It is a known exotic Plant Pest the economic consequences of an occurrence of which would be economically or otherwise harmful for Australia, and for which it is considered to be in the regional and national interest to be free of the Plant Pest.
b) It is a variant form of an established Plant Pest which can be distinguished by appropriate investigative and diagnostic methods and which, if established in Australia, would have a regional and national impact.
c) It is a serious Plant Pest of unknown or uncertain origin which may, on the evidence available at the time, be an entirely new Plant Pest or one not listed in Schedule 13 and which if established in Australia is considered likely to have an adverse economic impact regionally and nationally.
d) It is a Plant Pest already found in Australia that:
    (i) is restricted to a defined area through the use of regulatory measures intended to prevent further spread of the pest out of the defined area or into an endangered area; and
    (ii) has been detected outside the defined area; and
    (iii) is not a native of Australia; and
    (iv) is not the subject of any instrument for management which is agreed to be effective risk mitigation and management at a national level; and
    (v) is considered likely to have an adverse economic impact such that an emergency response is required to prevent an incident of regional and national importance.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of the EPPRD.

6.2 PLANTPLAN

Underpinning the EPPRD is PLANTPLAN, the agreed technical response plan for an EPP incident. It provides nationally consistent guidelines for response procedures, outlining the phases of an incursion, as well as the key roles and responsibilities of industry and government during each of the phases.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of PLANTPLAN.

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8. APPENDICES

Appendix 1: Known hosts of serpentine leafminer

The serpentine leafminer (*Liriomyza huidobrensis)* is highly polyphagous, with a wide host range across at least 15 families (Table 12). While host records for *L. huidobrensis* have been reviewed in previous studies (Weintraub et al. 2017), in order to facilitate trade and market access decisions, a thorough review was conducted as part of the Horticulture Innovation Australia funded Project MT16004 (R&E program for control, eradication and preparedness for vegetable leafminer), with a focus on evidence for lifecycle completion in the field. The results of this review, including commercial, ornamental and non-cultivated hosts of serpentine leafminer (*L. huidobrensis*), are presented in Table 13, Table 14 and Table 15 below. No guarantee can be made that these lists are exhaustive. Each record was scrutinized based on evidence provided showing completion of lifecycle within the field, and then included within the appropriate table. It is important to note that non-cultivated plants often serve as important reservoirs for *Liriomyza* pests (e.g. Schuster et al. 1991), and upon incursion into a new region, host ranges may expand to include native plant species. Moreover, host preferences may vary considerably across populations. For example, although chili (*Capsicum* spp.) is widely reported internationally as a favoured host of the closely related agromyzid *Liriomyza sativae*, the population of *L. sativae* which has established within the Torres Strait (IPPC 2017) has very rarely been observed to attack chili (despite it being an abundant garden host in the region) (Elia Pirtle pers. comm).

Number of host records for *L. huidobrensis* within plant families

Only records with either comprehensive or partial evidence of lifecycle completion in the field are included (see Table 13 and Table 14). Multiple records of the same species are included. Thus, the number of records does not indicate total number of affected species within each family. *Liriomyza huidobrensis* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 12 Number of host records for L. huidobrensis within plant families.

Family	Number of records
Asteraceae	81
Brassicaceae	36
Fabaceae	32
Solanaceae	29
Amaranthaceae	27
Cucurbitaceae	20
Caryophyllaceae	13
Apiaceae	12
Amaryllidaceae	10
Malvaceae	7
Ranunculaceae	7
Poaceae	6
Convolvulaceae	5
Lamiaceae	5

Family	Number of records
Polygonaceae	5
Chenopodiaceae	4
Gentianaceae	4
Passifloraceae	1
Plantaginaceae	4
Plumbaginaceae	3
Tropaeolaceae	3
Violaceae	3
Alstroemeriaceae	2
Araceae	2
Araliaceae	2
Balsaminaceae	2
Basellaceae	2
Campanulaceae	2
Euphorbiaceae	2
Iridaceae	2
Liliaceae	2
Primulaceae	2
Verbenaceae	2
Acanthaceae	1
Alismataceae	1
Apocynaceae	1
Asparagaceae	1
Asphodelaceae	1
Calceolariaceae	1
Hydrangeaceae	1
Linaceae	1
Linderniaceae	1
Menispermaceae	1
Onagraceae	1
Onagroideae	1
Oxalidaceae	1
Papaveraceae	1
Polemoniaceae	1
Rosaceae	1
Scrophulariaceae	1

Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*) has been provided within the scientific literature

This includes all records where the following conditions were met:

(1) Adults were reared from plant material infested with eggs or from larvae collected in the field; and

(2) Emerging adults were subsequently identified via morphology or molecular diagnostics; and

(3) Scientific names of hosts are reported

In addition, all records from Kenneth Spencer, the foremost authority on leafminer taxonomy, are included. Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included only if there are other records for species within this genus that met both conditions. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records confirming lifecycle completion for that same species that met both conditions. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. For all non-cultivated plants, and asterisk after the scientific name indicates that the Atlas of Living Australia reports this species present within Australia (Atlas of Living Australia 2020). Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report only partial confirmations of lifecycle completion are included in Table 14, while unverified records are included in Table 15. *Liriomyza huidobrensis* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 13 Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the serpentine leafminer (Liriomyza huidobrensis) has been provided within the scientific literature.

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial		*Beta* sp.		Amaranthaceae		Collins (1996)
Commercial	Beet; Chard; Beetroot	*Beta vulgaris*		Amaranthaceae	Nakamura et al. (2013); Rauf et al. (2000); Scheffer et al. (2001); Takano et al. (2008)	Andersen et al. (2008); Echevarria et al. (1994); He et al. (2002); (Koch and Waterhouse 2000); Korytkowski (2014); Korytkowski (1982); Ripa et al. (1995); Saunders et al. (1998); Spencer (1973); Weintraub et al. (2017); Wijesekara (2010); Kox et al. (2005)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Swisschard; Chard; Common beet	*Beta vulgaris*	*cicla*	Amaranthaceae	Hammad and Nemer (2000); Mujica and Kroschel (2011); Salvo and Valladares (2002)	He et al. (2001); He et al. (2002); Salvo and Valladares (1997); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial	Beetroot	*Beta vulgaris*	*rapacea*	Amaranthaceae	Salvo and Valladares (2002)	Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial		*Beta vulgaris*	*rubra*	Amaranthaceae		Shiao and Wu (2000)
Commercial	Beetroot	*Beta vulgaris*	*saccharifera*	Amaranthaceae		Hidalgo and Carballo (1991)
Commercial	Beetroot	*Beta vulgaris*	*vulgaris*	Amaranthaceae	Mujica and Kroschel (2011)	
Commercial	Spinach; Silverbeet	*Spinacia oleracea*		Amaranthaceae	Foba et al. (2015); Mujica and Kroschel (2011); Nakamura et al. (2013); Rauf et al. (2000)	Andersen et al. (2008); Bahlai et al. (2006); He et al. (2001); (Koch and Waterhouse 2000); Korytkowski (1982); Martin et al. (2005); Spencer (1973); Valladares et al. (1996); Kox et al. (2005)
Commercial	Leek; Leeks	*Allium ampeloprasum*		Amaryllidaceae	Rauf et al. (2000)	Hincapie et al. (1993)
Commercial	Onion; Shallot; Multiplier onion	*Allium cepa*		Amaryllidaceae	Foba et al. (2015); Rauf et al. (2000)	Andersen et al. (2002); Andersen et al. (2008); He et al. (2001); Hidalgo and Carballo (1991); Hincapie et al. (1993); Korytkowski (2014); Korytkowski (1982); Kuhnke et al. (1998); Martin et al. (2005); Pang et al. (2006); Rauf et al. (2000); Spencer (1973); Ueno (2006); Verjel-Manzano and Mejia-Florez (2000); Weintraub et al. (2017); (Koch and Waterhouse 2000)
Commercial	Scallion	*Allium cepa*	*aggregatum*	Amaryllidaceae	Mujica and Kroschel (2011)	Korytkowski (1982)
Commercial		*Allium cepa*	*cepa*	Amaryllidaceae		Scheffer et al. (2006)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Japanese bunching onion; Scallion; Welsh onion	*Allium fistulosum*		Amaryllidaceae	Rauf et al. (2000)	He et al. (2001); Hidalgo and Carballo (1991); Pang et al. (2006); Shepard et al. (1998); Shiao and Wu (2000); Ueno (2006); Wei et al. (2000)
Commercial	Garlic	*Allium sativum*		Amaryllidaceae	Rauf et al. (2000)	He et al. (2001); Hincapie et al. (1993); Pang et al. (2006); Saunders et al. (1998); Silva (1993); Wei et al. (2000)
Commercial		*Allium* sp.		Amaryllidaceae	Scheffer et al. (2001)	
Commercial	Celery	*Apium graveolens*		Apiaceae	Larrain and Munoz (1997); Rauf et al. (2000); Sivapragasam and Syed (1999)	Bahlai et al. (2006); de Clercq and Casteels (1992); He et al. (2001); He et al. (2002); Hidalgo and Carballo (1991); (Koch and Waterhouse 2000); Korytkowski (2014); Korytkowski (1982); Kuhnke et al. (1998); Martin et al. (2005); Pang et al. (2006); Romero-Zuniga et al. (1991); Shepard et al. (1998); Spencer (1973); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Wei et al. (2000); Weintraub (1999); Weintraub et al. (2017)
Commercial	Celery	*Apium graveolens*	*dulce*	Apiaceae	Mujica and Kroschel (2011)	He et al. (2001); Korytkowski (2014); Scheffer et al. (2006)
Commercial		*Apium* sp.		Apiaceae		Collins (1996)
Commercial	Carrot	*Daucus carota*		Apiaceae	Rauf et al. (2000)	Korytkowski (2014); Korytkowski (1982); Shepard et al. (1998)
Commercial		*Daucus carota*	*sativa*	Apiaceae		Korytkowski (2014)
Commercial	Carrot	*Daucus carota*	*sativus*	Apiaceae		Scheffer et al. (2006)
Commercial	Lettuce; Garden	*Lactuca sativa*		Asteraceae	Masetti et al. (2006); Mujica and Kroschel	Andersen et al. (2002); Andersen et al. (2008); Bahlai et al. (2006); de Clercq

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
	lettuce				(2011); Rauf et al. (2000); Takano et al. (2008)	and Casteels (1992); Echevarria et al. (1994); Godinho and Mexia (2000); He et al. (2002); Hincapie et al. (1993); (Koch and Waterhouse 2000); Korytkowski (2014); Korytkowski (1982); Kuhnke et al. (1998); Martin et al. (2005); Olivera and Bordat (1996); Weintraub et al. (2017); Parish et al. (2017); Salvo and Valladares (1997); Spencer (1973); Takano et al. (2008); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Weintraub et al. (2017)
Commercial		*Lactuca sativa*	*asparagina*	Asteraceae		Wei et al. (2000)
Commercial		*Lactuca sativa*	*capitata*	Asteraceae		Scheffer et al. (2006); Wei et al. (2000)
Commercial		*Lactuca sativa*	*intybeca*	Asteraceae		Shiao and Wu (2000)
Commercial	Indian nightshade	*Basella alba*		Basellaceae	Rauf et al. (2000)	He et al. (2001)
Commercial	Chinese cabbage; Petsai; Common yellow mustard	*Brassica campestris*		Brassicaceae	Rauf et al. (2000); Sivapragasam et al. (1999); Sivapragasam and Syed (1999)	Hidalgo and Carballo (1991); Pang et al. (2006); Romero-Zuniga et al. (1991)
Commercial		*Brassica campestris*	*pekinensis*	Brassicaceae	Mujica and Kroschel (2011)	He et al. (2001)
Commercial	Turnip	*Brassica campestris*	*rapa*	Brassicaceae	Mujica and Kroschel (2011)	Hincapie et al. (1993)
Commercial	Pak-choi cabbage; Petsai	*Brassica chinensis*		Brassicaceae	Rauf et al. (2000); Sivapragasam et al. (1999)	Andersen et al. (2008); Shepard et al. (1998)
Commercial	Leaf mustard; Mustard greens;	*Brassica juncea*		Brassicaceae	Rauf et al. (2000); Scheffer et al. (2001)	Andersen et al. (2008); Bahlai et al. (2006); He et al. (2001); Scheffer et al.

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
	Radish; Chinese mustard; Indian mustard; Mustard					(2006); Silva (1993); Wei et al. (2000)
Commercial	Gai lan; Cabbage; Broccoli; Caisin; Cabbage; Cauliflower; Field cabbage	*Brassica oleracea*		Brassicaceae	Rauf et al. (2000); Scheffer et al. (2001)	Bahlai et al. (2006); Echevarria et al. (1994); Korytkowski (2014); Korytkowski (1982); Pang et al. (2006); Saunders et al. (1998); Shepard et al. (1998)
Commercial	Kale	*Brassica oleracea*	*acephala*	Brassicaceae	Foba et al. (2015)	He et al. (2001)
Commercial	Cauliflower	*Brassica oleracea*	*botrytis*	Brassicaceae		He et al. (2001); Korytkowski (2014)
Commercial	Cabbage	*Brassica oleracea*	*capitata*	Brassicaceae	Mujica and Kroschel (2011)	He et al. (2001); Hincapie et al. (1993); Scheffer et al. (2006); Weintraub et al. (2017)
Commercial		*Brassica oleracea*	*caulorapa*	Brassicaceae		He et al. (2001)
Commercial		*Brassica oleracea*	*geminifera*	Brassicaceae		Weintraub et al. (2017)
Commercial	Broccoli	*Brassica oleracea*	*italica*	Brassicaceae		He et al. (2001); Scheffer et al. (2006); Shepard et al. (1998); Weintraub et al. (2017)
Commercial	Turnip; Rinsho; Chinese cabbage; Bokchoy	*Brassica rapa*		Brassicaceae	Rauf et al. (2000); Takano et al. (2008)	Collins (1996); Scheffer et al. (2006); Shepard et al. (1998); Valladares et al. (1999); Valladares et al. (2011)
Commercial	Pak choi; Pechay; Chinese	*Brassica rapa*	*chinensis*	Brassicaceae	Takano et al. (2008)	Andersen et al. (2008)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
	cabbage					
Commercial		*Brassica rapa*	*cultivar Ciaxin*	Brassicaceae		CABI (2019)
Commercial		*Brassica* sp.		Brassicaceae		Hincapie et al. (1993)
Commercial	Watercress	*Nasturtium officinale*		Brassicaceae	Rauf et al. (2000)	He et al. (2001); Weintraub et al. (2017)
Commercial		*Nasturtium* spp.		Brassicaceae		Valladares (1984)
Commercial	Radish; Chinese radish (Daikon); Wild radish; Garden radish; White radish	*Raphanus sativus*		Brassicaceae	Mujica and Kroschel (2011); Rauf et al. (2000)	He et al. (2001); Collins (1996); Echevarria et al. (1994); Shepard et al. (1998); Weintraub et al. (2017)
Commercial		*Raphanus sativus*	*sativus*	Brassicaceae		Scheffer et al. (2006)
Commercial	Sweet potato	*Ipomoea batatas*		Convolvulaceae	Rauf et al. (2000)	Shepard et al. (1998)
Commercial	Watermelon	*Citrullus lanatus*		Cucurbitaceae	Foba et al. (2015)	(Koch and Waterhouse 2000); Ripa et al. (1995)
Commercial	Cucumber	*Cucumis sativus*		Cucurbitaceae	Hammad and Nemer (2000); Masetti et al. (2006); Mujica and Kroschel (2011); Nakamura et al. (2013); Rauf et al. (2000)	Andersen et al. (2002); Weintraub et al. (2017); Hincapie et al. (1993); Kuhnke et al. (1998); Martin et al. (2005); Weintraub et al. (2017); Parish et al. (2017); Ripa et al. (1995); Shiao and Wu (2000); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Yabas et al. (1995)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial		*Cucumis* spp.		Cucurbitaceae		(Koch and Waterhouse 2000)
Commercial	Pumpkin; Winter squash; Squash	*Cucurbita maxima*		Cucurbitaceae	Foba et al. (2015); Mujica and Kroschel (2011)	Korytkowski (2014); Korytkowski (1982); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial	Courgette	*Cucurbita maxima*	*zapallito*	Cucurbitaceae	Salvo and Valladares (2002)	Salvo and Valladares (1997); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial	Butternut squash; Pumpkin; Crookneck squash	*Cucurbita moschata*		Cucurbitaceae	Foba et al. (2015)	Pang et al. (2006); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial		*Cucurbita moschata*	*medullosa*	Cucurbitaceae		Pang et al. (2006)
Commercial	Courgette; Zucchini; Pumpkin	*Cucurbita pepo*		Cucurbitaceae	Foba et al. (2015); Mujica and Kroschel (2011); Scheffer et al. (2001)	He et al. (2001); Korytkowski (1982); Olivera and Bordat (1996); Pang et al. (2006); Spencer (1973)
Commercial		*Cucurbita pepo*	*pepo*	Cucurbitaceae		Scheffer et al. (2006)
Commercial	Squash	*Cucurbita sp.*		Cucurbitaceae		Korytkowski (2014); Salvo and Valladares (1997); Valladares (1984); Valladares et al. (1996)
Commercial	Bitter gourd; Balsam pear	*Momordica charantia*		Cucurbitaceae	Foba et al. (2015)	Pang et al. (2006)
Commercial	Swartz Chayote	*Sechium edule*		Cucurbitaceae	Rauf et al. (2000)	He et al. (2001)
Commercial	Chickpea	*Cicer arietinum*		Fabaceae	Avalos et al. (2013)	
Commercial	Soybean; Soya bean	*Glycine max*		Fabaceae	Rauf et al. (2000)	He et al. (2001); Parish et al. (2017); Valladares (1984); Valladares et al. (1999)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Sweet Dolichos; Field bean	*Lablab purpureus*		Fabaceae	Foba et al. (2015)	
Commercial	Alfalfa; Lucerne	*Medicago sativa*		Fabaceae	Mujica and Kroschel (2011)	Echevarria et al. (1994); Gloria and Salas (1976); (Koch and Waterhouse 2000); Korytkowski (2014); Valladares (1984); Valladares et al. (1999); Valladares et al. (2011)
Commercial	Sieva; Lima bean; Lima bean	*Phaseolus lunatus*		Fabaceae	Sivapragasam et al. (1999)	
Commercial	Beans; Grean beans; Pole beans	*Phaseolus* sp.		Fabaceae		Scheffer et al. (2006)
Commercial	Green bean; French bean; Kidney bean; Bean; Common bean; Snap bean	*Phaseolus vulgaris*		Fabaceae	Foba et al. (2015); Larrain and Munoz (1997); Mujica and Kroschel (2011); Rauf et al. (2000); de Souza (1986); Hammad and Nemer (2000); Salvo and Valladares (2002)	Andersen et al. (2002); Andersen et al. (2008); Masetti et al. (2006); Olivera and Bordat (1996); Pang et al. (2006); Parish et al. (2017); Saunders et al. (1998); Verjel-Manzano and Mejia-Florez (2000); Echevarria et al. (1994); Godinho and Mexia (2000); Weintraub et al. (2017); He et al. (2001); He et al. (2002); Hidalgo and Carballo (1991); Hincapie et al. (1993); Iwasaki (2004b); Korytkowski (2014); Korytkowski (1982);

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
						Macdonald et al. (2003); Musundire et al. (2012); Okoth et al. (2014); Salvo and Valladares (1997); Shepard et al. (1998); Shiao and Wu (2000); Spencer (1973); Takano et al. (2008); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Weintraub et al. (2017); Yabas et al. (1995)
Commercial	Green pea	*Phaseolus vulgaris*	*vulgaris*	Fabaceae	Mujica and Kroschel (2011)	
Commercial	Garden pea; Pea; Snow pea; Sugar snap	*Pisum sativum*		Fabaceae	Sivapragasam et al. (1999); Foba et al. (2015); Larrain and Munoz (1997); Mujica and Kroschel (2011); Rauf et al. (2000); Scheffer et al. (2001); Sivapragasam and Syed (1999)	Andersen et al. (2002); Andersen et al. (2008); Bahlai et al. (2006); He et al. (2001); (Koch and Waterhouse 2000); Korytkowski (2014); Korytkowski (1982); Martin et al. (2005); Musundire et al. (2012); Okoth et al. (2014); Pang et al. (2006); Ripa et al. (1995); Salvo and Valladares (1997); Saunders et al. (1998); Silva (1993); Spencer (1973); Takano et al. (2008); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Wei et al. (2000); Weintraub et al. (2017); Yabas et al. (1995); Kox et al. (2005)
Commercial		*Pisum sativum*	*macrocarpenser*	Fabaceae		He et al. (2001)
Commercial	Snow pea	*Pisum* sp.		Fabaceae		Collins (1996); Scheffer et al. (2006); Shepard et al. (1998)
Commercial	Broad bean; Faba bean; Fava bean	*Vicia faba*		Fabaceae	Mujica and Kroschel (2011); Rauf et al. (2000); Salvo and Valladares (2002); Scheffer et al. (2001)	Blanchard (1938); Collins (1996); Echevarria et al. (1994); Gallegos 2000; Weintraub et al. (2017); He et al. (2001); He et al. (2002); (Koch and Waterhouse 2000); Korytkowski (1982); Musundire et al. 2011; Musundire et al. (2012); Noujeim et al. 2013; Okoth et al. (2014);

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
						Salvo and Valladares (1997); Saunders et al. (1998); Silva (1993); Takano et al. (2008); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Wei et al. (2000); Yabas et al. (1995)
Commercial		*Vicia* sp.		Fabaceae		Spencer (1973)
Commercial	Red bean; Black eyed pea; Cowpea	*Vigna sinensis*		Fabaceae	Rauf et al. (2000)	He et al. (2001); Shepard et al. (1998); Wei et al. (2000)
Commercial	Walp Cowpea; Yard-long bean; Long bean; Snakebean; Cowpea	*Vigna unguiculata*		Fabaceae	Foba et al. (2015); Rauf et al. (2000)	Hincapie et al. (1993)
Commercial		*Vigna unguiculata*	*unguiculata*	Fabaceae		Takano et al. (2008)
Commercial	Basil; Sweet basil; Thai basil	*Ocimum basilicum*		Lamiaceae	Mujica and Kroschel (2011); Rauf et al. (2000)	
Commercial	Okra	*Abelmoschus esculentus*		Malvaceae	Foba et al. (2015)	
Commercial	Pepper; Sweet pepper; Green pepper; Chili; Capsicum	*Capsicum annuum*		Solanaceae	Sivapragasam et al. (1999); Mujica and Kroschel (2011); Rauf et al. (2000); Sivapragasam and Syed (1999)	Calabretta et al. 1995; (Koch and Waterhouse 2000); Pang et al. (2006); Salvo and Valladares (1997); Saunders et al. (1998); Shepard et al. (1998); Shiao and Wu (2000); Spencer (1973); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Weintraub et al. (2017)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Yellow chili	*Capsicum baccatum*		Solanaceae	Mujica and Kroschel (2011)	
Commercial	Sweet pepper; Pepper; Chilli	*Capsicum* sp.		Solanaceae	Foba et al. (2015)	Romero-Zuniga et al. (1991)
Commercial	Tomato	*Lycopersicon esculentum*		Solanaceae	Mujica and Kroschel (2011); Rauf et al. (2000); Sivapragasam et al. (1999); Sivapragasam and Syed (1999)	Korytkowski (2014); Kox et al. (2005); He et al. (2001); (Koch and Waterhouse 2000); Okoth et al. (2014); Olivera and Bordat (1996); Osorio et al. 1983; Salvo and Valladares (1997); Shepard et al. (1998); Spencer (1973); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Collins (1996);
Commercial	Tomato	*Lycopersicon esculentum*	*esculentum*	Solanaceae		Scheffer et al. (2006)
Commercial	Tomato	*Solanum lycopersicum*		Solanaceae	Foba et al. (2015)	Andersen et al. (2008); Andersen et al. (2002); de Clercq and Casteels (1992); Godinho and Mexia (2000); Hanafi (2005); Hidalgo and Carballo (1991); Korytkowski (1982); Macdonald et al. (2003); Moura et al. (2014); Musundire et al. (2012); Weintraub et al. (2017); Pang et al. (2006); Parish et al. (2017); Saunders et al. (1998); Weintraub et al. (2017)
Commercial	Brinjal eggplant;	*Solanum*		Solanaceae	Foba et al. (2015); Rauf et	He et al. (2001); Korytkowski (1982);

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
	Eggplant; common eggplant; Pickling eggplant	*melongena*			al. (2000)	Salvo and Valladares (1997); Saunders et al. (1998); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Commercial		*Solanum sp.*		Solanaceae	Scheffer et al. (2001)	Andrade et al. (1989)
Commercial	Potato	*Solanum tuberosum*		Solanaceae	de Souza (1986); Foba et al. (2015); Larrain and Munoz (1997); Mujica and Kroschel (2011); Nino et al. (2009); Rauf et al. (2000); Salvo and Valladares (2002); Scheffer et al. (2001); Sivapragasam and Syed (1999)	Andersen et al. (2002); Andersen et al. (2008); Bahlai et al. (2006); Gallegos 2000; He et al. (2001); Hidalgo and Carballo (1991); Iwasaki (2004a); (Koch and Waterhouse 2000); Korytkowski (2014); Korytkowski (1982); Macdonald et al. (2003); Maharjan et al. 2014; Martin et al. (2005); Romero-Zuniga et al. (1991); Salvo and Valladares (1997); Saunders et al. (1998); Shepard et al. (1998); Spencer (1973); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011); Wijesekara (2010); Yabas et al. (1995)
Ornamental	Pot marigold	*Calendula officinalis*		Asteraceae	Salvo and Valladares (2002)	He et al. (2001); Korytkowski (1982); Salvo and Valladares (1997); Valladares (1984); Valladares et al. (1996); Valladares et al. (1999); Valladares et al. (2011)
Ornamental		*Calendula* sp.		Asteraceae		Weintraub et al. (2017)
Ornamental	Chinese aster	*Callistephus chinensis*		Asteraceae	Salvo and Valladares (2002)	de Clercq and Casteels (1992); Salvo and Valladares (1997); Valladares et al. (1999); Valladares et al. (2011); Wei et al. (2000); Valladares et al. (1996)
Ornamental	Garland chrysanthemum	*Chrysanthemum coronarium*		Asteraceae		Wei et al. (2000)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Ornamental	Garland chrysanthemum	*Chrysanthemum coronarium*	*coronarium*	Asteraceae	Tran (2009)	
Ornamental	Garland chrysanthemum; Chrysanthemum	*Chrysanthemum* sp.		Asteraceae	Scheffer et al. (2001)	Collins (1996); Hincapie et al. (1993); Pang et al. (2006); Salvo and Valladares (1997); Scheffer et al. (2006); Weintraub et al. (2017); Anderson et al. (2008)
Ornamental	Chrysanthemum	*Chrysanthemum* spp.		Asteraceae	Rauf et al. (2000)	
Ornamental	Dahlia	*Dahlia* sp.		Asteraceae	Rauf et al. (2000)	Collins (1996); Weintraub et al. (2017)
Ornamental	Gerbera; Barberton daisy; Transvaal daisy	*Gerbera jamesonii*		Asteraceae	Rauf et al. (2000)	Wei et al. (2000)
Ornamental		*Gerbera* sp.		Asteraceae		Weintraub et al. (2017)
Ornamental	Marigold; Aztec marigold	*Tagetes erecta*		Asteraceae	Mujica and Kroschel (2011)	Wei et al. (2000)
Ornamental		*Tagetes erecta*	*hybrids*	Asteraceae		de Clercq and Casteels (1992)
Ornamental	Marigold	*Tagetes* sp.		Asteraceae		Weintraub et al. (2017); Salvo and Valladares (1997); Valladares (1984); Valladares et al. (1996)
Ornamental		*Eustoma* sp.		Gentianaceae	Nakamura et al. (2013)	Weintraub et al. (2017)
Ornamental	`Swai tanah'	*Nasturtium indicum*		Tropaeolaceae	Rauf et al. (2000)	
Ornamental	Garden nasturtium	*Tropaeolum majus*		Tropaeolaceae	Salvo and Valladares (2002)	He et al. (2001); Salvo and Valladares (1997); Silva (1993); Valladares (1984)
Ornamental	Nasturtium	*Tropaeolum* sp.		Tropaeolaceae	Spencer (1983)	
Non cultivated	Amaranth; Bayam	*Amaranthus* sp.*		Amaranthaceae	Rauf et al. (2000); Sivapragasam et al. (1999)	Hidalgo and Carballo (1991); Romero-Zuniga et al. (1991); Shepard et al. (1998); Valladares et al. (1999); Valladares et al. (2011); Weintraub et al.

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
						(2017)
Non cultivated	Goosefoot; Lamb's quarters; Fat hen	*Chenopodium album**		Amaranthaceae	Salvo and Valladares (2002)	He et al. (2001); Silva (1993)
Non cultivated	Amaranth	*Deeringia amaranthoides**		Amaranthaceae	Rauf et al. (2000)	
Non cultivated	Penniwort	*Hydrocotyle* sp.*		Araliaceae	Salvo and Valladares (2002)	Salvo and Valladares (1997)
Non cultivated	Chick weed; Goatweed	*Ageratum conyzoides**		Asteraceae	Rauf et al. (2000)	
Non cultivated	Common burdock	*Arctium minus**		Asteraceae	Salvo and Valladares (2002)	Salvo and Valladares (1997); Valladares et al. (1999); Valladares et al. (2011)
Non cultivated	Sow thistle	*Emilia sonchifolia**		Asteraceae	Rauf et al. (2000); Scheffer et al. (2001)	Hincapie et al. (1993); Scheffer et al. (2006)
Non cultivated	American burnweed; Pilewort; Fireweed	*Erechtites hieracifolia** (currently under quarantine in Australia (CABI 2019))		Asteraceae	Rauf et al. (2000)	Romero-Zuniga et al. (1991)
Non cultivated	Indian lettuce	*Lactuca indica*		Asteraceae	Rauf et al. (2000); Sivapragasam et al. (1999)	
Non cultivated	Cinderella weed; Nodeweed	*Synedrella nodiflora**		Asteraceae	Rauf et al. (2000)	
Non	Wild cucumber	*Melothria indica*		Cucurbitaceae	Rauf et al. (2000)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
cultivated						
Non cultivated	Cutleaf ground cherry; Mullaca; Wild gooseberry	*Physalis angulata**		Solanaceae	Rauf et al. (2000)	Hidalgo and Carballo (1991)
Non cultivated	American black nightshade	*Solanum americanum*		Solanaceae	Rauf et al. (2000)	

Commercial, ornamental, and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*) has been provided within the scientific literature

This includes all records where:

(1) Adults were reared from field collected plant material infested with larvae, but no subsequent identification via morphology or molecular diagnostics was reported; or

(2) Laboratory feeding and oviposition preference experiments were conducted that did not confirm lifecycle completion; or

(3) Identification via morphology or molecular diagnostics was reported, but collection methods in the field were not reported. Lifestyle completion cannot be confirmed in these instances due to the potential use of non-specific collection methods (e.g. sticky traps or sweep nets); or

(4) Laboratory cultures confirmed lifecycle completion, but evidence of lifecycle completion in the field was not provided; or

(5) Only common names of plant were given, but the scientific name could be confidently inferred; or

(6) The record reports presence of *L. huidobrensis* on imported material detected at the border.

Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included here only if there are other records for species within this genus that met one of these conditions outlined above. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records for that same species that meet one of the conditions outlined above. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. For all non-cultivated plants, and asterisk after the scientific name indicates that the Atlas of Living Australia reports this species present within Australia (Atlas of Living Australia 2020). Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 13, while unverified records are included in Table 15. *Liriomyza huidobrensis* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 14 Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the serpentine leafminer (Liriomyza huidobrensis) has been provided within the scientific literature

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Quinoa	*Chenopodium quinoa*		Amaranthaceae	CABI (2019); Korytkowski (1982)
Commercial	Quinua	*Chenopodium quinoa*	Amarilla de Maranganí	Amaranthaceae	Yabar et al. (2002)
Commercial	Quinua	*Chenopodium quinoa*	Blanca de Junín	Amaranthaceae	Yabar et al. (2002)
Commercial		*Allium chinense*		Amaryllidaceae	He et al. (2001)
Commercial	Chive	*Allium schoenoprasum*		Amaryllidaceae	Spencer (1973)
Commercial		*Allium tuberosum*	*rottler*	Amaryllidaceae	He et al. (2001)
Commercial		*Bupleurum green* *(*most likely a *Bupleurum rotundifolium* cultivar)		Apiaceae	He et al. (2001)
Commercial		*Bupleurum* sp.		Apiaceae	Andersen and Hofsvang (2010)
Commercial	Coriander	*Coriandrum sativum*		Apiaceae	Weintraub et al. (2017); He et al. (2001); Koch and Waterhouse 2000
Commercial		*Daucus sativa*	*sativus*	Apiaceae	Wei et al. (2000); Hidalgo and Carballo (1991)
Commercial	Giant taro	*Alocasia macrorrhiza*		Araceae	Wei et al. (2000)
Commercial		*Colocasia esculenta*		Araceae	He et al. (2001)
Commercial	Carthamum; Safflower	*Carthamus tinctorius*		Asteraceae	He et al. (2001); Sunderlands et al. (1992)
Commercial		*Carthamus* sp.		Asteraceae	Andersen and Hofsvang (2010)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial		*Cichorium endivia*		Asteraceae	de Goffau 1991
Commercial	Endive	*Cichorium* sp.		Asteraceae	Weintraub et al. (2017); Salvo and Valladares (1997)
Commercial		*Crassocephalum rubens*		Asteraceae	Shiao and Wu (2000)
Commercial	Artichoke	*Cynara cardunculus*	*scolymus*	Asteraceae	Larrain et al. 2013
Commercial		*Cynara scolymus*		Asteraceae	Korytkowski (2014)
Commercial		*Cynara scolymus*	*scolymus*	Asteraceae	Korytkowski (1982)
Commercial		*Basella rubra*		Basellaceae	He et al. (2001)
Commercial	Gai lan; Asian broccoli	*Brassica alboglabra*		Brassicaceae	Bahlai et al. (2006); Martin et al. (2005)
Commercial		*Brassica botrytis*		Brassicaceae	Wei et al. (2000)
Commercial		*Brassica campestris spp.*		Brassicaceae	He et al. (2001)
Commercial		*Brassica chinese*		Brassicaceae	Wei et al. (2000)
Commercial		*Brassica napus*		Brassicaceae	Wei et al. (2000)
Commercial		*Brassica pekinensis*		Brassicaceae	Wei et al. (2000)
Commercial		*Brassica pekinensis pekinensis*		Brassicaceae	He et al. (2002)
Commercial	Quinoa	*Chenopodium quinoa*		Chenopodiaceae	Korytkowski (2014)
Commercial		*Spinacea oleracea*		Chenopodiaceae	Korytkowski (2014)
Commercial		*Ipomoea aquatica*		Convolvulaceae	He et al. (2001)
Commercial	Wax gourd; White goard; Chinese melon; China squash; Ash gourd	*Benincasa hispida*		Cucurbitaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Watermelon	*Citrullus vulgaris*		Cucurbitaceae	Korytkowski (2014); Korytkowski (1982)
Commercial	Melon; Cantaloupe; Muskmelon; Honey melon; Cassaba melon	*Cucumis melo*		Cucurbitaceae	EPPO (2020); CABI (2019); Valladares et al. (2011) ; Guimarães et al. (2009); Koch and Waterhouse 2000; Valladares et al. (1999); Valladares et al. (1996); Valladares (1984); Spencer (1973);
Commercial	Bottle gourd	*Lagenaria siceraria*		Cucurbitaceae	Kox et al. (2005)
Commercial		*Lagenaria* sp.		Cucurbitaceae	Shiao and Wu (2000)
Commercial	Angled luffa; Sing kwa; Towelsponge; Angled luffa	*Luffa acutangula*		Cucurbitaceae	He et al. (2002)
Commercial	Towel gourd	*Luffa cylindrica*		Cucurbitaceae	Shiao and Wu (2000); Valladares et al. (1996)
Commercial	Creeper bean	*Dolichos lablab*		Fabaceae	He et al. (2001)
Commercial		*Lotus* sp.		Fabaceae	Collins (1996)
Commercial	White clover	*Trifolium repens*		Fabaceae	He et al. (2001); Wei et al. (2000); Andrade et al. (1989)
Commercial		*Trigonella* sp.		Fabaceae	Collins (1996)
Commercial		*Vicia fava*		Fabaceae	Korytkowski (2014)
Commercial		*Vicia sativa*		Fabaceae	Weintraub et al. (2017); He et al. (2001)
Commercial	Mungbean; Green gram	*Vigna radiata*		Fabaceae	Andersen et al. (2002)
Commercial	Spearmint	*Mentha spicata*		Lamiaceae	Wei et al. (2000)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial		*Linum usitatissimum*		Linaceae	Spencer (1973)
Commercial	Barley	*Hordeum vulgare*		Poaceae	He et al. (2001)
Commercial		*Triticum aestivum*		Poaceae	He et al. (2001)
Commercial		*Zea mays*		Poaceae	He et al. (2001); Salvo and Valladares (1997)
Commercial	Wild rice	*Zizania latifolia*		Poaceae	Wei et al. (2000)
Commercial	Bell pepper	*Capsicum frutescens*		Solanaceae	He et al. (2002); He et al. (2001)
Commercial		*Lycium chinense*		Solanaceae	He et al. (2001)
Commercial		*Nicotiana* sp.		Solanaceae	Korytkowski (2014); Korytkowski (1982)
Commercial		*Nicotiana tabacum*		Solanaceae	Korytkowski (2014); He et al. (2001); Korytkowski (1982)
Commercial		*Solanum muricatum*		Solanaceae	Larrain 2002
Ornamental		*Thurnbergia alata*		Acanthaceae	Andersen and Hofsvang (2010)
Ornamental	Cock's comb	*Celosia argentea*		Amaranthaceae	He et al. (2001)
Ornamental		*Celosia argentea*	*cristata*	Amaranthaceae	He et al. (2001)
Ornamental		*Centella asiatica*		Apiaceae	He et al. (2001)
Ornamental		*Bupleurum griffithii*		Apiaceae	Andersen and Hofsvang (2010)
Ornamental		*Eryngium* sp.		Apiaceae	Andersen and Hofsvang (2010)
Ornamental		*Eryngium alpinum*		Apiaceae	Andersen and Hofsvang (2010)
Ornamental		*Eryngium foetidum*		Apiaceae	Andersen and Hofsvang (2010)
Ornamental		*Hydrocotyle umbellata*		Apiaceae	Korytkowski (2014)
Ornamental	Java waterdropwort	*Oenanthe javanica*		Apiaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Catharanthus roseus*		Apocynaceae	He et al. (2001)
Ornamental		*Chionodoxa luciliae*		Asparagaceae	He et al. (2001)
Ornamental		*Hemerocallis fulva*		Asphodelaceae	He et al. (2001)
Ornamental	Aster	*Aster* sp.		Asteraceae	Pang et al. (2006); Kox et al. (2005); Collins (1996)
Ornamental		*Argyranthemum frutescens*		Asteraceae	EPPO (2020); Andersen and Hofsvang (2010)
Ornamental		*Argyranthemum* sp.		Asteraceae	Weintraub et al. (2017); Andersen and Hofsvang (2010)
Ornamental	Daisy	*Bellis perennis*		Asteraceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Salvo and Valladares (1997); Valladares (1984)
Ornamental		*Callendula officinallis*		Asteraceae	Korytkowski (2014)
Ornamental		*Centaurea cyanus*		Asteraceae	He et al. (2001)
Ornamental		*Centaurea* sp.		Asteraceae	Andersen and Hofsvang (2010)
Ornamental		*Chrysanthemum fructescens*		Asteraceae	de Clercq and Casteels (1992)
Ornamental	Oxeye daisy	*Chrysanthemum leucanthemum*		Asteraceae	Valladares et al. (2011); Valladares et al. (1999); Valladares et al. (1996); Valladares (1984);
Ornamental	Chrysanthemum; Florists' chrysanthemum	*Chrysanthemum morifolium*		Asteraceae	Valladares et al. (2011); Wei et al. (2000); Valladares et al. (1999); Valladares et al. (1996) de Clercq and Casteels (1992); Andrade et al. (1989); Valladares (1984)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Cineraria cruenta*		Asteraceae	He et al. (2001)
Ornamental		*Cineraria* spp.		Asteraceae	Valladares (1984); Blanchard (1926)
Ornamental		*Conoclinium coelestinum*		Asteraceae	He et al. (2001)
Ornamental		*Cosmos bipinnatus*		Asteraceae	He et al. (2001)
Ornamental		*Craspedia globosa*		Asteraceae	He et al. (2001)
Ornamental	Dahlia	*Dahlia pinnata*		Asteraceae	He et al. (2001); Salvo and Valladares (1997); de Clercq and Casteels (1992);
Ornamental	Dahlia	*Dahlia variabilis*		Asteraceae	Weintraub et al. (2017); Valladares et al. (2011); Valladares et al. (1999); Valladares et al. (1996); Valladares (1984);
Ornamental		*Dendranthema morifolium*		Asteraceae	Cuthbertson et al. (2013)
Ornamental		*Gaillardia pulchella*		Asteraceae	He et al. (2001)
Ornamental		*Helianthus annus*		Asteraceae	Korytkowski (2014)
Ornamental		*Helianthus* sp.		Asteraceae	Weintraub et al. (2017); Collins (1996);
Ornamental	Indian aster	*Kalimeris indica*		Asteraceae	Shiao and Wu (2000)
Ornamental		*Leucanthemum* sp.		Asteraceae	Andersen and Hofsvang (2010); Kox et al. (2005)
Ornamental		*Osteospermum* sp.		Asteraceae	Weintraub et al. (2017); Andersen and Hofsvang (2010)
Ornamental		*Pyrethrum cinerariifolium*		Asteraceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental	Goldenrod	*Solidago sp.*		Asteraceae	Andersen and Hofsvang (2010); He et al. (2001)
Ornamental	Marigold; French marigold	*Tagetes patula*		Asteraceae	Wei et al. (2000)
Ornamental		*Tagetes tenuitolia*		Asteraceae	Valladares et al. (2011); Valladares et al. (1999); Salvo and Valladares (1997); Valladares (1984);
Ornamental		*Tanacetum parthenium*		Asteraceae	EPPO (2020); He et al. (2001)
Ornamental		*Zinnia elegans*		Asteraceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Valladares (1984)
Ornamental		*Zinnia augustifolia*		Asteraceae	Andersen and Hofsvang (2010)
Ornamental		*Zinnia* sp.		Asteraceae	Kox et al. (2005)
Ornamental		*Impatiens balsamina*		Balsaminaceae	He et al. (2001)
Ornamental		*Impatiens caeruleum*		Balsaminaceae	He et al. (2001)
Ornamental		*Eruca* sp.		Brassicaceae	Andersen and Hofsvang (2010)
Ornamental		*Eruca vesicaria*		Brassicaceae	Andersen and Hofsvang (2010)
Ornamental		*Lobularia maritima*		Brassicaceae	He et al. (2001)
Ornamental		*Matthiola incana*		Brassicaceae	He et al. (2001)
Ornamental		*Matthiola sp.*		Brassicaceae	He et al. (2001)
Ornamental		*Calceolaria crenatiflora*		Calceolariaceae	He et al. (2001)
Ornamental		*Campanula medium*		Campanulaceae	He et al. (2001)
Ornamental		*Platycodon grandifloras*		Campanulaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Trachelium* sp.		Campanulaceae	Andersen and Hofsvang (2010)
Ornamental		*Dianthus barbatus*		Caryophyllaceae	He et al. (2001)
Ornamental	Carnation	*Dianthus caryophyllus*		Caryophyllaceae	EPPO (2020); Valladares et al. (2011); He et al. (2001); (Koch and Waterhouse 2000); Valladares et al. (1999); Shepard et al. (1998); Salvo and Valladares (1997); Valladares (1984)
Ornamental	Indian pink; China pink	*Dianthus chinensis*		Caryophyllaceae	He et al. (2001); de Clercq and Casteels (1992)
Ornamental		*Dianthus hybridus*		Caryophyllaceae	He et al. (2001)
Ornamental		*Dianthus* sp.		Caryophyllaceae	Andersen and Hofsvang (2010); Kox et al. (2005); Collins (1996);
Ornamental	Showy baby's breath	*Gypsophila elegans*		Caryophyllaceae	Valladares et al. (2011); He et al. (2002); He et al. (2001); Valladares et al. (1999)
Ornamental		*Gypsophila muralis*		Caryophyllaceae	Kox et al. (2005)
Ornamental	Gypsophila; Baby's breath	*Gypsophila paniculata*		Caryophyllaceae	Cure and Cantor 2003; He et al. (2001); Wei et al. (2000)
Ornamental	Gypsophila	*Gypsophila* sp.		Caryophyllaceae	Weintraub et al. (2017); Collins (1996); Andrade et al. (1989); Sunderlands et al. (1992)
Ornamental		*Gypsophila perfecta*		Caryophyllaceae	Andersen and Hofsvang (2010)
Ornamental		*Pharbitis purpurea*		Convolvulaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Euphorbia marginata*		Euphorbiaceae	He et al. (2001)
Ornamental		*Lathyrus latifolius*		Fabaceae	Valladares et al. (2011); Valladares et al. (1999); Valladares (1984)
Ornamental		*Lathyrus odoratus*		Fabaceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Valladares (1984)
Ornamental		*Lupin* sp.		Fabaceae	Collins (1996)
Ornamental		*Lupinus rassel*	*Russell hybrid*	Fabaceae	He et al. (2001)
Ornamental		*Lupinus* sp.		Fabaceae	Kox et al. (2005)
Ornamental	Fenugreek	*Trigonella foenum-graecum*		Fabaceae	Andersen and Hofsvang (2010)
Ornamental	Exacum	*Exacum* sp.		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Exacum affine*		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Eustoma grandiflorum*		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Eustoma russellianum*		Gentianaceae	He et al. (2001); Wei et al. (2000)
Ornamental		*Lisianthus* sp.		Gentianaceae	Andersen and Hofsvang (2010); Weintraub et al. (2017); Collins (1996)
Ornamental		*Lisianthus russelianus*		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Scaevola aemula*		Goodeniaceae	Andersen and Hofsvang (2010)
Ornamental		*Hydrangea macrophylla*		Hydrangeaceae	He et al. (2001)
Ornamental		*Freesia refracta*		Iridaceae	He et al. (2001)
Ornamental	Sword lily	*Gladiolus hybridus*		Iridaceae	He et al. (2001); Wei et al. (2000)
Ornamental		*Lamium* sp.		Lamiaceae	Andersen and Hofsvang (2010)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Moluccella laevis*		Lamiaceae	He et al. (2001)
Ornamental		*Moluccella* sp.		Lamiaceae	Andersen and Hofsvang (2010)
Ornamental		*Salvia splendens*		Lamiaceae	He et al. (2001)
Ornamental		*Lilium davidii*		Liliaceae	He et al. (2001)
Ornamental		*Lilium longiflorum*		Liliaceae	He et al. (2001)
Ornamental		*Torenia* sp.		Linderniaceae	Kox et al. (2005)
Ornamental		*Althaea rosea*		Malvaceae	Korytkowski (2014)
Ornamental		*Hibiscus trionum*		Malvaceae	He et al. (2001)
Ornamental		*Godetia amoena*		Onagraceae	He et al. (2001)
Ornamental		*Oenothera rosea*		Onagroideae	He et al. (2001)
Ornamental		*Papaver rhoeas*		Papaveraceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Salvo and Valladares (1997); Valladares (1984)
Ornamental	Snapdragon	*Antirrhinum majus*		Plantaginaceae	Martin et al. (2005)
Ornamental		*Limonium latifolium*		Plumbaginaceae	He et al. (2001)
Ornamental		*Limonium tataricum*		Plumbaginaceae	He et al. (2001)
Ornamental		*Myosotis sylvatica*		Plumbaginaceae	He et al. (2001)
Ornamental		*Lagurus ovatus*		Poaceae	He et al. (2001)
Ornamental	Phlox	*Phlox drummondii*		Polemoniaceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Salvo and Valladares (1997); de Clercq and Casteels (1992); Valladares (1984)
Ornamental		*Primula acaulis*		Primulaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Primula obconica*		Primulaceae	He et al. (2001); Salvo and Valladares (1997); de Clercq and Casteels (1992)
Ornamental		*Delphinium grandiflorum*		Ranunculaceae	He et al. (2001)
Ornamental		*Delphinium* sp.		Ranunculaceae	He et al. (2001)
Ornamental		*Nigella damascena*		Ranunculaceae	He et al. (2001)
Ornamental		*Ranunculus asiaticus*		Ranunculaceae	Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Valladares et al. (1996)
Ornamental		*Ranunculus sceleratus*		Ranunculaceae	Wei et al. (2000)
Ornamental	Buttercup	*Ranunculus* sp.		Ranunculaceae	Collins (1996)
Ornamental		*Ranunculus viridis*		Ranunculaceae	He et al. (2001)
Ornamental		*Rosa chinese*		Rosaceae	Wei et al. (2000)
Ornamental		*Rosa* sp.		Rosaceae	Andersen and Hofsvang (2010)
Ornamental		*Diascia* sp.		Scrophulariaceae	Andersen and Hofsvang (2010); Saethre (1996)
Ornamental		*Nicotiana alata*		Solanaceae	de Clercq and Casteels (1992)
Ornamental		*Petunia hybida*		Solanaceae	He et al. (2001)
Ornamental	Petunia	*Petunia* sp.		Solanaceae	Weintraub et al. (2017); Valladares et al. (2011); Valladares et al. (1999); Salvo and Valladares (1997); Collins (1996); Hincapie et al. (1993)
Ornamental		*Petunia* spp.		Solanaceae	Valladares (1984)
Ornamental		*Petunia* spp.	*hybrids*	Solanaceae	de Clercq and Casteels (1992)
Ornamental	Vervain	*Verbena officinalis*		Verbenaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Verbena* sp.		Verbenaceae	Weintraub et al. (2017); Collins (1996); de Clercq and Casteels (1992)
Ornamental		*Viola tricolor*		Violaceae	Valladares et al. (2011); Valladares et al. (1999); Salvo and Valladares (1997); Valladares (1984)
Ornamental		*Viola tricolor*	*vannortensis*	Violaceae	He et al. (2001)
Ornamental		*Viola yedoensis*		Violaceae	He et al. (2001)
Non cultivated		*Sagittaria sagittifolia**		Alismataceae	He et al. (2001)
Non cultivated		*Alstomeria* sp.*		Alstroemeriaceae	Collins (1996)
Non cultivated		*Alstroemeria aurea**		Alstroemeriaceae	He et al. (2001)
Non cultivated		*Amaranthus caudatus**		Amaranthaceae	He et al. (2001)
Non cultivated		*Amaranthus lividus**		Amaranthaceae	He et al. (2001)
Non cultivated		*Amaranthus manostanus*		Amaranthaceae	He et al. (2001)
Non cultivated	Rough pigweed	*Amaranthus retroflexus**		Amaranthaceae	He et al. (2001); Hincapie et al. (1993)
Non cultivated		*Amaranthus viridis**		Amaranthaceae	Shiao and Wu (2000); Verjel-Manzano and Mejia-Florez (2000); Spencer (1973)
Non cultivated		*Chenopodium ambrosioides** (Korytkowski (2014) reports as *C. abrossoides*)		Amaranthaceae	Korytkowski (2014); Valladares et al. (2011); He et al. (2001); Valladares et al. (1999); Valladares (1984); Korytkowski (1982)
Non cultivated		*Chenopodium abrossoides*		Amaranthaceae	
Non cultivated		*Chenopodium hirsutum*		Amaranthaceae	Korytkowski (2014)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Non cultivated		*Chenopodium murale**		Amaranthaceae	Korytkowski (1982)
Non cultivated		*Chenopodium paniculatum*		Amaranthaceae	Andrade et al. (1989)
Non cultivated		*Gomphrena globose**		Amaranthaceae	He et al. (2001)
Non cultivated		*Iresine herbstii**		Amaranthaceae	Wei et al. (2000)
Non cultivated		*Unknown Amaranthaceae*		Amaranthaceae	Salvo and Valladares (1997)
Non cultivated		*Hydrocotyle umbellate**		Araliaceae	Hincapie et al. (1993); Hidalgo and Carballo (1991); Korytkowski (1982)
Non cultivated		*Arctium lappa**		Asteraceae	Scheffer et al. (2006); Silva (1993)
Non cultivated		*Artemisia annua*		Asteraceae	He et al. (2001)
Non cultivated	Black jack; Spanish needle; hairy beggarticks	*Bidens pilosa**		Asteraceae	Weintraub et al. (2017); Pang et al. (2006); Hidalgo and Carballo (1991); Romero-Zuniga et al. (1991)
Non cultivated	Bidens	*Bidens sp.**		Asteraceae	Salvo and Valladares (1997)
Non cultivated		*Carduus crispus*		Asteraceae	He et al. (2001)
Non cultivated		*Conyza canadensis**		Asteraceae	Wei et al. (2000)
Non cultivated		*Conyza* sp.*		Asteraceae	Valladares et al. (2011); Valladares et al. (1999); Salvo and Valladares (1997)
Non cultivated		*Crepis pulchra*		Asteraceae	Salvo and Valladares (1997)
Non cultivated		*Dichrocephala auriculata*		Asteraceae	He et al. (2001)
Non cultivated		*Echinops ritro*		Asteraceae	He et al. (2001)
Non cultivated		*Erigeron breviscapus*		Asteraceae	He et al. (2001)
Non cultivated		*Galinsoga caracasana*		Asteraceae	Andrade et al. (1989); Spencer (1983); Spencer (1973)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Non cultivated	Quickweed; Shaggy soldier	*Galinsoga ciliata*		Asteraceae	Hidalgo and Carballo (1991); Romero-Zuniga et al. (1991); Andrade et al. (1989)
Non cultivated		*Galinsoga parviflora**		Asteraceae	Wei et al. (2000); Hincapie et al. (1993)
Non cultivated		*Helichrysum bracteatum**		Asteraceae	Wei et al. (2000)
Non cultivated		*Helipterum roseum**		Asteraceae	He et al. (2001)
Non cultivated		*Hemistepta lyrate**		Asteraceae	He et al. (2001)
Non cultivated	Spiny sow thistle; Prickly sow thistle	*Sonchus asper**		Asteraceae	He et al. (2001); Hincapie et al. (1993)
Non cultivated		*Sonchus brachyotus*		Asteraceae	He et al. (2001)
Non cultivated		*Sonchus lunatus*		Asteraceae	Andrade et al. (1989)
Non cultivated	Common sow thistle	*Sonchus oleraceus**		Asteraceae	Weintraub et al. (2017); He et al. (2001); Hidalgo and Carballo (1991); Romero-Zuniga et al. (1991)
Non cultivated		*Sonchus* sp.*		Asteraceae	Korytkowski (2014); Korytkowski (1982)
Non cultivated		*Taraxacum mongolicum*		Asteraceae	He et al. (2001)
Non cultivated		*Barbarea* sp.*		Brassicaceae	Shiao and Wu (2000)
Non cultivated		*Capsella bursa-pastoris**		Brassicaceae	Weintraub et al. (2017); He et al. (2001); Andrade et al. (1989)
Non cultivated		*Cardamine hirsute**		Brassicaceae	He et al. (2001)
Non cultivated		*Rorippa indica*		Brassicaceae	Scheffer et al. (2006); He et al. (2001)
Non cultivated		*Rorippa montana*		Brassicaceae	He et al. (2001)
Non cultivated		*Rorippa palustris**		Brassicaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Non cultivated		*Malachium aguaticum*		Caryophyllaceae	He et al. (2001)
Non cultivated		*Silene gallica**		Caryophyllaceae	Andrade et al. (1989)
Non cultivated		*Stellaria media**		Caryophyllaceae	Mujica (2007); He et al. (2001); Wei et al. (2000)
Non cultivated		*Stellaria yunnansis*		Caryophyllaceae	He et al. (2001)
Non cultivated		*Calystegia hederacea*		Convolvulaceae	He et al. (2001)
Non cultivated		*Calystegia sepium**		Convolvulaceae	Wei et al. (2000)
Non cultivated	Castor bean; Castor oil bush	*Ricinus communis**		Euphorbiaceae	He et al. (2001)
Non cultivated	Black medic	*Medicago lupulina**		Fabaceae	He et al. (2001)
Non cultivated		*Medicago minima**		Fabaceae	He et al. (2001)
Non cultivated		*Melilotus suaveolens*		Fabaceae	He et al. (2001)
Non cultivated		*Vicia tetrasperma**		Fabaceae	He et al. (2001)
Non cultivated		*Leonurus heterophyllus*		Lamiaceae	He et al. (2001)
Non cultivated		*Althaea rosea*		Malvaceae	He et al. (2001); Korytkowski (1982)
Non cultivated		*Malva rotunditolia*		Malvaceae	He et al. (2001)
Non cultivated		*Malva verticillate**		Malvaceae	He et al. (2001); Wei et al. (2000)
Non cultivated		*Sida* sp.*		Malvaceae	Scheffer et al. (2006)
Non cultivated		*Stephania delavayi*		Menispermaceae	He et al. (2001)
Non cultivated		*Oxalis corniculate**		Oxalidaceae	He et al. (2001)
Non cultivated		*Plantago asiatica*		Plantaginaceae	He et al. (2001)
Non cultivated	Broad leaved plantain	*Plantago major**		Plantaginaceae	He et al. (2001)
Non cultivated		*Veronica anagallis-aquatica**		Plantaginaceae	He et al. (2001)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Non cultivated		*Setaria viridis**		Poaceae	Wei et al. (2000)
Non cultivated		*Polygonum amphibium*		Polygonaceae	He et al. (2001)
Non cultivated	Knotweed	*Polygonum aviculare**		Polygonaceae	He et al. (2001)
Non cultivated		*Polygonum hydropiper**		Polygonaceae	He et al. (2001)
Non cultivated		*Polygonum nepalense*		Polygonaceae	He et al. (2001)
Non cultivated		*Rumex acetosa**		Polygonaceae	He et al. (2001)
Non cultivated		*Nemesia strumose**		Scrophulariaceae	He et al. (2001)
Non cultivated		*Datura ferox**		Solanaceae	Valladares et al. (2011); Valladares et al. (1999); Salvo and Valladares (1997)
Non cultivated		*Datura stramonium**		Solanaceae	Mujica (2007); He et al. (2001); Hincapie et al. (1993)
Non cultivated		*Nicotiana glauca**		Solanaceae	Korytkowski (2014); Korytkowski (1982)
Non cultivated		*Nicotiana physaloides*		Solanaceae	He et al. (2001)
Non cultivated	Black nightshade	*Solanum nigrum**		Solanaceae	He et al. (2001)
Non cultivated		*Solanum oleracelus*		Solanaceae	Noujeim et al. (2013)

Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the serpentine leafminer (*Liriomyza huidobrensis*)

This includes all records for which no evidence was presented within the record for lifecycle completion and no evidence was shown for confident species identification, or evidence could not suitably be accessed for verification. We are not disputing the veracity of these records - we simply did not have enough information available to verify if these hosts support lifecycle completion in the field. This includes all records where:

(1) Adults were collected by sweep netting the presumed host, but neither larval activity nor lifecycle completion was confirmed; or

(2) A scientific name could not reliably be assumed from a reported common name; or

(3) Collection and identification methodology were unclear or unreported; or

(4) The record could not be accessed or suitably translated to confirm collection and identification methodology.

For some unverified species within this table, there may be other congeneric species that have been either comprehensively or partially confirmed as a host plant, and thus these would appear within Table 13 and Table 14. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). For all non-cultivated plants, and asterisk after the scientific name indicates that the Atlas of Living Australia reports this species present within Australia (Atlas of Living Australia 2020). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 13, while records that report only partial confirmations of lifecycle completion are included in Table 14. *Liriomyza huidobrensis* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 15 Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the serpentine leafminer (Liriomyza huidobrensis).

Host type	Common names	Scientific name	Variety	Family	Records
Commercial		*Sambucus* sp.		Adoxaceae	Weintraub et al. (2017)
Commercial	Leek	*Allium porrum*		Amaryllidaceae	Weintraub et al. (2017)
Commercial		*Levisticum officinale*		Apiaceae	Weintraub et al. (2017)
Commercial		*Petroselinum* sp.		Apiaceae	Hincapie et al. (1993)
Commercial		*Asparagus officinalis*		Asparagaceae	Silva (1993)

Host type	Common names	Scientific name	Variety	Family	Records
Commercial		*Cichorium intybus*		Asteraceae	Valladares et al. (1996); Valladares (1984); Valladares et al. (1999); Valladares et al. (2011); Weintraub et al. (2017)
Commercial		*Cannabis sativa*		Cannabaceae	EPPO (2020)
Commercial		*Humulus scandens*		Cannabaceae	Weintraub et al. (2017)
Commercial	Common cornsalad	*Valerianella locusta*		Caprifoliaceae	CABI (2019)
Commercial	Lentil; Lentil bean	*Lens culinaris*		Fabaceae	(Koch and Waterhouse 2000)
Commercial	Black gram	*Vigna mungo*		Fabaceae	Weintraub et al. (2017)
Commercial	Passionfruit	*Passiflora* sp.		Passifloraceae	Kahinga et al. (2017)
Ornamental		*Oenanthe benghalensis*		Apiaceae	Weintraub et al. (2017)
Ornamental		*Chrysanthemum indicum*		Asteraceae	EPPO (2020)
Ornamental		*Dahlia imperialis*		Asteraceae	Spencer (1983)
Ornamental		*Dendranthema x grandiflorum*		Asteraceae	EPPO (2020)
Ornamental	Sunflower; Common sunflower	*Helianthus annuus*		Asteraceae	Korytkowski (1982); Valladares et al. (1999); Valladares et al. (2011)
Ornamental		*Leucanthemum vulgare*		Asteraceae	EPPO (2020)
Ornamental		*Leucanthemum x superbum*		Asteraceae	EPPO (2020)
Ornamental	Butterweed	*Senecio cruentus*		Asteraceae	Calabretta et al. 1995
Ornamental		*Tanacetum vulgare*		Asteraceae	EPPO (2020)
Ornamental		*Primula x polyanthus*		Primulaceae	EPPO (2020)
Ornamental		*Ranunculus chinensis*		Ranunculaceae	Weintraub et al. (2017)
Ornamental		*Ranunculus sieboldii*		Ranunculaceae	Weintraub et al. (2017)
Ornamental		*Viola philippica*		Violaceae	Weintraub et al. (2017)
Non cultivated		*Trianthema portulacastrum**		Aizoaceae	Mujica (2007)

Host type	Common names	Scientific name	Variety	Family	Records
Non cultivated		*Alternanthera philox*	*eroides*	Amaranthaceae	Weintraub et al. (2017)
Non cultivated		*Amaranthus hybridus**		Amaranthaceae	Mujica (2007)
Non cultivated		*Chenopodium hircinum*		Amaranthaceae	Korytkowski (1982)
Non cultivated		*Artemisia argyi*		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Carduus nutans**		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Conyza bonariensis**		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Eclipta prostrata**		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Galinsoga urticifolia*		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Gnaphalium affine*		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Gynura crepidioides**		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Schistocarpha platyphylla*		Asteraceae	Weintraub et al. (2017)
Non cultivated		*Taraxacum officinale**		Asteraceae	Valladares et al. (1999); Valladares et al. (2011)
Non cultivated		*Diplotaxis muralis**		Brassicaceae	Mujica (2007)
Non cultivated		*Stellaria alsine*		Caryophyllaceae	Weintraub et al. (2017)
Non cultivated		*Vaccaria pyramidata**		Caryophyllaceae	Iwasaki (2004a)
Non cultivated	Chipilín	*Crotalaria longirostrata*		Fabaceae	Weintraub et al. (2017)
Non cultivated		*Streptocarpus sp.*		Gesneriaceae	Weintraub et al. (2017)
Non cultivated		*Leonurus sibiricus*		Lamiaceae	Weintraub et al. (2017)
Non cultivated		*Stachys arvensis**		Lamiaceae	Mujica (2007)
Non cultivated		*Alcea* sp.*		Malvaceae	Weintraub et al. (2017)
Non cultivated	Purslane	*Portulaca oleracea**		Portulacaceae	CABI (2019); Hincapie et al. (1993)

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Appendix 2: Leaf mining damage caused by established species

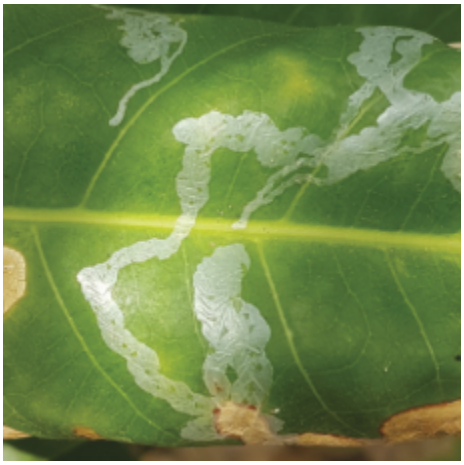
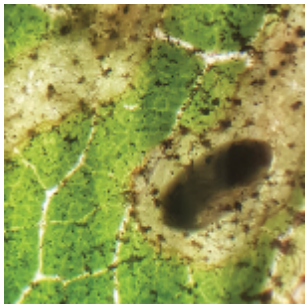
Table 16 provides a summary of established (native or naturalised) leaf mining species, their host overlap with pea leafminer and features that can be used to differentiate between serpentine leafminer and established leafminers.

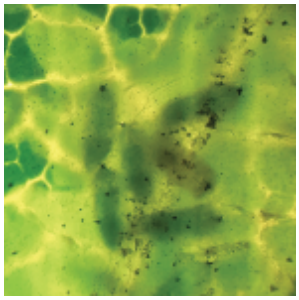
It should be noted that because host plant records are poor for many native or naturalised species, there could be many more affected hosts than included here. The host overlap between serpentine leafminer and several native leafminer underscores the importance of sample collection and molecular diagnostics.

Figure 8 provides some general guidelines for differentiating between Dipteran and Lepidopteran leafminer larvae.

Table 16 Leaf mining damage caused by Australian dipterans or lepidopterans on leaves of cultivated crops and common weeds of cultivated crops

LEAFMINER	DISTINGUISHING FEATURES FROM SERPENTINE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH SERPENTINE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Liriomyza brassicae	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from serpentine leafminer)	Brassicaceae Capparaceae Tropaeolaceae Fabaceae	
Liriomyza chenopodii	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from serpentine leafminer)	Amaranthaceae Caryophyllaceae Any native hosts are as yet unrecorded (*L. chenopodii* is a native species)	

LEAFMINER	DISTINGUISHING FEATURES FROM SERPENTINE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH SERPENTINE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Phytomyza syngenesiae	Pupation occurs inside the leaf mine.  Adults easily distinguishable from serpentine leafminer.	Asteracea Apiaceae Fabaceae	
Tropicomyia polyphtya	Larvae are upper surface epidermal feeders (deeper parenchymatous tissues is not eaten). As such, mines have a silvery, film like appearance. Pupation occurs inside the mine.  Adults easily distinguishable from serpentine leafminer	Highly polyphagous Common hosts include plants in the Passifloraceae, Euphorbiaceae, Fabaceae, Rubiaceae, Ruaceae, and Solanaceae families, and more.	
Ophiomyia solanicola	Pupation occurs inside the leaf mine.  Adults easily distinguishable from serpentine leafminer	Solanaceae	

LEAFMINER	DISTINGUISHING FEATURES FROM SERPENTINE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH SERPENTINE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Ophiomyia alysicarpi	Mint green pupae can be found inside mines  Adults easily distinguishable from serpentine leafminer	*Alysicarpus* sp. *Desmodium* sp.	
Ophiomyia cornuta	Adults easily distinguishable from serpentine leafminer	*Scaevola* sp. *Goodenia* sp.	
epidopteran leafminer	Lepidopteran larvae can be distinguished from dipteran larvae on the spot, via a hand lens (Figure 10). For some lepidopteran species, when the larva is in its final stages, it forms a large blister at the end of the mine (see images on the right). Serpentine leafminer larvae do not create such blisters.	Polyphagous (incl. Fabaceae, Eucalypts, native trees, etc)	

LEAFMINER	DISTINGUISHING FEATURES FROM SERPENTINE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH SERPENTINE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
			 

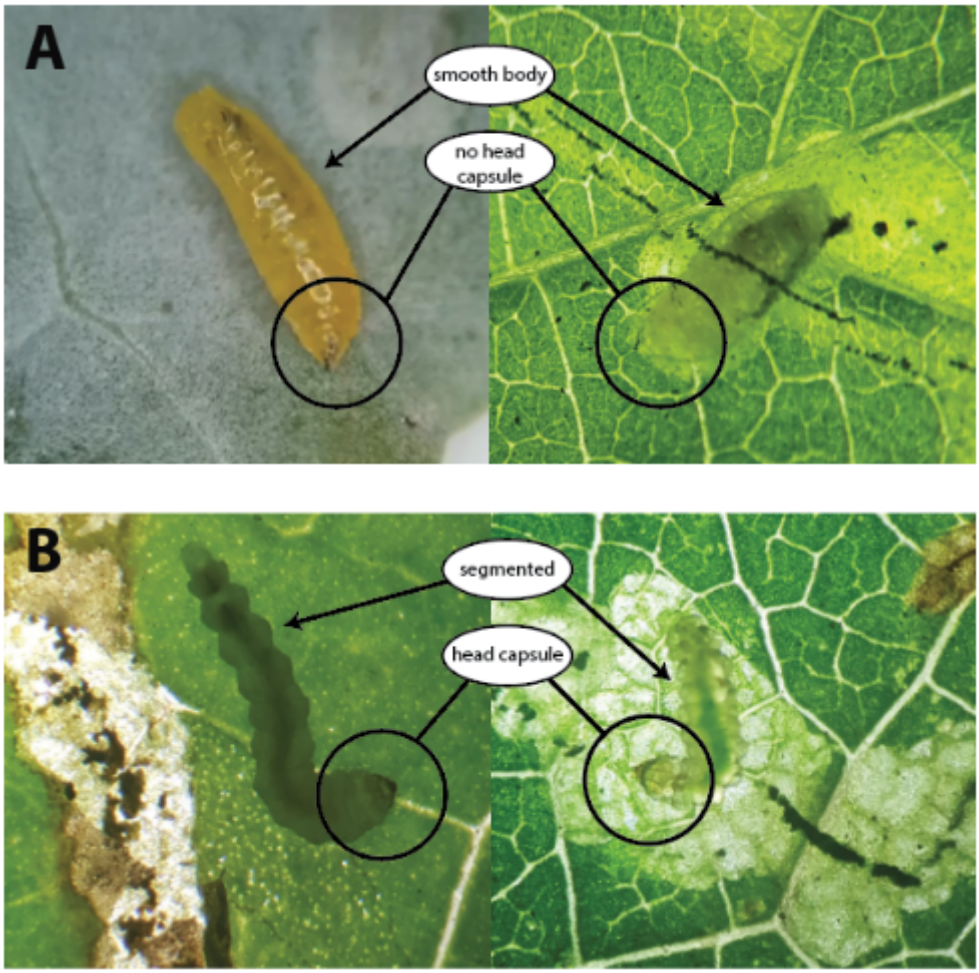


Figure 8 General guidelines for distinguishing dipteran vs lepidopteran leafminer larvae. Dipteran leafminer larvae (A) lack a head capsule and have a smooth body without clear segments. Lepidopteran leafminer larvae (B) have a distinct head capsule and are more visibly segmented. When attempting to distinguish a dipteran from lepidopteran leafminer larva on the spot, it is helpful to backlight the leaf as it is viewed under a hand lens, or to carefully excise the larva. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

Surveillance for *L. huidobrensis* in Australia will be confounded by the presence of other leafmining insects already present in Australia, many of which create mines that are indistinguishable to a casual observer, and even in some cases indistinguishable to experts.

Table 17 lists genera that include hosts of *L. huidobrensis* but do not appear to contain of other Agromyzids within Australia or Australasia. It is important to note that host plant records for non-pest leafminer flies are highly incomplete, and some of the genera included herein could host leafminer flies that have not yet been reported. Host records for weeds and native plants are expected to be more incomplete than for cultivated crops and ornamentals, and despite poor host records for native leafminer. We can be more confident that native leafminer are not currently a notable presence within the genera containing cultivated crops and ornamentals, where sampling effort is expected to be higher. As such, leafmining damage detected within the genera included in these tables should be regarded as highly suspicious.

Error! Reference source not found. indicates where known overlap exists between hosts of *L. huidobrensis*, *L. trifolii* or *L. sativae* and native or naturalised agromyzids in Australasia.

*Table 17 Host genera of L. huidobrensis, L. sativae or L trifolii (including only records with comprehensive or partial evidence for lifecycle completion in the field, see Appendix 1) without any reported overlap with native or naturalised agromyzids already present in Australasia (see **Error! Reference source not found.** for known overlap). For each host, preferences of the three exotic Liriomyza are indicated (LH = L. huidobrensis; LT = L. trifolii; LS = L. sativae).*

Genus	Family	Exotic *Liriomyza*	Host type
Allium	Amaryllidaceae	LH; LT; LS	Commercial
Coriandrum	Apiaceae	LH; LT; LS	Commercial
Eryngium	Apiaceae	LH	Commercial
Petroselinum	Apiaceae	LT	Commercial
Colocasia	Araceae	LH	Commercial
Cichorium	Asteraceae	LH	Commercial
Crassocephalum	Asteraceae	LH	Commercial
Basella	Basellaceae	LH	Commercial
Eruca	Brassicaceae	LH	Commercial
Spinacea	Chenopodiaceae	LH	Commercial
Benincasa	Cucurbitaceae	LH; LT; LS	Commercial
Coccinia	Cucurbitaceae	LT	Commercial
Lagenaria	Cucurbitaceae	LH; LT; LS	Commercial
Luffa	Cucurbitaceae	LH; LT; LS	Commercial
Momordica	Cucurbitaceae	LH; LT; LS	Commercial
Sechium	Cucurbitaceae	LH	Commercial
Trichosanthes	Cucurbitaceae	LT	Commercial
Arachis	Fabaceae	LT	Commercial
Cicer	Fabaceae	LH; LT; LS	Commercial
Cyamopsis	Fabaceae	LT	Commercial
Lens	Fabaceae	LS	Commercial
Lotus	Fabaceae	LH	Commercial
Lupin	Fabaceae	LH	Commercial
Medicago	Fabaceae	LH; LT; LS	Commercial
Trigonella	Fabaceae	LH; LT	Commercial
Vicia	Fabaceae	LH; LT; LS	Commercial
Mentha	Lamiaceae	LH; LS	Commercial
Origanum	Lamiaceae	LS	Commercial
Linum	Linaceae	LH	Commercial
Abelmoschus	Malvaceae	LH; LT; LS	Commercial
Corchorus	Malvaceae	LT	Commercial
Gossypium	Malvaceae	LT; LS	Commercial
Hibiscus	Malvaceae	LH; LT	Commercial
Zizania	Poaceae	LH	Commercial
Lycium	Solanaceae	LH	Commercial
Thurnbergia	Acanthaceae	LH	Ornamental
Celosia	Amaranthaceae	LH; LT	Ornamental
Bupleurum	Apiaceae	LH	Ornamental
Centella	Apiaceae	LH	Ornamental
Oenanthe	Apiaceae	LH	Ornamental
Catharanthus	Apocynaceae	LH	Ornamental
Asclepias	Asclepiadaceae	LT	Ornamental
Chionodoxa	Asparagaceae	LH	Ornamental
Hemerocallis	Asphodelaceae	LH	Ornamental
Argyranthemum	Asteraceae	LH	Ornamental

Genus	Family	Exotic *Liriomyza*	Host type
Calendula	Asteraceae	LH; LT; LS	Ornamental
Centaurea	Asteraceae	LH	Ornamental
Conoclinium	Asteraceae	LH	Ornamental
Cosmos	Asteraceae	LH	Ornamental
Dimorphotheca	Asteraceae	LT	Ornamental
Echinacea	Asteraceae	LT	Ornamental
Eupatorium	Asteraceae	LT	Ornamental
Felicia	Asteraceae	LS	Ornamental
Flaveria	Asteraceae	LT	Ornamental
Kalimeris	Asteraceae	LH	Ornamental
Osteospermum	Asteraceae	LH	Ornamental
Pyrethrum	Asteraceae	LH	Ornamental
Tanacetum	Asteraceae	LH	Ornamental
Zinnia	Asteraceae	LH; LT; LS	Ornamental
Impatiens	Balsaminaceae	LH	Ornamental
Nasturtium	Brassicaceae	LH; LS	Ornamental
Calceolaria	Calceolariaceae	LH	Ornamental
Campanula	Campanulaceae	LH	Ornamental
Platycodon	Campanulaceae	LH	Ornamental
Trachelium	Campanulaceae	LH; LT	Ornamental
Dianthus	Caryophyllaceae	LH; LT	Ornamental
Gypsophila	Caryophyllaceae	LH; LT; LS	Ornamental
Bauhinia	Fabaceae	LS	Ornamental
Lathyrus	Fabaceae	LH	Ornamental
Lupinus	Fabaceae	LH; LS	Ornamental
Eustoma	Gentianaceae	LH; LT	Ornamental
Exacum	Gentianaceae	LH	Ornamental
Lisianthus	Gentianaceae	LH; LT; LS	Ornamental
Freesia	Iridaceae	LH	Ornamental
Gladiolus	Iridaceae	LH	Ornamental
Salvia	Lamiaceae	LH; LT	Ornamental
Lilium	Liliaceae	LH	Ornamental
Torenia	Linderniaceae	LH	Ornamental
Althaea	Malvaceae	LH	Ornamental
Moringa	Moringaceae	LS	Ornamental
Godetia	Onagraceae	LH	Ornamental
Oenothera	Onagroideae	LH	Ornamental
Dendrobium	Orchidaceae	LS	Ornamental
Papaver	Papaveraceae	LH	Ornamental
Antirrhinum	Plantaginaceae	LH; LT; LS	Ornamental
Myosotis	Plumbaginaceae	LH	Ornamental
Lagurus	Poaceae	LH	Ornamental
Phlox	Polemoniaceae	LH; LT; LS	Ornamental
Primula	Primulaceae	LH	Ornamental
Aquilegia	Ranunculaceae	LS	Ornamental
Delphinium	Ranunculaceae	LH	Ornamental
Nigella	Ranunculaceae	LH	Ornamental
Prunus	Rosaceae	LS	Ornamental

Genus	Family	Exotic *Liriomyza*	Host type
Rosa	Rosaceae	LH; LT	Ornamental
Diascia	Scrophulariaceae	LH	Ornamental
Viola	Violaceae	LH	Ornamental
Sagittaria	Alismataceae	LH	Non cultivated
Alternanthera	Amaranthaceae	LT	Non cultivated
Amaranthus	Amaranthaceae	LH; LT; LS	Non cultivated
Deeringia	Amaranthaceae	LH	Non cultivated
Gomphrena	Amaranthaceae	LH	Non cultivated
Iresine	Amaranthaceae	LH	Non cultivated
Alocasia	Araceae	LH	Non cultivated
Hydrocotyle	Araliaceae	LH; LT; LS	Non cultivated
Artemisia	Asteraceae	LH; LT	Non cultivated
Baccharis	Asteraceae	LT	Non cultivated
Carduus	Asteraceae	LH	Non cultivated
Dichrocephala	Asteraceae	LH	Non cultivated
Echinops	Asteraceae	LH	Non cultivated
Eclipta	Asteraceae	LT	Non cultivated
Emilia	Asteraceae	LH	Non cultivated
Erechtites	Asteraceae	LH; LT	Non cultivated
Galinsoga	Asteraceae	LH; LT; LS	Non cultivated
Helipterum	Asteraceae	LH	Non cultivated
Hemistepta	Asteraceae	LH	Non cultivated
Hymenopappus	Asteraceae	LT	Non cultivated
Lipochaeta	Asteraceae	LS	Non cultivated
Melanthera	Asteraceae	LT	Non cultivated
Parthenium	Asteraceae	LT	Non cultivated
Synedrella	Asteraceae	LH; LT	Non cultivated
Tridax	Asteraceae	LT	Non cultivated
Verbesina	Asteraceae	LS	Non cultivated
Xanthium	Asteraceae	LT	Non cultivated
Cordia	Boranginaceae	LT	Non cultivated
Capsella	Brassicaceae	LH	Non cultivated
Rorippa	Brassicaceae	LH	Non cultivated
Silene	Caryophillaceae	LH; LT	Non cultivated
Malachium	Caryophyllaceae	LH	Non cultivated
Spergula	Caryophyllaceae	LT	Non cultivated
Commelina	Commelinaceae	LT	Non cultivated
Calystegia	Convolvulaceae	LH	Non cultivated
Convolvulus	Convolvulaceae	LT	Non cultivated
Pharbitis	Convolvulaceae	LH	Non cultivated
Ceratosanthes	Cucurbitaceae	LS	Non cultivated
Melothria	Cucurbitaceae	LH	Non cultivated
Melilotus	Fabaceae	LH; LS	Non cultivated
Poissonia	Fabaceae	LS	Non cultivated
Lamium	Lamiaceae	LH	Non cultivated
Leonurus	Lamiaceae	LH	Non cultivated
Melissa	Lamiaceae	LS	Non cultivated
Anoda	Malvaceae	LS	Non cultivated

Genus	Family	Exotic *Liriomyza*	Host type
Malva	Malvaceae	LH; LS	Non cultivated
Sida	Malvaceae	LH; LS	Non cultivated
Oxalis	Oxalidaceae	LH; LT	Non cultivated
Piriqueta	Passifloraceae	LT	Non cultivated
Linaria	Plantaginaceae	LT	Non cultivated
Veronica	Plantaginaceae	LH	Non cultivated
Limonium	Plumbaginaceae	LH	Non cultivated
Polygala	Polygalaceae	LT	Non cultivated
Rumex	Polygonaceae	LH	Non cultivated
Cardiospermum	Sapindaceae	LT	Non cultivated
Nemesia	Scrophulariaceae	LH	Non cultivated
Datura	Solanaceae	LH; LT; LS	Non cultivated
Nicotiana	Solanaceae	LH; LT	Non cultivated
Kallstroemia	Zygophyllaceae	LT	Non cultivated
Tribulus	Zygophyllaceae	LT	Non cultivated

Table 18 Host genera of either L. huidobrensis, L. sativae or L trifolii with known overlap with native or naturalised agromyzids already present in Australasia. Some of these species are stem miner, which should be straightforward to distinguish from the leaf mining damage caused by L. huidobrensis, L. sativae or L trifolii. Records of L. sativae host plants within the Torres Strait Islands (TSI) and on the Cape York Peninsula (CYP) are included.

Host genus	Host family	Australasian Agromyzid genera	Sources
Beta	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977); Spencer (1990)
Chenopodium	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Spinacia	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Apium	Apiaceae	*Melanagromyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Daucus	Apiaceae	*Phytomyza*	Spencer (1973)
Asparagus	Asparagaceae	*Hexomyza*	Benavent Corai et al. (2005)
Ageratum	Asteraceae	*Melanagromyza*	Spencer (1977)
Arctium	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Aster	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Bellis	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Bidens	Asteraceae	*Melanagromyza*	Spencer (1977)
Callistephus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Carthamus	Asteraceae	*Melanagromyza*	Benavent Corai et al. (2005)
Chrysanthemum	Asteraceae	*Ophiomyia, Phytomyza, Melanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Cineraria	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cirsium	Asteraceae	*Phytomyza*	Spencer (1977)
Conyza	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Coreopsis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Craspedia	Asteraceae	*Liriomyza*	Benavent Corai et al. (2005)
Crepis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cynara	Asteraceae	*Phytomyza*	Spencer (1973)
Dahlia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Erigeron	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005); Spencer (1977)
Gaillardia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gerbera	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gnaphalium	Asteraceae	*Melanagromyza, Malanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Helianthus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Helichrysum	Asteraceae	*Liriomyza, Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Lactuca	Asteraceae	*Liriomyza*, Phytomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1973)
Leucanthemum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Picris	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Senecio	Asteraceae	*Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1977)
Solidago	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005)
Sonchus	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)

Host genus	Host family	Australasian Agromyzid genera	Sources
Tagetes	Asteraceae	*Liriomyza**	Present in TSI/CYP
Taraxacum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Tecoma	Bignoniaceae	*Liriomyza**	Present in TSI/CYP
Barbarea	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Brassica	Brassicaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Cardamine	Brassicaceae	*Liriomyza*	Spencer (1977)
Diplotaxis	Brassicaceae	*Liriomyza*	Spencer (1977)
Hirschfeldia	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Matthiola	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977)
Raphanus	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Silene	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Stellaria	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Cleome	Cleomaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP
Ipomoea	Convolvulaceae	*Melanagromyza*	Benavent Corai et al. (2005)
Citrullus	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucumis	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucurbita	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Euphorbia	Euphorbiaceae	*Tropicomyia*	Spencer (1977)
Ricinus	Euphorbiaceae	*Liriomyza**	Present in TSI/CYP
Cajanus	Fabaceae	*Melanagromyza, Ophiomyia*	Spencer (1973); Spencer (1977)
Canavalia	Fabaceae	*Ophiomyia*	Spencer (1973)
Cassia	Fabaceae	*Tropicomyia, Ophiomyia*	Spencer (1977); Spencer (1990)
Crotalaria	Fabaceae	*Liriomyza*, Ophiomyia*	Present in TSI/CYP; Spencer (1973); Spencer (1977)
Dolichos	Fabaceae	*Ophiomyia*	Spencer (1973); Spencer (1977)
Glycine	Fabaceae	*Melanagromyza, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Indigofera	Fabaceae	*Liriomyza*, Ophiomyia*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977); Spencer (1990)
Lablab	Fabaceae	*Ophiomyia*	Spencer (1973)
Macroptilium	Fabaceae	*Liriomyza*, Tropicomyia*	Present in TSI/CYP; Spencer (1977)
Millettia	Fabaceae	*Unknown*	Pirtle pers. comm.
unknown	Fabaceae	*Melanagromyza*	Spencer (1977)
Phaseolus	Fabaceae	*Ophiomyia, Tropicomyia, Melanagromyza, Ophiomyla*	Spencer (1973); Spencer (1977)
Pisum	Fabaceae	*Liriomyza, Phytomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Senna	Fabaceae	*Tropicomyia*	Spencer (1977)
Sesbania	Fabaceae	*Liriomyza**	Present in TSI/CYP
Trifolium	Fabaceae	*Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Vigna	Fabaceae	*Liriomyza*, Melanagromyza, Tropicomyia, Ophiomyia*	Present in TSI/CYP; Pirtle et al. (2020); Spencer (1973); Spencer (1977); Spencer 1977

Host genus	Host family	Australasian Agromyzid genera	Sources
Scaevola	Goodeniaceae	*Liriomyza, Ophiomyia*	Benavent Corai et al. (2005); Pirtle pers. comm.; Spencer (1977)
Hydrangea	Hydrangeaceae	*Tropicomyia*	Spencer (1977)
Ocimum	Lamiaceae	*Liriomyza**	Pirtle et al. (2020)
Stephania	Menispermaceae	*Tropicomyia*	Spencer (1977); Spencer (1990)
Passiflora	Passifloraceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977); Spencer (1990)
Turnera	Passifloraceae	*Liriomyza**	Pirtle et al. (2020)
Plantago	Plantaginaceae	*Liriomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977);
Hordeum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
unknown	Poaceae	*Agromyza, Cerodontha*	Spencer (1973)
Setaria	Poaceae	*Agromyza*	Benavent Corai et al. (2005)
Triticum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
Zea	Poaceae	*Pseudonapomyza*	Spencer (1973)
Portulaca	Portulacaceae	*Liriomyza*	Benavent Corai et al. (2005)
Ranunculus	Ranunculaceae	*Phytomyza, Napomyza*	Spencer (1977); Spencer (1990)
Passiflora	Resedaceae	*Tropicomyia*	Spencer (1990)
Coffea	Rubiaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Citrus	Rutaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Capsicum	Solanaceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977)
Cestrum	Solanaceae	*Tropicomyia*	Spencer (1977)
Lycopersicum	Solanaceae	*Liriomyza**	Present in TSI/CYP
Petunia	Solanaceae	*Liriomyza**	Present in TSI/CYP
Physalis	Solanaceae	*Liriomyza**	Present in TSI/CYP; Pirtle et al. (2020)
Solanum	Solanaceae	*Ophiomyia, Liriomyza**	Pirtle pers. comm.; Present in TSI/CYP; Spencer (1977)
Tropaeolum	Tropaeolaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Lantana	Verbenaceae	*Calycomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Stachytarpheta	Verbenaceae	*Liriomyza**	Pirtle et al. (2020)
Verbena	Verbenaceae	*Calycomyza*	Benavent Corai et al. (2005)

* Includes *Liriomyza sativae* within TSI/CYP

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Appendix 3: Permits for the control of serpentine leafminer and other *Liriomyza* spp. on different host crops

Table 19 lists the agriculturally important hosts of serpentine leafminer (refer to Appendix 1) and notes where suitable pesticides are available and approved by the APVMA for control on each host crop. Blank cells denote where suitable pesticides have not yet been approved or identified. Refer to the APVMA permit database (https://portal.apvma.gov.au/permits) or the most up to date information on current leafminer permits.

Table 19 Insecticides control options for different crop groups

Relevant Crop group	Crop/host plant	Pesticide	Permit number(s)/ status	Target pest (as per permit)	Expiration date	Withholding information/notes	Suitable for management or eradication[2]
Brassica vegetables	Bok choy; pak-choy; turnip	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Broccoli	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Broccoli	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

[2] Some pesticides will be more suitable for eradication than for the ongoing management of the pest. This is because either they will need to be used at high rates to be effective, there are potential resistance issues, or the pesticide is not likely to fit into existing IPM systems as well as alternative pesticides. Other chemicals provide good control of the pest and are also compatible with IPM systems and will be more suitable for the ongoing management of the pest. To highlight this, pesticides are noted for either eradication, or for management and eradication.

Brassica vegetables	Brussels sprouts	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Brussels sprouts	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cabbage	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cauliflower	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cauliflower	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L.*	30 June 2024	3 days (harvest) Do not graze or cut for	Management or eradication

				sativae)		stock food	
Brassica vegetables	Indian mustard	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Bulb vegetables	Garlic	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Garlic	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	30 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Bulb vegetables	Shallots	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Shallots	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Cereal grains	Barley	No suitable pesticides identified					
Fruit trees (non-bearing)	Any trees that will not bear fruit for the next 12 months	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Cucumber	Cyromazine	PER81867	*Liriomyza*	30 November	7 days (harvest)	Management or

vegetables (Cucurbits – including melons)				*sativae* and *L. huidobrensis*	2023	Do not graze or cut for stock food	eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits –	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

including melons)							
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Watermelon	Abamectin	PER81876	*Liriomyza*	30 April 2024	7 days (harvest)	Management or

vegetables (Cucurbits – including melons)				*sativae* and *L. huidobrensis*		Do not graze or cut for stock food	eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Zucchini	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables	Capsicum, chili*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:*	31 December 2023	1 day (harvest) Do not graze or cut for	Management or eradication

(other than cucurbits)				*L. sativae, L trifolii, L. huidobrensis*)		stock food	
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Eggplant	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Okra*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (other than cucurbits)	Okra*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Okra*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Herbs	Basil	No suitable pesticides identified					
Herbs	Oregano	No suitable pesticides identified					

Herbs	Parsley[3]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management
Head lettuce	Lettuce (Head type only)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Lettuce	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Leafy vegetables (including brassica leafy vegetables)	Spinach	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Spinach	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Leafy vegetables	Spinach and silverbeet	Chlorantraniliprole	PER87631	*Liriomyza* spp. (including*:*	30 June 2024	3 days (harvest) 7 days (grazing or	Management or eradication

[3] Note: Parsley is not a recorded host for *L. sativae* but may be a host of other *Liriomyza* spp. and is listed as one of the crops that the permit covers.

(including brassica leafy vegetables)				*L. sativae, L brassicae, L. huidobrensis*)		cutting for stock food)	
Legume vegetables	Common bean; French beans, kidney beans	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	7 days (harvest) 7 days (grazing and hay)	Management
Legume vegetables	Faba bean, broad bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Faba bean, broad bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas,	Spinetoram	PER87878	*Liriomyza* spp.	28 February 2023	3 days (harvest); 14 days (grazing and	Management or eradication

	field pea					hay) Do not allow dairy cattle to graze treated forage	
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest); 3 days (grazing and hay)	Management
Legume vegetables	Red bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Red bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Not classified	Field mustard	No suitable pesticides identified					
Not classified	Lucerne	No suitable pesticides identified					
Nursery stock (non-food)	Various non-food nursery plants (e.g. chrysanthemum etc.)	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Nursery stock	Seedlings, tubes	Abamectin	PER88977	Leafminers	30	Not required when	Management or

(non-food)	and plugs, potted colour, trees and shrubs, foliage plants, palms, grasses, fruiting plants (non-bearing), cut flowers and ornamentals.	Azadirachtin Cyromazine Emamectin Chlorantraniliprole + thiamethoxam Cyantraniliprole Indoxacarb Spinetoram		(*Liriomyza* spp.) including Vegetable leafminer (*Liriomyza sativae*)	November, 2022	used as directed	eradication
Oilseeds	Cotton	Spinetoram has been identified as suitable for the control of Vegetable leafminer and other *Liriomyza* spp. on cotton as overseas and Australian use patterns are comparable. However, no permit has been applied for this use to date					
Pulses	Pulses	Dimethoate	PER89184	*Liriomyza sativae, Liriomyza trifolii, Liriomyza huidobrensis*	31 March 2025	Harvest: Do not harvest for 14 days after application. Grazing: Do not graze or cut for stock food for 14 days after application.	Management
Root and tuber vegetables	Beets (beetroot, silver beet, sugar beets)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Beets (beetroot, silver beet, sugar beets)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Carrot	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Carrot	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber	Potato	Abamectin	PER81876	*Liriomyza sativae* and	30 April 2024	14 days (harvest) Do not graze or cut for	Management or eradication

vegetables				*L. huidobrensis*		stock food	
Root and tuber vegetables	Potato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Potato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Radish	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Radish	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Celery	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[4]	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication

[4] Note: Rhubarb is not a recorded host for *L. sativae or L. huidobrensis* but may be a host of other *Liriomyza* spp. and is part of the 'Stalk and Stem Vegetable Crop Group' that the permit covers.

Stalk and stem vegetables	Rhubarb[3]	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication

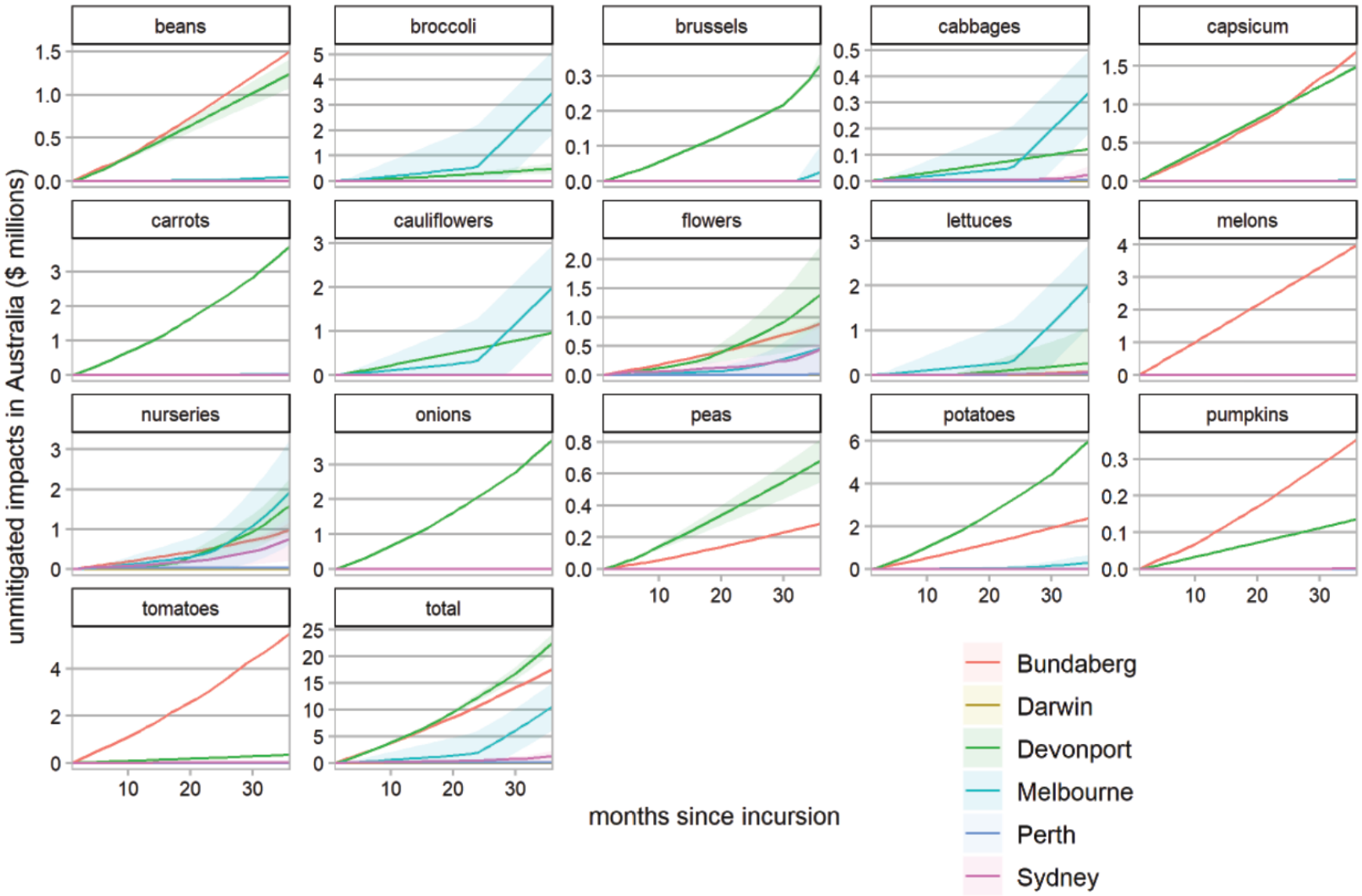
Appendix 4: Additional impact prediction outputs

Table 20 State level accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. huidobrensis *at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preferences hosts may be overestimated while impacts of high preference hosts may be underestimated. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.*

	INCURSION LOCATION															
	NSW			NT		QLD			SA	TAS	VIC		WA			
Crop	**Griffith**	**Sydney**	**Western Sydney**	**Darwin**	**Katherine**	**Bowen**	**Bundaberg**	**Toowoomba**	**Adelaide**	**Devonport**	**Ballarat**	**Melbourne**	**Albany**	**Carnarvon**	**Geraldton**	**Perth**
Beans	0.00 (0.00)	0.00 (0.00)	0.05 (0.00)	0.00 (0.00)	0.00 (0.00)	1.32 (0.00)	1.50 (0.00)	0.03 (0.02)	0.00 (0.00)	1.17 (0.00)	0.02 (0.01)	0.05 (0.02)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Broccoli	0.03 (0.00)	0.00 (0.00)	0.03 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.46 (0.01)	0.00 (0.00)	0.38 (0.00)	1.07 (1.08)	3.49 (1.63)	0.04 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.01)
Brussels	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	2.41 (0.00)	0.32 (0.00)	0.02 (0.06)	0.03 (0.08)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Cabbages	0.00 (0.00)	0.02 (0.03)	1.20 (0.02)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.35 (0.01)	0.10 (0.00)	0.12 (0.00)	0.12 (0.12)	0.34 (0.16)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Capsicum	0.04 (0.00)	0.00 (0.00)	0.07 (0.00)	0.00 (0.00)	0.00 (0.00)	3.68 (0.00)	1.69 (0.00)	0.00 (0.01)	0.00 (0.00)	1.47 (0.00)	0.08 (0.21)	0.01 (0.02)	0.04 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Carrots	0.02 (0.00)	0.00 (0.01)	0.02 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.07 (0.02)	0.00 (0.00)	3.72 (0.00)	0.05 (0.04)	0.03 (0.03)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.02 (0.03)
Cauliflowers	0.00 (0.00)	0.01 (0.01)	0.05 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.44 (0.01)	0.00 (0.00)	0.97 (0.00)	0.57 (0.64)	2.00 (0.96)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.01)
Flowers	0.00 (0.00)	0.44 (0.48)	3.87 (0.48)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.88 (0.07)	0.53 (0.18)	0.64 (0.04)	0.63 (0.02)	1.42 (0.61)	0.47 (0.51)	0.12 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.00)
Lettuces	0.00	0.05	0.61	0.00	0.00	0.00	0.07	1.47	0.00	0.01	0.97	2.00	0.00	0.00	0.00	0.02

	INCURSION LOCATION															
	(0.00)	(0.05)	(0.05)	(0.00)	(0.00)	(0.00)	(0.00)	(0.02)	(0.00)	(0.00)	(0.56)	(0.91)	(0.00)	(0.00)	(0.00)	(0.02)
Melons	0.90 (0.00)	0.00 (0.00)	0.03 (0.00)	0.00 (0.00)	0.17 (0.00)	0.17 (0.00)	3.98 (0.00)	0.11 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Nurseries	0.89 (0.00)	0.63 (0.46)	5.47 (0.54)	0.00 (0.00)	0.00 (0.00)	0.29 (0.00)	0.97 (0.19)	3.94 (1.86)	2.55 (0.02)	0.66 (0.01)	4.16 (2.53)	1.95 (1.31)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.03 (0.01)
Onions	0.09 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.21 (0.01)	0.00 (0.00)	3.71 (0.00)	0.01 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Peas	0.00 (0.00)	0.00 (0.00)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.29 (0.00)	0.00 (0.00)	0.00 (0.00)	0.63 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Potatoes	2.14 (0.00)	0.01 (0.02)	0.32 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	2.40 (0.00)	0.51 (0.01)	0.05 (0.00)	5.99 (0.00)	2.54 (0.29)	0.30 (0.37)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.01)
Pumpkins	0.63 (0.00)	0.00 (0.00)	0.04 (0.00)	0.00 (0.00)	0.00 (0.00)	0.31 (0.00)	0.35 (0.00)	0.20 (0.01)	0.00 (0.00)	0.13 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Tomatoes	0.25 (0.00)	0.02 (0.02)	0.92 (0.08)	0.00 (0.00)	0.00 (0.00)	9.11 (0.00)	5.50 (0.00)	0.29 (0.02)	0.00 (0.00)	0.34 (0.00)	0.18 (0.55)	0.00 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Total	**5.00 (0.00)**	**1.19 (1.01)**	**12.68 (1.08)**	**0.00 (0.00)**	**0.17 (0.00)**	**14.88 (0.00)**	**17.65 (0.24)**	**8.62 (2.09)**	**5.76 (0.06)**	**20.26 (0.03)**	**11.21 (3.35)**	**10.67 (4.44)**	**0.22 (0.00)**	**0.00 (0.00)**	**0.00 (0.00)**	**0.09 (0.08)**

Figure 9 Australia wide accumulating unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. huidobrensis *at key entry and establishment points across Australia.*



Appendix 11: *Liriomyza sativae* Continency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Rohan Burgess [1], Dr. Peter Ridland [2], Dr. Elia Pirtle [3]

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[2] The University of Melbourne

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December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Liriomyza sativae Contingency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Rohan Burgess [1], Dr. Peter Ridland [2], Dr. Elia Pirtle [3]

[1] Plant Health Australia

[2] The University of Melbourne

[3] Cesar Australia

December 7, 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

CONTINGENCY PLAN

VEGETABLE LEAFMINER (*LIRIOMYZA SATIVAE*)



Images: Elia Pirtle, Cesar Australia Pty Ltd

December 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

This Contingency Plan has been authored by Rohan Burgess (Plant Health Australia), Dr. Peter Ridland (The University of Melbourne) and Dr. Elia Pirtle (Cesar Australia), with contributions from Dr. Sharyn Taylor (Plant Health Australia), Dr. James Maino (Cesar Australia), and Dr. Paul Umina (Cesar Australia).

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1. Purpose and background of this contingency plan 6
2. Pest details 7
2.1 Background and impact of pest 7
2.2 Life cycle 8
2.3 Host range 9
2.4 Signs and symptoms 9
2.5 Dispersal 12
2.6 Current geographic distribution 12
2.7 Risk of establishment in Australia 15
2.8 Risk of spread within Australia 16
2.8.1 Risk pathways – international and domestic 16
2.8.2 Predicting spread potential 21
2.9 Risk of economic impact within Australia 23
3. Diagnostic information 25
3.1 Diagnostic considerations 25
4. Surveillance and collection of samples 27
4.1 Surveillance 27
4.1.1 Technical information for planning surveys 27
4.1.2 Delimiting surveys in the event of an incursion into new areas 30
4.1.3 Activities for general surveillance immediately following a detection 32
4.2 Collection and treatment of samples 32
4.2.1 If collecting for morphological diagnosis 32
4.2.2 If collecting for molecular diagnosis 34
5. Immediate response to detection 35
5.1 Tracing 35
5.2 Quarantine and movement controls 36
5.3 Destruction strategy 36
5.3.1 Priorities and considerations 36
5.3.2 Decontamination of tools and equipment 36
5.3.3 Chemical control 37
5.3.4 Physical control options 44
5.3.5 Biological control 44
5.4 Decision support for eradication 44
6. Emergency Plant Pest Response Deed and PLANTPLAN 49
6.1 Emergency Plant Pest Response Deed 49
6.2 PLANTPLAN 50
7. References 51
8. Appendices 57
Appendix 1: Known hosts of vegetable leafminer 57
Number of host records for the vegetable leafminer (*Liriomyza sativae*) within plant families. 57
Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*) has been provided within the scientific literature 59

Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*) has been provided within the scientific literature....74
Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*)....79
References....81
Appendix 2: Leaf mining damage caused by established species....83
References....94
Appendix 3: Permits for the control of *Liriomyza* spp. on different host crops....96
Appendix 4: Additional impact prediction outputs....113

1. PURPOSE AND BACKGROUND OF THIS CONTINGENCY PLAN

Vegetable leafminer (*Liriomyza sativae*) has recently been discovered in the Torres Strait (2008) and the northern tip of Cape York Peninsula in Queensland (2015) (IPPC 2015; 2017).

This Contingency Plan provides background information on vegetable leafminer to assist in determining the requirements for the initial response to a detection of this species in other regions of Australia. Only key information for immediate response is provided in this document.

Additional information can be found in the following supporting material:

- Awareness material such as the fact sheets from Plant Health Australia (PHA), AUSVEG, and commonwealth, state and territory jurisdictions.
    - www.planthealthaustralia.com.au/pests/vegetable-leaf-miner/
    - www.business.qld.gov.au/industries/farms-fishing-forestry/agriculture/crop-growing/priority-pest-disease/vegetable-leafminer
    - www.agriculture.gov.au/biosecurity/australia/naqs/naqs-target-lists/vegetable_leaf_miner
    - www.agriculture.gov.au/pests-diseases-weeds/plant/leaf-miner
    - https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/
- Overseas websites with additional information
    - https://gd.eppo.int/taxon/LIRISA
    - www.cabi.org/cpc/datasheet/30960
    - https://cipotato.org/riskatlasforafrica/liriomyza-sativae/
    - http://entnemdept.ufl.edu/creatures/veg/leaf/vegetable_leafminer.htm
    - http://ipm.ucanr.edu/PMG/r783300911.html

2.PEST DETAILS

Common name:	Vegetable leafminer
Scientific name:	*Liriomyza sativae* Blanchard
Synonyms:	*Agromyza subpusilla* *Lemurimyza lycopersicae* *Liriomyza canomarginis* *Liriomyza guytona* *Liriomyza minutiseta* *Liriomyza munda* *Liriomyza propepusilla* *Liriomyza pullata* *Liriomyza subpusilla* *Liriomyza verbenicola*
Taxonomic position:	Class: Insecta Order: Diptera Family: Agromyzidae Genus: *Liriomyza* Species: *Liriomyza sativae*

2.1 Background and impact of pest

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. Nearly all species are very host-specific, but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world.

The vegetable leafminer (*L. sativae*) is of concern as it has a wide host range including many horticulture crops in which it can cause significant yield losses and quality reductions (see Appendix 1). Vegetable leafminer has also been reported to vector the potyviruses Celery mosaic virus and Watermelon mosaic virus, both viruses have been reported from Australia and affect celery and cucurbits respectively (Zitter and Tsai 1977). The vegetable leafminer is widespread overseas being present in North and South America, Asia, Africa, and occasional outbreaks in Europe.

This species was first detected in the Torres Strait (August 2008) and has recently been detected on mainland Australia near the north Queensland town of Seisia (May 2015) (IPPC 2015; 2017). There appears to have been multiple introductions of this pest into the Torres Strait (Blacket et al., 2015). This genetic diversity may have management implications. Due to its presence in far North Queensland there is a significant risk that it may be able to spread south into production regions where it may have major impacts on Australian plant industries, especially the vegetable, production nursery and melon industries.

2.2 Life cycle

The lifecycle (Figure 1) of the vegetable leafminer consists of an egg inserted by a female just under the surface of a leaf of a host plant. The egg hatches in 2-5 days. The larvae then begin to feed within the leaf creating tunnels or mines that get larger as the larvae moult and mature. After passing through three larval stages in 4-7 days, the larva leaves the plant to form a puparium in the soil underneath the host plant. Larvae may also occasionally fail to leave the leaf and pupate on the leaf surface or may potentially fall from the plant to pots/trays underneath. After 7-14 days an adult emerges and begins to reproduce.

The times taken to complete each life stage vary depending on host and temperature. For example, Haghani et al. (2007), studied development times on cucumber at different temperatures. They found that total development time ranged between 40.4 and 9.3 days, with development time decreasing with increasing temperatures. They also reported that the limits of development were 10 and 40°C. Similarly, a study by Cost-Lima et al., (2010) examined vegetable leafminer development on cowpea at different temperatures and relative humidity levels. They found that adult longevity decreased with increasing temperatures.

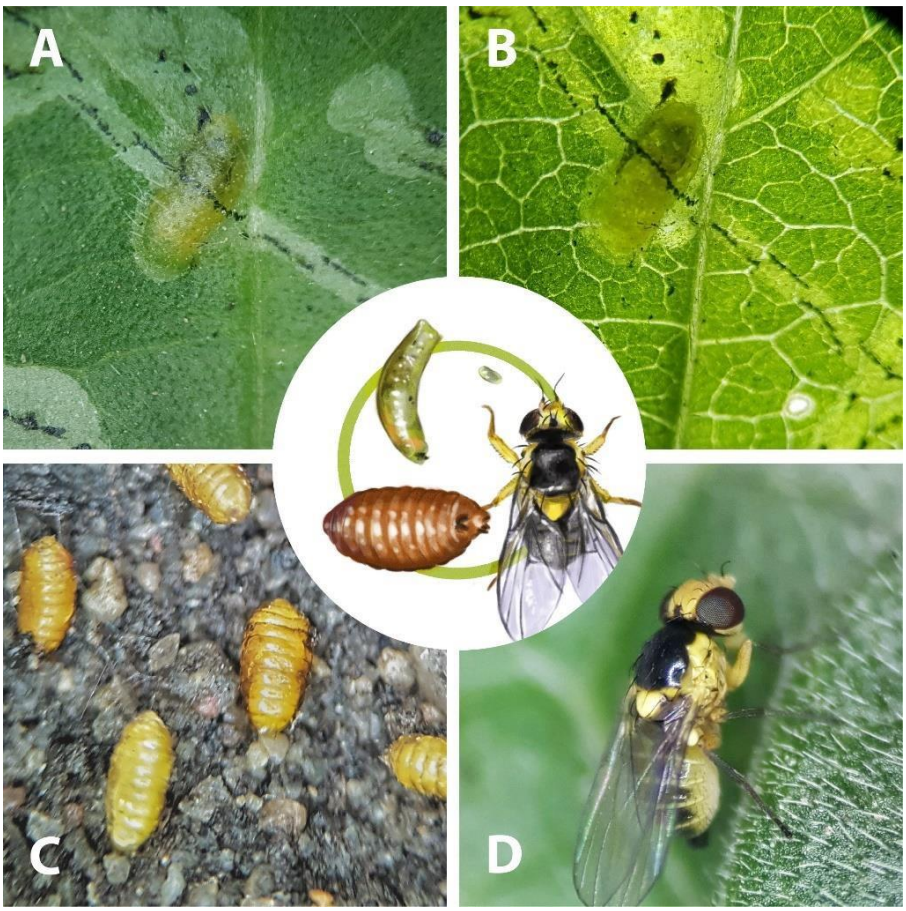


Figure 1 A larvae visible within a leaf mine, with top down lighting (A), and backlighting (B); pupa in the soil (C) and adult (D). Images: Elia Pirtle, Cesar Australia *Pty Ltd*

2.3 Host range

Vegetable leafminer is a polyphagous pest of many agricultural and ornamental hosts (see Appendix 1). It has been recorded from 120 plant species representing at least 23 plant families, although its preferred hosts tend to be in the Cucurbitaceae, Fabaceae and Solanaceae families (Spencer 1973, 1990).

Important agricultural hosts include: brassica vegetables, leafy vegetables (e.g. spinach and lettuce), cucurbits (e.g. pumpkins, cucumbers), melons, legume vegetables and pulse crops, tomatoes, potatoes, capsicum, a wide range of ornamental species (e.g. petunia, chrysanthemum and marigold), cotton, lucerne, herbs (e.g. basil), onions and clover..

A detailed host list is included in Appendix 1.

2.4 Signs and symptoms

Adult flies are small (1.5-1.6 mm long with a wing length of 1.3-1.8 mm) with a shiny black mesonotum and yellow markings on the head and body (Figure 2).

Female flies use their ovipositor to puncture the leaves of the host plants causing wounds which serve as sites for feeding (by both males and females) or oviposition. Feeding punctures appear as white speckles on the upper leaf surface and are ~0.2 mm in diameter (Figure 4). The appearance of the punctures does not differ between *Liriomyza* species so cannot be used to separate species.

Pale coloured leaf mines are created by the feeding larvae and are the most obvious symptom of infestations of *Liriomyza* spp. Larvae produce leaf mines that are serpentine and increase in size along their length (Figure 3 and Figure 4).

Factsheets with more information on what to look for in the field can be found at the following websites:

- www.planthealthaustralia.com.au/pests/vegetable-leaf-miner/
- www.business.qld.gov.au/industries/farms-fishing-forestry/agriculture/crop-growing/priority-pest-disease/vegetable-leafminer
- www.agriculture.gov.au/biosecurity/australia/naqs/naqs-target-lists/vegetable_leaf_miner
- http://entnemdept.ufl.edu/creatures/veg/leaf/vegetable_leafminer.htm
- https://ausveg.com.au/app/uploads/2019/07/Vegetable-leafminer.pdf



Figure 2. Adult vegetable leafminer. Image: Elia Pirtle, Cesar Australia *Pty Ltd*



Figure 3. Vegetable leafminer leaf mines on siratro left(left) and cucurbit (right,), note mines increase in width as the larvae ages. Images: Elia Pirtle, Cesar Australia *Pty Ltd*



Figure 4 Stippling and leaf mines on snake bean caused by vegetable leafminer. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

2.5 Dispersal

Typically, *Liriomyza* leafminers are considered to have invaded countries via the movement of infested plants (generally ornamentals such as chrysanthemum) (Spencer 1989). While fully formed mines should be readily visible, signs of early infestations are much less obvious and are easily overlooked (Spencer 1989).

Agromyzid flies are considered as "moderate fliers" (Yoshimoto and Gressitt 1964) and in agricultural situations, the flies tend to remain close to their target crops, often only moving very short distances between host plants (Zehnder and Trumble 1984a; Fenoglio et al., 2019). Lei et al., (2002) reported that in a flight mill experiment vegetable leafminer could fly up to 8.22 km but average flight distances of <1.0 km were reported at 18-36°C. Moreover, agromyzids such as vegetable leafminer may have the capacity to move longer distances by wind dispersal (White 1970; Glick 1960).

However, windborne dispersal is likely to be a minor pathway of leafminer dispersal in Australia, and human-mediated pathways should be prioritised, particularly given the accumulation of research pointing to this as a major contributor to the movement of invasive species (Capinha et al. 2015; Wichmann et al. 2009). Such pathways include transport of contaminated produce, or "hitch-hiking" adults associated with the large volume of human movement.

In recent years, the vegetable leafminer has spread from New Guinea, through several islands in the Torres Strait to Seisia on the northern tip of Cape York Peninsula, possibly by the spread of plant material or natural dispersal. This suggests that there is the potential to spread from Seisia south into other horticulture regions, particularly during the tourist season in Far North Queensland. Plant material should be considered as a potential pathway for the southward spread of this pest. There are currently movement restrictions in place to stop the movement of plant material south from the far north of Cape York Peninsula, however there is still a risk that the pest could spread southwards.

2.6 Current geographic distribution

The vegetable leafminer is thought to have originated in the Americas and since spread to Asia, and Africa. Small outbreaks have also been reported in Europe, which have been eradicated each time (CABI/EPPO, date unknown). The current status of the distribution worldwide is listed in the EPPO (2020) database.

The current distribution of this pest is presented in Table 1.

Table 1. Countries where vegetable leafminer is known to occur

COUNTRY/REGION	REFERENCE
American Samoa	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Antigua and Barbuda	Kroschel et al., (2016)
Argentina	Civelek (2002); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Australia (only reported from Torres Strait, and Far North Queensland)	IPPC (2015; 2017)

COUNTRY/REGION	REFERENCE
Bahamas	Kroschel et al., (2016); PHA (2008)
Bangladesh	Bhuiya et al., (2011)
Barbados	Alam (1985); Kroschel et al., (2016); PHA (2008)
Brazil	Araujo et al., (2013); Kroschel et al., (2016); PHA (2008)
Cameroon	Civelek (2002); Kroschel et al., (2016); Martinez and Bordat (1996); PHA (2008)
Canada (glasshouses in Ontario)	Kroschel et al., (2016); McClanahan (1980); PHA (2008)
Chile	Gonzalez (1989); Kroschel et al., (2016); PHA (2008)
China	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005); Tran et al., (2007)
Colombia	Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Cook Islands	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Costa Rica	Kroschel et al., (2016); PHA (2008); Spencer (1983)
Cuba	Kroschel et al., (2016); PHA (2008)
Democratic Republic of the Congo[1]	Kroschel et al., (2016)
Dominica	Kroschel et al., (2016); PHA (2008)
Dominican Republic	Kroschel et al., (2016); Martinez et al., (1993); PHA (2008)
Egypt	PHA (2008); Scheffer and Lewis (2005)
Ethiopia[1]	Kroschel et al., (2016)
Federated States of Micronesia	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
French Guiana	Kroschel et al., (2016); PHA (2008)
French Polynesia	Blacket et al., (2015); PHA (2008)
Guadeloupe	Kroschel et al., (2016); PHA (2008); Spencer et al., (1992)
Guam	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Guatemala	Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Honduras	Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
India	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Indonesia	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Iran	Kroschel et al., (2016); PHA (2008); Saryazdi et al., (2014)
Israel	Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Jamaica	Civelek (2002); Kroschel et al., (2016); PHA (2008)

[1] Kroschel et al., (2016) infer presence based on overseas interceptions on crops originating from this country

COUNTRY/REGION	REFERENCE
Japan	Kroschel et al., (2016); Blacket et al., (2015): PHA (2008); Tran et al., (2007)
Jordan	Kroschel et al., (2016); PHA (2008)
Kenya	Gitonga et al., (2010); Kroschel et al., (2016)
Malaysia	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Martinique	Etienne and Martinez (2013); Kroschel et al., (2016); PHA (2008)
Mexico	Kroschel et al., (2016); PHA (2008); Trumble and Alvarado-Rodriguez (1993)
Montserrat	Kroschel et al., (2016); PHA (2008)
Netherlands Antilles	Kroschel et al., (2016); PHA (2008)
New Caledonia	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Nicaragua	Kroschel et al., (2016); PHA (2008); Piedrahita and Fabio (2016)
Nigeria	Civelek (2002); Kroschel et al., (2016); PHA (2008)
Northern Mariana Islands	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Oman	Civelek (2002); Kroschel et al., (2016); PHA (2008)
Panama	Kroschel et al., (2016); PHA (2008)
Papua New Guinea	Blacket et al., (2015)
Peru	Civelek (2002); Mujica and Kroschel (2011); Kroschel et al., (2016); PHA (2008)
Philippines	PHA (2008); Scheffer and Lewis (2005)
Puerto Rico	Acosta et al., (1986); Kroschel et al., (2016); PHA (2008)
Samoa	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Saudi Arabia	PHA (2008); Scheffer and Lewis (2005)
South Africa[2]	Kroschel et al., (2016); PHA (2008)
Sri Lanka	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
St. Kitts and Nevis (on the Island of Saint Kitts/Saint Christopher)	Etienne and Martinez (2003); Kroschel et al., (2016); PHA (2008)
St. Lucia	CABI/EPPO (date unknown); PHA (2008)
St. Vincent and Grenadines	Kroschel et al., (2016)
Sudan	Civelek (2002); Kroschel et al., (2016); Martinez and Bordat (1996); PHA (2008)

[2] Kroschel et al., (2016) infer presence based on overseas interceptions on crops originating from this country

COUNTRY/REGION	REFERENCE
Tanzania[2]	Kroschel et al., (2016)
Thailand	Blacket et al., (2015): Kroschel et al., (2016); PHA (2008)
Timor Leste	Brito and Supartha (2016); PHA (2008)
Trinidad and Tobago	Kroschel et al., (2016); PHA (2008)
Turkey	Civelek (2002); Kroschel et al., (2016); PHA (2008)
United States of America (various mainland states and Hawaii)	Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Uganda[2]	Kroschel et al., (2016)
Uzbekistan	Drugova and Zlobin (2003); Kroschel et al., (2016); PHA (2008)
Vanuatu	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008)
Venezuela	Civelek (2002); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005)
Vietnam	Blacket et al., (2015); Kroschel et al., (2016); PHA (2008); Scheffer and Lewis (2005); Tran et al., (2007)
Yemen	Civelek (2002); Kroschel et al., (2016); PHA (2008)
Zimbabwe	Kroschel et al., (2016); PHA (2008)

2.7 Risk of establishment in Australia

Detailed studies of overwintering of vegetable leafminer pupae in China found no indication of pupal diapause and suggested the -2°C isotherm of the minimum mean temperature of January (northern hemisphere winter) was the overwintering range limit for vegetable leafminer (Chen & Kang 2005 a, b).

Cesar Australia has created a model of the potential geographic distribution of the vegetable leafminer in Australia, the results of which are summarised below (Figure 5). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

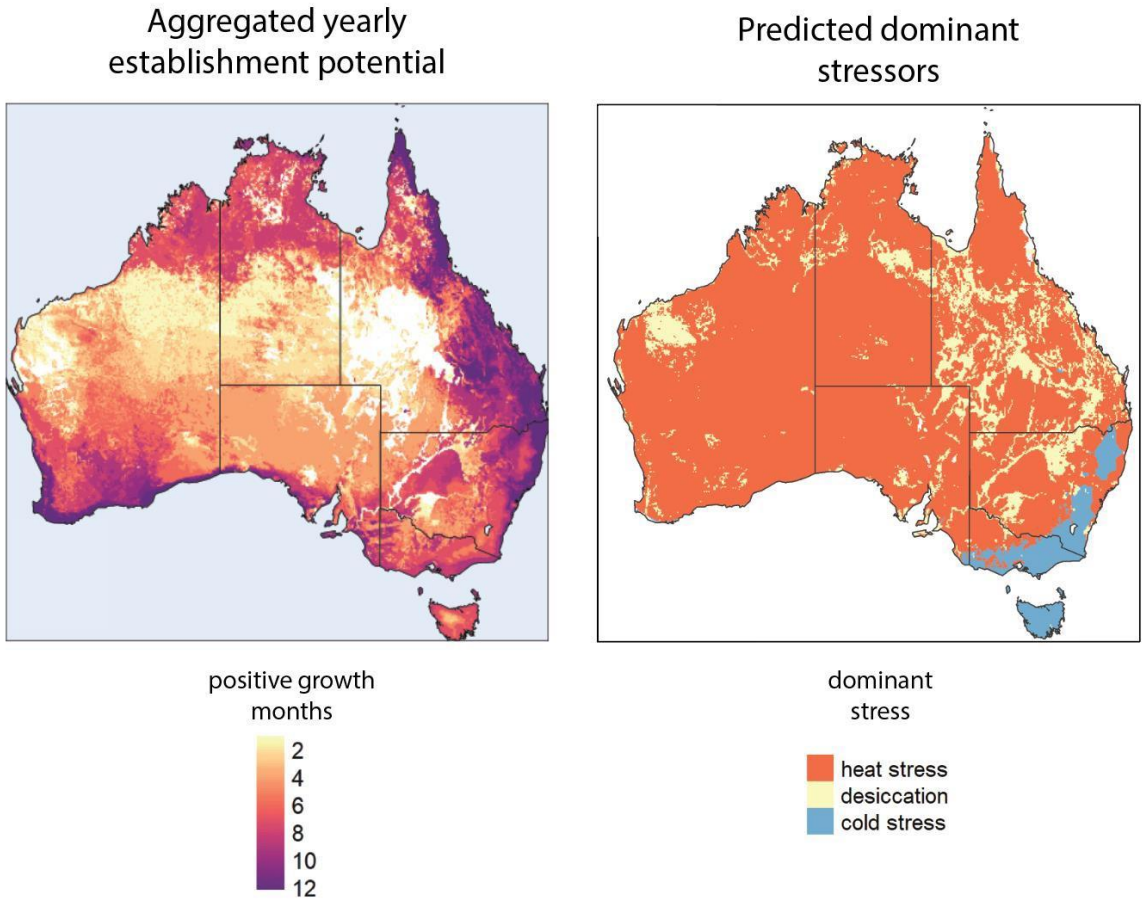


Figure 5 Aggregated yearly establishment potential of vegetable leafminer as the number of months across the year with increasing population sizes) based on temperature and moisture constraints (left) and predicted dominant stressors based on the highest mortality rate from desiccation, cold, and heat stress across the year (right).

2.8 Risk of spread within Australia

2.8.1 Risk pathways – international and domestic

This species could potentially enter new areas by natural spread and by hitchhiking on goods, aircraft, vehicles, or the movement of plant material (DAWR 2016). Table 2 lists some of the potential international risk pathways and control measures in place. These may also broadly applicable to other *Liriomyza* species.

Table 2 International risk pathways for the spread of the pest and control measures

RISK PATHWAY	DESCRIPTION	CONTROL MEASURES
Cut flowers on regulated imports	Infested cut flowers moved to residential dwellings that contain suitable hosts and the insect transfers to a host.	Border inspections
Infected plant material on passenger or mail pathways	Infested plant material moved to residential dwellings that contain suitable hosts and the insect transfers to a host.	Border inspections
Natural dispersal from PNG	Ongoing spread from PNG to Torres Strait and/or Far North Queensland via traditional movements, winds or flight.	Ongoing surveillance via the Northern Australia Quarantine Strategy Domestic quarantine
Infested plant material and natural spread of vegetable leafminer from FNQ	Plant material or pupae in soil spread to new areas in baggage, vehicles or camping equipment.	Domestic quarantine

Additionally, as the larvae of *Liriomyza* spp. leave the plant to pupate, pupae may also be spread with crop debris or soil associated with infested areas. Table 3 lists some of the potential risk pathways and potential domestic control measures to reduce the spread of vegetable leafminer. These are also broadly applicable to other *Liriomyza* species.

Table 3. Risk pathways for the spread of the pest and potential domestic control measures

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Cut flowers (host plants)	Cut flowers that are a host of vegetable leafminer (see Appendix 1) pose a significant risk. The life of cut flowers is long enough to allow the completion of the pest's lifecycle (CABI and EPPO, date unknown). Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Hitchhiking adults and pupae may be transported on cut flowers.	The application of systemic insecticides may be required before allowing cut flowers grown in an infested area to be moved outside the infested area. Further information on chemical control is provided in Section 5.3. 3. All cut flowers leaving infested areas will also need to be visually inspected for signs of leafminers before leaving the area.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Cut flowers (non-host plants)	Cut flowers that are not a host of vegetable leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host cut flowers are transported with or spent time in close proximity with host cut flowers. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	To reduce the risk of spread, cut flowers that are not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with vegetable leafminer.).
Nursery Stock (containerised host plants)	Nursery stock that is a host plant for vegetable leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Containerised nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae may also be present in the soil of containerised stock. Larvae emerge from the leaf and pupate in the soil under host plants meaning potting mix/soil can also potentially spread the pest. Larvae very occasionally attach to leaves or surrounding non-soil surfaces to pupate. Adults may potentially spread with containerised nursery stock as a hitchhiker.	Visual inspection of containerised nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae in the soil (see Figure 3, however pupae will be very hard to detect as they are mixed into the substrate) or attached to leaves and container surfaces. To reduce the risk of spread, containerised nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) should also be considered. Further information on chemical control is provided in Section 5.3.3.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Nursery Stock (bare rooted host plants)	Nursery stock that is a host plant for vegetable leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Bare rooted nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae are less likely to be present in bare rooted stock than containerised stock. However, upon emergence, if larvae are unable to find a substrate to bury into, they will attach to any surface available (including tray walls or plant leaves) meaning pupae can potentially still spread via bare rooted stock. Adults may potentially spread with bare rooted nursery stock as a hitchhiker.	Visual inspection of bare rooted nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae attached to leaves or surfaces surrounding the roots (see Figure 3). To reduce the risk of spread, bare rooted nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Further information on chemical control is provided in Section 5.3.3.
Nursery Stock (non-host plants)	Nursery stock that is not a known host plant of vegetable leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host stock spent time in close proximity with host stock. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	Visual inspection of nursery stock for the presence of adults resting on leaves. To reduce the risk of spread, nursery stock that is not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with vegetable leafminer. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered as a precaution to limit the potential for spread.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Non-commercially grown seedlings and plants	People and small businesses selling/trading seedlings at a local level (e.g. at farmers markets, school fetes, roadside stalls, etc.) pose similar threats to those posed by nursery stock, (i.e. plants can spread eggs, larvae, pupae and potentially adults via host plants to new areas). However, being less regulated this is a difficult pathway to control and therefore potentially poses a higher risk than commercial production nursery businesses.	Should an incursion occur, effort will be needed to identify those involved in the non-commercial trade of seedlings to reduce the risks that they pose. Similar to commercial nursery stock, host plants should not be moved from infested to non-infested areas without being inspected and treated with an appropriate systemic insecticide. Likewise, non-host crops grown in proximity to host plants should be inspected and/or treated as a precaution if grown in an area known to be infested with vegetable leafminer, as there is a small risk that non-host crops could potentially spread adult leafminers as hitchhikers. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered. Further information on chemical control is provided in Section 5.3.3.
Plant material including hay, plant debris and crop waste	Hay, plant debris and crop waste could potentially spread pupae and adults to new areas. Larvae are not expected to be able to survive long after plant debris begins to wilt and rot (once they cannot feed on the plant, the larvae must vacate the leaf to begin pupation or die), however pupae will survive in plant debris and crop waste. Adults may also hitchhike on hay, plant debris and crop waste.	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without being treated with an appropriate insecticide. Further information on chemical control is provided in Section 5.3.3.
Soil	Larvae emerge from the leaf and pupate in the soil under host plants (Capinera 2014). Soil therefore poses a potential risk for moving the pest to new areas.	Minimise potential spread by reducing the spread of soil off infested sites (e.g. by cleaning down machinery between properties).
Tools, equipment,	Tools, equipment, and machinery	Clean tools, equipment and

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
and machinery	used on farm in the vicinity of host plants pose a low risk of spreading the pest, however, may transport pupae via soil deposits, or may transport adults.	machinery before moving off infested sites.
Transport vehicles	Vehicles in the vicinity of host plants could allow the pest to hitchhike (in cabins etc.) to new areas (DAWR 2016).	Clean vehicles before moving off infested sites.
Conveyances (includes crates, boxes, bins, pallets)	Crates, bins, pallets, etc. that are clean of soil are unlikely to spread leafminers.	As a precaution, clean conveyances before moving between sites to reduce any risk of spread.
Seed	There are no expected pathways for leafminer spread by the movement of seed.	Not applicable.
Fruit (without leaves)	There are no expected pathways for leafminer spread by the movement of fruit (without leaves). It is expected the process of harvesting and cleaning fruit will remove leaf and soil material that could spread larvae and pupae and would cause adults to disperse rather than hitchhike with fruit to new areas.	Not applicable.
Fresh leafy or green vegetables (including peas)	Mines can occur on leaves of a wide range of green vegetables. There is therefore a risk that leafminer eggs or larvae (and potentially pupae and hitchhiking adults) could be spread to a new area via the movement of infested leafy vegetables. Mines do not occur on fruiting structures, with the exception that *Liriomyza huidobrensis* can form mines in the pods of snow peas and disrupt market access (Gitonga et al. 2010). It is not clear if this risk extends to *Liriomyza sativae*, though there is photographic evidence of *L. sativae* mining in field pea pods (Cranshaw 2005).	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without undertaking suitable treatments to reduce the risk of pest spread.

2.8.2 Predicting spread potential

Cesar Australia has created a model of the potential rate of area invasion by the vegetable leafminer at entry points across Australia, the results of which are summarised below (Figure 6). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

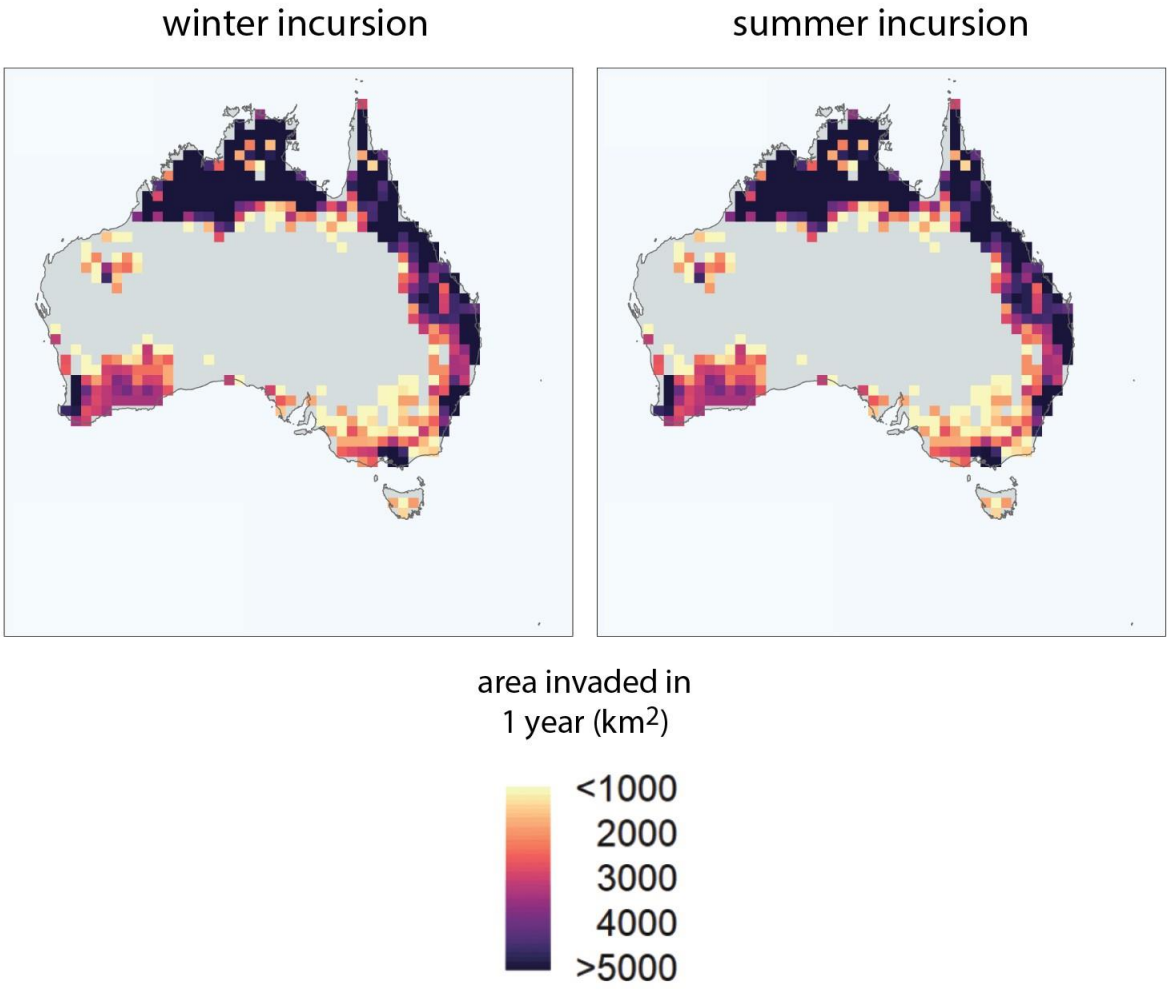


Figure 6 Rate of spread from locations across Australia. For each grid cell an incursion is simulated with the mean predicted area invaded by L. sativae in one year shown by the colour gradient. The mean predicted area is estimated from 10 replicated simulations. Incursions were commenced in July for a winter incursion and January for a summer incursion.

2.9 Risk of economic impact within Australia

Cesar Australia has created a model of the potential economic impacts of the vegetable leafminer in Australia, the results of which are summarised below (Table 4, Table 5 and Figure 4Figure 7). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

Table 4 Impact potential of Vegetable leafminer on affected commodities in terms of international collated reports of proportions crop value lost due to direct feeding damage.

Host crop	Proportion crop value lost			Sources
	Min	Max	Mean	
Beans	0.10	1.00	0.55	Mujica 2016
Capsicum	0.18	1.00	0.18	Wolfenbarger et al. 1963
Celery	1.00	1.00	1.00	Guzman et al 1979
Cucumber	0.30	1.00	0.30	Zhao and Kang 2000
Potatoes	0.10	1.00	0.43	Wolfenbarger et al. 1948; Mujica 2016
Tomatoes	0.02	1.00	0.23	Johnson et al. 1983; Wolfenbarger et al. 1966; Schuster 1977; Waterhouse and Norris 1987; Wyatt et al. 1984; Mujica 2016
Alfalfa	0.80	1.00	0.80	Spencer 1973
Eggplant	0.10	1.00	0.55	Mujica 2016
Musk melon	0.30	1.00	0.30	Zhao and Kang 2000
Okra	0.10	1.00	0.55	Mujica 2016
Peas	0.10	1.00	0.55	Mujica 2016
Squash	0.01	1.00	0.17	Sharma et al. 1980
Watermelon	0.30	1.00	0.65	Palumbo 2020; Zhao and Kang 2000

Table 5 Australia wide accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. sativae *at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preferences hosts may be overestimated while impacts of high preference hosts may be underestimated. Predictions were made using ESIM framework and are the culmination of the establishment, spread and impact modules (with each level of risk building upon the previous). For impacts calculated at state levels, see Appendix 4, Table 18 for additional information. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.*

	Incursion location					
Crop	***Bundaberg***	***Darwin***	***Devonport***	***Melbourne***	***Perth***	***Sydney***
Beans	13.23 (0.44)	5.82 (1.36)	3.36 (1.80)	0.87 (0.42)	0.63 (1.35)	4.27 (1.17)
Broccoli	11.70 (1.02)	6.24 (1.73)	8.17 (2.00)	8.91 (0.22)	2.92 (0.73)	5.66 (1.90)
Brussels	1.86 (1.06)	0.41 (0.32)	1.52 (0.27)	1.28 (0.06)	0.03 (0.08)	0.67 (0.19)
Cabbages	6.53 (0.24)	3.94 (0.69)	2.69 (1.12)	1.59 (0.49)	0.92 (0.58)	4.59 (0.64)
Capsicum	5.76 (0.22)	1.02 (0.48)	2.61 (0.36)	0.81 (0.08)	0.90 (0.02)	0.94 (0.58)
Carrots	9.32 (0.56)	7.46 (2.52)	6.98 (2.54)	0.59 (0.48)	7.48 (1.13)	4.34 (0.96)
Cauliflowers	5.13 (0.55)	2.82 (0.78)	4.55 (0.99)	4.35 (0.14)	1.04 (0.27)	2.98 (0.88)
Flowers	31.46 (1.22)	17.69 (1.53)	21.14 (4.83)	16.83 (2.75)	5.18 (1.82)	25.52 (1.27)
Lettuces	16.37 (1.29)	8.08 (1.39)	11.32 (2.40)	10.83 (0.48)	3.73 (1.03)	8.11 (2.03)
Melons	4.90 (0.27)	1.68 (0.46)	0.20 (0.38)	0.02 (0.02)	1.14 (0.11)	0.31 (0.10)

Crop	Incursion location					
	Bundaberg	***Darwin***	***Devonport***	***Melbourne***	***Perth***	***Sydney***
Nurseries	73.02 (2.58)	39.89 (5.61)	36.68 (10.16)	30.38 (4.10)	14.43 (4.82)	43.98 (4.01)
Onions	5.77 (0.38)	3.83 (1.41)	6.13 (1.30)	0.48 (0.29)	2.77 (0.63)	2.40 (0.57)
Peas	0.82 (0.09)	0.17 (0.19)	1.22 (0.20)	0.39 (0.11)	0.01 (0.02)	0.27 (0.16)
Potatoes	10.59 (0.65)	4.18 (1.65)	12.43 (1.99)	3.76 (0.64)	3.34 (0.43)	4.50 (0.89)
Pumpkins	4.27 (0.19)	2.13 (0.59)	0.64 (0.60)	0.14 (0.16)	0.53 (0.43)	1.52 (0.42)
Tomatoes	19.40 (1.04)	3.98 (1.53)	4.97 (1.70)	5.16 (0.89)	0.40 (0.30)	5.10 (1.22)
Total	**220.12 (6.35)**	**109.34 (17.84)**	**124.60 (27.90)**	**86.40 (9.78)**	**45.45 (13.02)**	**115.19 (14.71)**

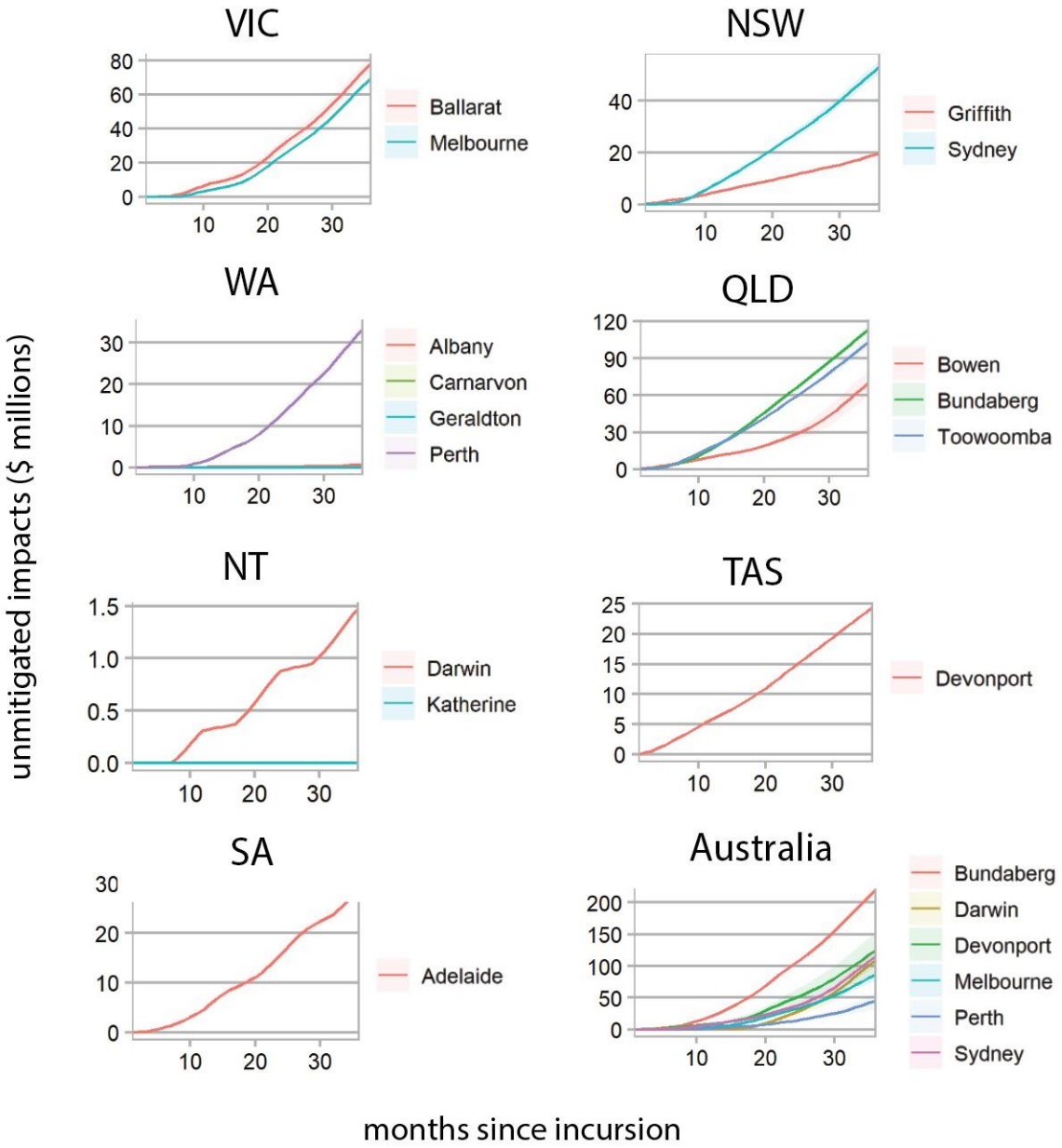


Figure 7 State level accumulating unmitigated impacts in millions of dollars across all host crops after 3 years resulting from a spring incursion of L. sativae at key entry and establishment points. For state level impact accumulation by crop type, see Appendix 4, Figure 11 for more information.

3. DIAGNOSTIC INFORMATION

Morphological diagnosis to a species level requires adult male flies. These can be collected by sweep netting, yellow sticky traps or collecting leaves with larvae in them and allowing the larvae to develop into adults. Alternatively, molecular tools can be used to identify larvae, pupae and adult flies.

An International Plant Protection Convention (IPPC) Diagnostic Protocol (IPPC 2016) and a European and Mediterranean Plant Protection Organization (EPPO) diagnostic protocol for *Liriomyza* spp. (EPPO 2005) are available and should be referred to for the diagnosis of suspected *Liriomyza* spp. It should be noted that these protocols do not include information on environmental DNA (eDNA). Additional information on the use of eDNA for *Liriomyza* spp. leafminer diagnosis can be found at Pirtle et al., *in press*.

An information portal for polyphagous agromyzid leafminer identification is available at https://keys.lucidcentral.org/keys/v3/leafminers/index.htm, which includes species-specific fact sheets, and a pictorial Lucid3 key that allows users to make a preliminary identification of pest agromyzids and to be able to distinguish these species from non-pest endemic agromyzids. A factsheet for *Liriomyza sativae* is accessible here: https://keys.lucidcentral.org/keys/v3/leafminers/key/Polyphagous%20Agromyzid%20Leafminers/Media/Html/Liriomyza_sativae.htm

3.1 Diagnostic considerations

Considerations when triaging samples of suspected vegetable leafminer for diagnostics include:

- One of the greatest challenges in detection and subsequent management of *Liriomyza* species overseas has been the difficulty in correctly identifying the species (Reitz et al., 2013), which can have considerable overlap in adult morphology, host range and damage symptoms left on a plant. In the presence of indigenous leafminer, it may be difficult to detect a new incursion, or determine the limits of an infestation (Powell 1981).
- There are also several difficult to distinguish Agromyzid species present within Australia that are morphologically indistinguishable from vegetable leafminer to a non-expert (including *L. brassicae* and *L. chenopodii*). This is also true for some Agromyzids that are exotic to Australia (including *L. trifolii* and *L. huidobrensis*). In many cases, specimens must be distinguished by expert taxonomists, or via molecular means.
- Within Australia, there at least 50 leafminer species (family Agromyzidae) (and presumably many more that have not yet been described) that create leaf mines that look similar to vegetable leafminer and may appear on the same host plants. Some of the most common include *Liriomyza brassicae* (which shares several brassica hosts with vegetable leafminer), *Liriomyza chenopodii* (which shares cultivated beets with vegetable leafminer), and *Phytomyza syngenesiae (*which shares cultivated asters with vegetable leafminer, but the adult form is readily distinguishable from vegetable leafminer) (Appendix 2). Moreover, there are an unknown number of additional Dipteran and Lepidopteran (Figure 10) species that share the leaf mining habit.
- Of these species that create similar leaf mining damage, some can be distinguished from vegetable leafminer via their mine characteristics, and their pupal or adult forms. (see Appendix 2, IPCC 2016, Salvo and Valladares 2004). However, the high degree of individual variation between each leaf mine means there can always be exceptions to these guidelines. For example, while vegetable leafminer pupate outside the leaf, there are rare instances where a larva will fail to emerge and complete pupation within the leaf mine.
- Due to the high degree of morphological overlap of adults and visual overlap of damage, sample collection (of flies of any life stage and/or leaf mines) will be paramount for identifying and delimiting potential vegetable leafminer incursions.

- Surveyors should be familiar with the vegetable leafminer host plant list (Appendix 1), as well as with commonly observed signs of indigenous leafminer damage and the extent of known overlap with indigenous Agromyzids for common host genera (Appendix 2) before conducting vegetable leafminer surveillance. For example, damage caused by the closely related *L. brassicae* is essentially indistinguishable from vegetable leafminer and is very common on brassica crops (Appendix 2). Crops such as tomato, cucumbers and melons, on the other hand, have little to no reported overlap with indigenous Agromyzids (Appendix 2). Mines detected on these crops during surveillance activities should be considered highly suspicious.
- Molecular techniques can be used for diagnostics of Liriomyza spp. IPPC 2016) and have the advantage of being able to be used on all life stages of the insect. If using molecular techniques, it is important that surveyors understand what material to collect and how to preserve specimens appropriately, refer to Section 4.2.2 for further information.

4. SURVEILLANCE AND COLLECTION OF SAMPLES

4.1 Surveillance

4.1.1 Technical information for planning surveys

Detection and delimiting surveys are required to determine the extent of outbreaks, ensure areas free of the pest retain market access, and ensure appropriate quarantine zones are established to contain outbreaks.

Initial surveillance priorities for vegetable leafminer should consider the following:

- Visual surveillance for the leaf mines created by vegetable leafminer is expected to be the most reliable way to detect new incursions. This is because the serpentine leaf mines created by leafminer larvae persist permanently on a leaf, extending the detection window beyond the lifespan of the fly itself. In comparison, sweep netting requires active populations, as do yellow sticky traps. While sticky traps can be left in place for extended periods to increase the chance of catching an adult, in open field settings, relying solely on sticky traps would be impractical due to the very large number of traps that would be needed to be placed and processed, to reach a statistically acceptable detection likelihood for small or isolated populations.
- In closed cropping systems, sticky traps become much more practical, but should still be accompanied by visual surveillance.
- Sticky traps will have a high level of bycatch (i.e. other insects will be caught in the trap), which can increase processing time for manual identification. However, molecular diagnostic methods are also available for bulk invertebrate samples collected via a sticky trap (see Section 4.2.2.2).
- When undertaking visual surveillance in plant crops, the serpentine leaf mines are the most conspicuous symptoms to look for, though stippling from oviposition and feeding may also be apparent.
- Mines are expected to be more common on the edges of fields and in the upper portions of the plant (Weintraub 2001).
- The vegetable leafminer has an extensive list of broad leaf weed hosts (see Section 2.3) and therefore surveillance in broadleaf weeds (such as siratro, castor bean, sow thistles, etc.) as well as cultivated crops should be conducted.
- Larvae, pupae and adults can be identified using molecular methods (refer to Section 3).
- Only adult male leafminers can be identified using morphological methods.
    - Adults can be captured using yellow sticky traps, sweep netting (IPPC 2016), or rearing from infested material. The latter is the most practical and reliable method for collecting adult flies. Infested leaves can be placed into an insectarium to develop into adults (IPPC 2016). Techniques for rearing Agromyzids are described in Griffiths (1962) and Fisher et al., (2005).
- Collectors should be made aware of common signs of leafminers and note symptomatic plants when conducting surveillance for vegetable leafminer.

- Australia is already home to several leaf miner species, both native and naturalised, which can be confused for vegetable leafminer. Surveyors should be made aware of the native and naturalized species they might expect to see in the particular region and crops they are surveying.
- Early detection surveillance will be most effective if timed when vegetable leafminer population growth potential is greatest. The seasonality of pea leafminer activity at major centers is provided in Table 6. A biophysical risk forecasting tool has been developed that allows users to explore changing vegetable leafminer growth potential and spread risk across the year at any location in Australia. The interactive version of this tool can be accessed here.

Table 6 Major Australian growing regions showing risk seasonality for L. sativae *activity across the year. Green means highest establishment risk (where the population growth potential was greater than 50% of the maximum predicted growth rate) yellow means lower establishment risk (where the population growth potential was between 1 and 50% of the maximum), and red means lowest establishment risk (where predicted growth rates are negative, meaning populations are predicted to decline).*

State	Incursion point	Seasonality
		Jan Feb Mar Apr May Jun Jul Aug Sep Oct Nov Dec
Western Australia	Perth	
	Geraldton	
	Carnarvon	
	Bunbury	
	Albany	
Victoria	Thorpdale	
	Melbourne	
	Ballarat	
Tasmania	Smithton	
	Devonport	
South Australia	Mt. Gambier	
	Loxton	
	Adelaide	
Queensland	Toowoomba	
	Bundaberg	
	Bowen	
	Atherton	
Northern Territory	Katherine	
	Darwin	
New South Wales	Sydney	
	Griffith	

Further information on factors to consider when conducting surveillance, including some information

on trap placement in different crops, can be found in the "Early detection toolkit for surveillance of the vegetable leafminer (*Liriomyza sativae*)" a report prepared by Cesar Australia as part of the Horticulture Innovation funded project MT16004. A copy of the report is available by contacting Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

4.1.1.1 Surveillance tools

Yellow sticky traps and sweep netting can be used as supplementary methods to visual searches during delimiting surveillance activities, particularly when suspicious damage has been observed, or for monitoring existing outbreaks.

- Sweep netting is expected to be most effective when employed near recently active leaf mines, and is less effective near old leaf mines (see Figure 8).



Figure 8. Younger mines (on the left) are more white-coloured with more distinct black trails of frass. Frass becomes less distinct as mines age, and mines develop a brown colour (right). Younger mines are expected to give the best sweep netting success. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

- Agromyzid flies are attracted to the colour yellow (Chandler 1981, Affeldt et al., 1983, Yudin et al., 1987), and can therefore be captured on yellow sticky traps. Yellow sticky traps are more effective for *Liriomyza* adults than other types of traps, such as funnel traps and yellow water pans (Chavez and Raman 1987). Vacuum sampling has also been shown to be a less effective means of sampling adult leafminer (Weintraub 2001).
- A great deal of effort overseas has been dedicated to improving the effectiveness of yellow sticky traps for *Liriomyza* adults (Heinz et al 1992) including the modifications of size, shape, adhesives, lures, height and orientation. For example, several studies report a strong effect of trap height on the number and species trapped (Trumble and Nakakihara 1983, Zehnder and Trumble 1984a, Zoebisch and Schuster 1990), however these results do not always appear consistent and may be difficult to extrapolate across different crop types. Moreover, optimal height may vary considerably between *Liriomyza* species (Zehnder and Trumble 1984a). See the "Early detection toolkit for surveillance of the vegetable leafminer (*Liriomyza sativae*)" for more information. General guidelines are:
    - Traps should be placed (vertically oriented) at the edges of paddocks, preferably on the upwind side of paddocks, and near broadleaf weed reservoirs if possible.
    - Optimal trap height may vary with crop type, but generally speaking traps should be placed around canopy height.

- Traps should be placed in the afternoon, and left for a minimum of 24 hours, to ensure traps are active during the peak flight time for vegetable leafminer, which occurs between 7:00am and 11:00am.
- Traps may be left in place for up to one month, at which point they should be collected, to prevent excessive degradation of DNA and allow for molecular confirmation of vegetable leafminer caught on the trap. If traps are placed outdoors and exposed to the elements and/or high levels of bycatch they should be collected sooner than one month.

- Experimental lures developed from the extracted volatiles of known plant hosts have been shown to be attractive to vegetable leafminer in field trapping trials (e.g. Wei et al., 2005). Methyl salicylate has been shown to attract *Liriomyza bryoniae* when used on a yellow sticky trap (Būda and Radžiute 2008), while lures made from spruce, basil, juniper or clove oil have been shown to attract *L. huidobrensis* (Gorski 2005). See the "Early detection toolkit for surveillance of the vegetable leafminer (*Liriomyza sativae*)" for more information.

4.1.2 Delimiting surveys in the event of an incursion into new areas

Following a new detection of vegetable leafminer, the following guidelines are recommended for delimiting surveillance:

- Surveillance to delimit a detection of vegetable leafminer should take into account tracing information as outlined in Section 5.1 to determine potential pathways for movement of material from the site of the initial detection.
- Surveillance should be a combination of the following:
  - Visual inspection in high risk areas (e.g. areas with suitable hosts).
  - Sweep nets to collect adults around infested host plants.
  - Yellow sticky traps, particularly in closed cropping systems and near infested plants.
- Surveillance should be accompanied with awareness material, signs and personal visits to households and businesses within the surveillance zone and any buffer zones.

The suggested surveillance protocol for vegetable leafminer is as follows:

1. Delimiting surveillance should be conducted immediately upon detection of any incursions within a radius within 1 km of the incursion site, based on natural spread dynamics of agromyzid leafminer. The Surveillance zone may be increased as deemed necessary for the individual response.
2. At each site, choose a crop that is a preferred host plant of vegetable leafminer (refer to Section 2.3).
3. Choose a survey path that preferences the following areas if possible:
   - Paddocks and weeds near transport routes and unloading areas. These are preferred, because a major long-distance pathway of leafminer spread is human assisted movement.
   - Paddock edges should be included in the survey path, with the 'incoming wind side' preferred, as short distance movements of leafminer can be driven by wind.

- Weeds along paddocks should be included in the survey path (broadleaf weeds, but not grasses).

4. Leafminer damage may be patchy at low densities.
5. At low densities, leaf mines have a relatively low detectability, despite their characteristic appearance. Slower search speeds will improve detection likelihood, however, when time is limited, search speed trades off with the total area covered (reducing the number of possible encounters). This trade-off is important in realistic paddock monitoring scenarios.
    - *When time is not limited*: To achieve a detection probability of 20%, 50%, and 80% respectively, when leaf mine density is about 1% of plants affected, the mean (and 95% CI) search time required per metre is 11.2 (4.8, 20.5), 34.8 (15.0, 63.8), and 80.8 (34.9, 148.2) seconds spend 1 minute searching per 1 m^2 of area (based on data collected during vegetable leafminer survey effort trials).
    - *When time is limited*: The optimal search speed, balancing area coverage with sensitivity, when leaf mine density is about 1% of plants, is about 10 seconds per 1 m^2 of area. Further benefits are not achieved by increasing search speed beyond this level to achieve greater area coverage.
6. A survey plan should take into consideration any variables regarding the environment or surveyor that might reduce detectability and increase the effort spent accordingly. These variables might include:
    - *Plant type*
        i. *Leaf density:* leafminer damage may be harder to spot in plants that are very dense and leafy, than in plants with larger, sparser or flatter leaves.
        ii. *Plant age:* it may be easier to spot leaf mines in young plants with few leaves and more open space between plants than in larger mature, overlapping plants.
        iii. *Colour:* leafminer damage may be harder to spot against light coloured leaves than against darker leaves.
        iv. *Height*: Leafminer damage in plants that are at eye level may be easier to spot than crops that are low to the ground.
        v. *Time of day/direction of survey relative to the sun*: Survey success may be higher when surveyors are walking with their back to the sun, rather than walking into the sun.
    - *Residues on plants:* Chemical residue on plants may reduce the contrast between the mine and the leaf, or obscure mine patterns with background residue patterns.
    - *Damage from other pests/disease on plants:* damage caused by other pests or disease may obscure leaf mines, or lead to false positive detections.
    - *Surveyor experience:* Inexperienced surveyors may have a learning curve.
    - *Weather conditions:* Surveys during poor weather should be avoided when possible.
7. If suspicious damage is detected, samples should be collected (see Section 4.2), and yellow sticky traps should be placed around the affected area, in an attempt to capture adults and diagnose the fly responsible for the damage.
    a. If vegetable leafminer are confirmed, visual surveillance along with sticky traps should be used to monitor the edges of the outbreak zone.

8. If no suspicious damage is detected, the initial round of delimiting surveillance should be followed with a second round occurring after one week.
    a. This second round is necessary as leafminer eggs are undetectable in the leaf, and hatch after about 2-5 days. A crop that appears clean during the initial survey may still develop symptoms within a week.
    b. After the first revisit, any further revisits should occur on a monthly basis. One lifecycle of the vegetable leafminer is estimated to take 2 to 4 weeks.

4.1.3 Activities for general surveillance immediately following a detection

Given the wide host range of this pest and its noticeable impact it may be a suitable target for general surveillance programs that request submission of images and/or samples from the public.

To establish a general surveillance program, the following will be required:

- Use of online or app reporting tools such as MyPestGuide to promote the submission of reports of suspected detections.
- Factsheets to provide information on the pest, symptoms, impacts and reporting mechanisms.
- Media releases to describe the impact of the pest, surveillance programs and activities within the response program.
- Information for industry communication channels including articles for industry newsletters, magazines and websites, information for Twitter feeds and Facebook, and presentations for industry talks.
- A website to provide information for the public and for commercial businesses and links to other relevant sites.
- Release and promotion of information on details for physical sample collection and submission, as well as information on how to take and submit images of flies/leaf mines
- Broader awareness campaigns should consider literature (brochures and factsheets) in several languages.
- A non-expert friendly surveillance and sample collection guide for vegetable leafminer, as well as other useful information, can be found here.

4.2 Collection and treatment of samples

4.2.1 If collecting for morphological diagnosis

Of the four life stages (egg, larva, pupa and adult) only adult males are identifiable to a species level using morphological features.

4.2.1.1 Collection of adult male specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the foliage. However, the most practical and reliable method is the collection of leaves with mines containing pupae or mature larvae which can then be reared until adult flies emerge (refer to Section 4.2.1.3).

4.2.1.2 How to collect sticky traps for identification

Sticky traps should be left in the field for up to one month (but preferably 2 weeks or less), to allow morphological identifications. Upon collection, sticky traps should be folded lightly in half sticky side inwards (not pressed shut), and placed in a sealed, padded plastic bag. Traps should be sent within a week of collection (and kept in cool, dry place in the meantime, but not frozen).

4.2.1.3 How to collect plant samples for rearing

For rearing of adult flies, mined leaves containing pupae or mature larvae can be collected into an insectarium and kept in a constant-temperature room for regular checking. Simple insectariums include sealed partially inflated ziplock bags, or sealed rigid plastic containers or jars. A piece of paper towel should be placed into the insectarium along with the infested leaves to regulate humidity. Once pupae emerge from the leaves, they should be transferred carefully with a wet paintbrush to a new rigid container, which contains a layer of damp sand but no leaf material. Pupae can be placed directly onto the damp sand. Techniques for rearing Agromyzids are described in the IPCC diagnostic protocol for *Liriomyza* (IPPC 2016, available from here), in Griffiths (1962) and Fisher et al., (2005).

4.2.1.4 How to preserve plant samples for identification

Identifying the plant species affected by suspected vegetable leafminer damage can aid diagnoses, particularly when surveying in weeds. To preserve a plant for identification, leaves can be stored between sheets of dry newspaper. However, if plant material with suspected leaf mines is pressed and dried, there will be no chance of rearing out adult flies from those leaf mines. Therefore, whenever plant identity is in question, infested material should be stored in an insectarium for rearing, and an accompanying pressed sample of uninfested leaves should be taken to facilitate for identification of the host.

4.2.1.5 How to preserve leafminers

Adults caught in sweep nets or reared in insectariums can be placed in 70% ethanol and stored indefinitely for morphological purposes. For adults reared in insectariums, it is advised to collect these adults in the afternoon, as emergences tend to occur in the morning, and enough time should be given for wings to fully expand before collection.

4.2.1.6 How to transport leafminers

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.1.7 How to transport plant samples

Plant samples may be collected if the host needs to be identified. Queensland Herbarium (2016) provides information on how to collect and preserve herbarium specimens. Samples should then be taken to state herbaria for identification.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.2 If collecting for molecular diagnosis

All life stages can potentially be diagnosed using molecular methods, including empty leaf mines.

4.2.2.1 Collection of specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the foliage. However, as other life stages can be diagnosed using molecular methods, leaves with mines containing pupae or mature larvae should also be collected for analysis. Rearing these life stages in an insectarium is not necessary.

4.2.2.2 How to collect sticky traps

Upon collection, sticky traps should be either:

- Placed inside a plastic sleeve that has been cut along one long side and along the bottom, the trap and sleeve can then be sealed in a plastic zip lock bag.
- Folded in half, sticky side inwards, and placed in a sealed plastic bag.

Traps should be sent within a week of collection (and kept in a cool, dry place in the meantime, but never frozen).

4.2.2.3 How to collect plant samples

Plant samples with suspected vegetable leafminer damage should be collected for molecular confirmation. If it is possible to discern a live larvae inside a leaf mine (which can be spotted through the leaf mine as a small yellow maggot, either by eye or with a hand lens, see Figure 9), these samples are always preferable. However, mined leaves that appear empty can also be collected. At least three samples should be collected to provide a 90% probability of detecting trace DNA that is present (assuming three technical qPCR test replicates are conducted, and the mines were stored into 100% ethanol immediately after collection) (Pirtle et al *in press*). Empty mine samples that look youngest should be preferred, as they have the greatest chance of containing a live larva, however DNA can successfully be extracted even from mines that are up to one month old (See Figure 8).



Figure 9. A larva inside a leaf mine. Larvae are visible to the eye in later stages of development. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

4.2.2.4 How to preserve plant samples

Mined leaves containing pupae, mature larvae, or mined leaves that appear empty can all be placed straight into 100% Ethanol. Small mined leaves should be rolled and placed into the tube of absolute ethanol. For larger leaves, the mine should be cut out, with care taken to avoid cutting across the mine, and the cut out should be rolled and placed into the tube of absolute ethanol. These samples should be frozen and mailed within a week of collection.

Due to the very small amounts of DNA within leaf mines, the odds of sample contamination is much higher than when working with whole flies. Careful hygiene must be employed when collecting and preserving suspicious leaf mine samples. Mines from different host plants or different locations should never be stored in the same container. Samples should either be immediately preserved into on site, or should be placed into unused, clean, sealed plastic bags on site and preserved within the day. If samples from multiple hosts or sites are being processed together at a field lab location, all equipment must be sterilised between each host/location (i.e. cleaning counter tops, sterilizing tweezers and other tools).

Leaf mines may also be preserved onto FTA cards, however DNA quality is somewhat lower than for preservation into 100% ethanol (Pirtle et al *in press*). To preserve leaf mines onto FTA cards, remove the mined leaf from plant and push the leaf mine face down onto the FTA card. Smear the mine thoroughly on the FTA card, for about 30 seconds, and then close the card and place in small sealed plastic bag. Do not rub mines from different host plants/sites onto the same FTA card, and do not store FTA cards from different host plants/sites in the same sealed bag. Store the FTA cards in a cool, dark, dry place and send for analysis within a week of collection

4.2.2.5 How to preserve leafminers

Specimens required for molecular diagnostic work, including larvae, pupae or adults, should be killed and preserved in absolute ethanol, frozen at -80°C, or preserved on an FTA card.

4.2.2.6 How to transport leafminers and plant samples

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

5.IMMEDIATE RESPONSE TO DETECTION

5.1 Tracing

Detection and delimiting surveys are required to delimit the extent of the outbreak, ensure areas free of the pest retain market access, and ensure that appropriate quarantine zones are established.

Extensive tracing (trace forward and trace back) may be feasible as *Liriomyza* spp. can be readily dispersed by the movement of infested plant material, soil and crop debris. The focus should be on high risk linkages including premises linked directly with the initial detection, particularly where

movements of plant material (such as nursery plants and seedlings), or soil have occurred as these pathways may move vegetable leafminers over long distances.

Further information on possible risk pathways are presented in Section 0 and in Table 3.

5.2 Quarantine and movement controls

If Restricted or Quarantine Areas are practical, no plant material should be moved from the infested to non-infested areas without first being inspected and appropriately treated. The size of the Restricted or Quarantine Area will be dependent on the type and scale of the incursion. However, a zone 1 km in radius should be considered for initial delimiting surveillance activities Section 4.1.2). This zone can then be expanded as needed depending on the specific situation.

Voluntary movement control should be considered for urban/residential detections. Voluntary controls would involve negotiation with residents to undertake inspection and treatment of goods prior to movement from infested areas. Residents should be advised on measures to minimise the inadvertent transport of the pest from the infested area to non-infested areas. Voluntary compliance is likely to be implemented for urban areas using awareness campaigns to highlight high risk goods/situations and appropriate treatments.

5.3 Destruction strategy

5.3.1 Priorities and considerations

If eradication of an outbreak is considered feasible, the priority will involve the destruction of the pest. Adult leafminers may be present around host plants; eggs, larvae and pupae may be present in the leaves of host plants, and pupae may be present within the first 5 cm of soil under host plants.

Considerations include:

- The infested plants may need to be sprayed with a suitable insecticide (refer to Section 5.3.3), then bagged and then buried (or tilled/ploughed directly into the soil if a large area is affected).
- Other host plants in the immediate area should be inspected and treated as needed.
- Soil and plant media in greenhouses should be treated (heat or chemically) to destroy any pupae that may be present in the soil.
- Machinery and equipment used on site may need to be washed down to minimise the potential risk of spreading the pest between sites.

5.3.2 Decontamination of tools and equipment

Machinery, equipment, tools and clothes should be cleaned and disinfected between sites to minimise the potential to spread vegetable leafminer, and other pests and diseases that may be present on the site, to new areas.

For points to consider when decontaminating, refer to the PLANTPLAN Guideline document:

Disinfection and Decontamination (PHA, 2015[3]). which includes guidelines for the destruction and decontamination of tools used when working with emergency plant pests.

Several state agriculture departments have also produced publications on cleaning down machinery and vehicles and should be referred to if decontamination is required, examples include:

- www.dpi.nsw.gov.au/ data/assets/pdf_file/0010/545554/procedure-decontamination-vehicles-and-equipment.pdf.
- www.daf.qld.gov.au/ data/assets/pdf_file/0011/58178/cleandown-procedures.pdf.
- http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/weeds/weedstop-vehicle-hygiene-program/machinery-hygiene.

5.3.3 Chemical control

Overseas, *Liriomyza* leafminers are most damaging when parasitoids are removed due to the use of non-selective insecticides (Cesar Australia 2018a; Murphy and LaSalle 1999). For the ongoing management of leafminers it will therefore be important that chemicals are selected that are reasonably selective and have low toxicity towards key parasitoids.

EPPO (2009) provides guidance on how to manage and eradicate vegetable leafminer infestations in Europe. The EPPO recommends that foliar sprays are applied at weekly intervals until control has been achieved. Importantly they suggest rotating chemicals and modes of action (where possible) each treatment (Table 7). In greenhouse environments the use of a suitable space treatment is also suggested (EPPO 2009). Based on this, it would be ideal if pesticides from different mode of action groups are available for each host crop, as this would allow pesticides to be rotated rather than relying on a single product being continuously used to manage/treat the pest.

[3] www.planthealthaustralia.com.au/biosecurity/incursion-management/plantplan/

Table 7. An ideal chemical rotation schedule used for eradication of Liriomyza leafminers in Europe (refer to EPPO 2009)

WEEK	PESTICIDE USED TO TREAT INFESTED AREA
Week 1	Chemical A
Week 2	Chemical B
Week 3	Chemical C
Week 4 onwards	Repeat above sequence

In 2017-18 Cesar Australia carried out a literature review on the chemical control of vegetable leafminer considering pesticides based on their efficacy and their impact on Integrated Pest Management (IPM) systems (Cesar Australia 2018b). A copy of the report is available by contacting Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

The report notes that vegetable leafminer populations in Indonesia and Vietnam have developed resistance to synthetic pyrethroids. The report also identified several potential pesticides that would fit in with IPM systems (i.e. are not too damaging to biocontrol organisms) and are effective at controlling the pest overseas.

APVMA permits have been applied for a variety of pesticides based on the literature review's findings and considering the similarity of overseas and Australian use patterns with respect to rate, application methods, etc. Table 8 provides a summary of the permits that have been granted to date and also notes mode of action, and leafminer life stages targeted by the pesticide.

As the vegetable leafminer can affect many plant species (including agricultural and ornamental species) (refer to Section 2.3), it is important that all potential hosts are covered by at least one, but preferably multiple, pesticide permits. Appendix 3 provides a summary of the pesticides that can be used on each crop type and host and should be used as a guide when treating new vegetable leafminer outbreaks, or if eradication is unsuccessful, managing the pest. As permits are approved, they will be available from the APVMA permit database: https://portal.apvma.gov.au/permits or by contacting the APVMA directly.

Table 8. Insecticide permits that have been applied for or granted by the APVMA for the control of vegetable leafminer in Australia (as of July 2020)

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[4]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Abamectin (Mode of Action (MOA) 6)	Contact and translaminar activity	Very effective against adults and larval stages. All larval stages appear susceptible (Cesar Australia 2018b). There are also some reports of ovicidal activity against the related *L. trifolii* (Saryazdi et al., 2012) and *L. huidobrensis* (Mujica et al., 1999).	Fruiting vegetables-cucurbits	600-1,200 ml/ha (suppression only)	PER81876	30/4/2024
			Fruiting vegetables – other than cucurbits (excluding sweet corn and mushroom)	600-1,200 ml/ha (suppression only)		
			Leafy vegetables (except lettuce)	600-1,200 ml/ha (suppression only)		
			Legume vegetables	600-1,200 ml/ha (suppression only)		
			Root and tuber vegetables	600-1,200 ml/ha (suppression only)		
			Bulb onions	600-1,200 ml/ha (suppression only)		
			Cabbage	600-1,200 ml/ha (suppression only)		
			Celery	600-1,200 ml/ha (suppression only)		
			Rhubarb[5]	600-1,200 ml/ha (suppression only)		
			Bulb vegetables except onions (including leeks, spring onion)	600 ml/ha (suppression only)		

[4] Refer to Appendix 4 for detailed information on chemical control options.
[5] Note: Rhubarb is not a recorded host for vegetable leafminer but may be a host of other exotic *Liriomyza* spp. and is part of the 'Stalk and Stem Vegetable Crop Group' that the permit covers.

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[4]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Cyromazine (MOA 17)	Contact and translaminar activity	Not effective against adults but being an insect growth regulator is very effective against larval stages (Cesar Australia 2018b). Experiments on the related *L. trifolii* found that Cyromazine was very effective against first and second instar but less effective against late instar larvae (Yathom et al., 1986).	Nursery stock (non-food)	333 g in 20 L water/10 tonne potting mix 950 g/ha or 95 g/100 L applied in 1,000 L/ha.	PER83506	31/10/2022
			Fruit trees (non-bearing)			
			Broccoli	1 kg/ha or 150g/100L water	PER81867	30/11/2023
			Fruiting vegetables – cucurbits			
			Fruiting vegetables – other than cucurbits (excluding mushrooms and corn)			
			Head lettuce			
			Legume vegetables			
			Root and tuber vegetables			
			Stalk and stem vegetables			
			Brassicas (excluding broccoli) **Note: this crop must be destroyed if treated and must not be made available for human consumption**			

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[4]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Cyromazine (MOA 17)	Contact and translaminar activity	Not effective against adults but being an insect growth regulator is very effective against larval stages (Cesar Australia 2018b). Experiments on the related *L. trifolii* found that Cyromazine was very effective against first and second instar but less effective against late instar larvae (Yathom et al., 1986).	Leafy vegetables (excluding head lettuce) **Note: this crop must be destroyed if treated and must not be made available for human consumption**	1 kg/ha or 150g/100L water	PER81867	30/11/2023
Dimethoate (MOA 1B)	Contact and systemic activity	Dimethoate is reported to control *Liriomyza* spp. adults and larvae however resistance is known overseas (Tran and Takagi 2005)	Pulse crop (grains)	75ml (400 g a.i/L products)/100L water	PER89184	31/3/2025
Emamectin benzoate (MOA 6)	Contact and translaminar activity	Same mode of action as abamectin. Reported efficacy against larvae, adults and eggs (**cesar** 2018b)	Brassica vegetables (including broccoli, Brussels sprouts, cabbage, cauliflower)	250 to 300 g/ha (44 g a.i/kg products); or 650 to 780 mL/ha (17 g a.i/L products) (suppression only)	PER87563	30/6/2024

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[4]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Spinetoram (MOA 5)	Contact, systemic and translaminar activity	Spinetoram has shown efficacy against vegetable leafminer adults and larvae (Shimokawatoko et al., 2012)	Vegetable legumes (peas and beans)	400 ml (48 g a.i)/ha	PER87878	28/2/2023
Spirotetramat (MOA 23)	Contact, systemic and translaminar activity	Affects the development of larvae and has little impact on adults (Ring 2019)	Capsicum	400ml (96 g a.i)/ha	PER88640	31/5/2023
			Celery			
			Chili			
			Eggplant			
			Green beans			
			Lettuce (head and leafy)			
			Parsley			
			Rhubarb			
			Snow & sugar snap peas			
			Tomato			

Research undertaken as part of the Hort Innovation project MT16004 has identified the suitability of the following as potential control options for *Liriomyza* spp.:

- Azadirachtin (a systemic insecticide that acts as an insect growth regulator, MOA unknown) on ornamental and floriculture plants.
- Cyantraniliprole (a systemic insecticide, MOA 28) on bulb vegetables, fruiting vegetables, cucurbits, melons, and potatoes (note residue data will be required to support this use pattern).
- Diazinon (a contact insecticide, MOA 1B) on beans, melons, beetroot, lettuce, parsnips, tomatoes, and potatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Dimethoate (a systemic insecticide, MOA 1B) on green beans, peas, tomatoes, capsicums, and potatoes. Note can have significant impact on beneficial insects and not fit well with IPM systems.
- Emamectin Benzoate (an insecticide with contact and translaminar activity, MOA 6) on brassicas, leafy vegetables, fruiting vegetables, cucurbits, and melons
- Indoxocarb (an insecticide with contact and some translaminar activity, MOA 22A) on tomatoes.

- Permethrin (an insecticide with contact activity, MOA 3A) on tomatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Pyrethrins (an insecticide with contact activity but very limited residual activity, MOA 3A) on brassicas, leafy vegetables, lettuce, fruiting vegetables, and legume vegetables.
- Spinetoram (an insecticide with contact, systemic and translaminar activity, MOA 5) on cotton.

Permits for these use patterns have not been applied to date. However, in the event of an incursion, emergency permits for these use patterns could be developed.

5.3.4 Physical control options

Destruction of host plants and deep ploughing of crop residues can assist with leafminer control as adult leafminers have difficulty emerging from pupae buried deeply in soil.

Removal of weed and volunteer hosts of the leafminer and planting susceptible crops away from weed hosts can also assist with the ongoing management of the pest by removing potential leafminer reservoirs.

5.3.5 Biological control

Although not specifically for eradication, biological controls will assist in lowering leafminer populations and will be an important part of the management of vegetable leafminer should eradication not be possible in the event of an outbreak.

Parasitoids often provide effective suppression of leafminers in the field when non-selective insecticides are not used (Ridland et al., 2020). History has also shown that invading *Liriomyza* populations are rapidly exploited by endemic Agromyzid parasitoids (Ridland et al., 2020). For these reasons it will be important that any pesticides used for the ongoing management of leafminers are compatible with Integrated Pest Management (IPM) systems.

Seventy-one parasitoid species have been recorded from vegetable leafminer (Ridland et al., 2020). Agromyzid parasitoids tend not to be very host-specific and Australia already has several endemic species that would likely be effective against the vegetable leafminer. *Diglyphus isaea* is one such parasitoid that occurs in Australia and is one of two parasitoids (the other being *Dacnusa sibirica*) that are mass reared overseas for the biological control of *Liriomyza* species (Ridland et al., 2020).

The importation of biological control agents is not warranted at this time as the adventive parasitoids such as *D. isaea*, already occur in Australia and would likely be suitable for the management of leafminers assuming pesticides are selected appropriately and are compatible with IPM systems.

5.4 Decision support for eradication

A summary of key activities associated with different scenarios associated with a detection of vegetable leafminer is provided in Table 9. Differing physical detection scenarios have been chosen to highlight operational differences in urban environments compared with agricultural and horticultural environments.

A summary of factors to be considered for eradication or alternative action is provided in Table 10.

Table 9. Recommended responses and considerations for differing scenarios of detection of vegetable leafminer

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Commercial glasshouse/ protected cropping setting	➢ Destruction of hosts within glasshouses could be considered to limit food sources or refuges (see Section 5.3.4). ➢ Chemical sprays to eradicate larvae and adults (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and in glasshouses managed by the same business as the infested glasshouse (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around the Infected Premises (IP). ➢ Focus on known hosts. ➢ Surveillance at linked properties. ➢ See Section 4 for points to consider when establishing a surveillance program. Key points for glasshouses are: • Undertake visual surveillance of all host plants. • Consider using visual surveys, sweep nets and yellow sticky traps.
Open agricultural/ horticultural setting	➢ Spray infested area using appropriate insecticide (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and survey jointly managed properties (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around each IP. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance awareness campaigns may be beneficial. ➢ Surveillance at linked properties should occur.

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Urban environment	➢ Chemical sprays to eradicate larvae and adults. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls in infested areas could be undertaken by permit or as voluntary movement controls (coupled with awareness campaign for urban areas).	➢ Surveillance on host plants within 1 km zone around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Surveillance to be undertaken in conjunction with awareness campaigns for households providing information on the pest and symptoms. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ Surveillance at linked properties.
Open natural environment (e.g. detection on roadsides or national park)	➢ Treatment of detections in a 1 km zone pending delimiting surveillance. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls on plant material from infested areas could be undertaken by permit or as voluntary movement controls. ➢ Movement restrictions may be considered pending outcomes of delimiting surveillance.	➢ Surveillance on host plants within 1 km zone around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance and awareness campaigns should be considered. ➢ Surveillance should be undertaken at linked properties.

Table 10. Summary of factors to be considered in determining whether eradication or alternative action will be taken for an incursion of a vegetable leafminer

FACTORS TO CONSIDER REGARDING THE TECHNICAL FEASIBILITY OF ERADICATION
• **The population size and population structure associated with the initial detection.** • **The cost effectiveness of recommended control technique options.** - Multiple applications of several different pesticides over a period of time (weeks/months) may be required to successfully eradicate vegetable leafminer populations. • **The ability to remove or destroy all vegetable leafminer by the recommended control techniques.** - Permission to enter private premises for surveillance and treatment must be considered. - Determination of whether treatments can effectively eradicate populations within premises or environments. • **The recommended control techniques are publicly acceptable.** - Chemical treatments in residences or backyards may not be acceptable or will require negotiation with residents. • **Whether emergency containment measures can effectively be put in place to contain the outbreak.** • **Whether there are control methods, commonly employed for endemic pests, that may prevent the establishment of, or be impacted by the treatment, for vegetable leafminer.** • **Legislative impediments to undertaking an eradication response.** - If not a prescribed pest, confirmatory diagnosis will be required under legislation in some jurisdictions before powers to enter premises and undertake treatments can be enacted. - Delimiting surveillance in urban environments may be restricted to front yards and environs rather than entry into premises (unless permission from the resident can be negotiated). - Emergency permits will be required for chemical treatments. Amendments to chemical labels will be required for longer term management of the pest if eradication is not possible. • **The ability to delimit the known area of infestation.** - Determination of linked properties in an urban detection may be unfeasible given the ability of adult leafminers to disperse at a local level. High risk linkages such as the recent purchase of seedlings, cut flowers or other commodities that could vector the pest should be investigated. • **The ability to identify and close the pathway for entry of the pest into Australia or from Cape York Peninsula into southern production areas of Australia.** - Pathways into Australia such as on nursery plants from overseas are managed via Post Entry Quarantine. - Natural dispersal is a possible pathway from neighbouring countries into northern Australia. Northern Australia Quarantine Strategy (NAQS) surveys offer an early detection mechanism to detect natural introductions from the north. If an outbreak is detected, the pathway will need to be identified and closed, where possible. - Pathways into southern production areas are regulated. For example, the Coen Information and Inspection Centre asks that fruit and plant material be surrendered to reduce the risk of pests moving south from Coen. - If an outbreak occurs south of Cape York Peninsula, it could be due to the existing population moving south or it could be due to a new incursion from overseas. In both cases the pathway will need to be identified and closed. • **The dispersal ability of the pest.** - Vegetable leafminer has the ability to naturally spread over short distances through flight or wind assisted dispersal. - Nursery material that is a host of vegetable leafminer (including containerised plants, seedlings, bare rooted plants, etc.; see Appendix 1 for host list) and cut flowers that are host plants are the most likely means of spread to new areas. They may carry all life stages of the

pest and will need to be managed to reduce the spread of the pest to new areas. Nursery material and cut flowers that are not a host of vegetable leafminer are at significantly lower risk (but may still transport adults across short periods if they have been in close proximity with host plants).

- Plant material (potentially including fresh hay and crop debris) and soil could also facilitate the spread of the pest (via adults and pupae) to new areas. Tools, equipment, machinery and vehicles are lower risk but may still transport hitchhiking adults or pupae via soil deposits.
- Vegetable leafminer is considered unlikely to travel with harvested fruit (without leaves) as the process of harvesting or moving produce is likely to cause adults to disperse and, provided soil and leaf material is absent, it is unlikely to spread larvae and pupae. Leafminer are also not expected to travel via the movement of seed.
- Fresh green or leafy vegetables or fresh legume vegetables (e.g. snow peas) may allow the spread of eggs and larvae and potentially adults and pupae. Therefore, movement restrictions or mandatory treatments may be needed for some vegetable crops.
- See Table 3 for further details on possible dispersal pathways.

- **The capability to detect vegetable leafminer at very low densities for the purpose of declaring freedom, and that all sites affected by the pest have or can be found.**
  - Experimental lures for vegetable leafminer exist (see Section 4.1.1.1). However, surveillance typically relies on the use of yellow sticky traps as well as sweep netting and visual inspection of host plants for mining activity.
- **The ability to put into place surveillance to confirm proof of freedom.**
  - Surveillance options are available for vegetable leafminer and include visual inspection of host plants for mines, yellow sticky traps and sweep netting.
- **Whether community consultation activities have or will be undertaken.**
  - In an urban environment, community consultation will be critical to assist in securing public support for delimiting surveillance and an eradication program.
  - Given the symptoms caused by this pest on a wide range of plants, general surveillance activities should be implemented comprising of awareness material, media releases, web-based reporting tools and the Exotic Plant Pest Hotline (1800 084 881).

6. EMERGENCY PLANT PEST RESPONSE DEED AND PLANTPLAN

The Emergency Plant Pest Response Deed (EPPRD) and PLANTPLAN should be referred to, in conjunction with this contingency plan when developing a Response Plan in the event of a vegetable leafminer detection outside its current range.

6.1 Emergency Plant Pest Response Deed

PHA is the custodian of the EPPRD. This is a formal legally binding agreement between PHA, the Australian Government, all state and territory governments and national plant industry body signatories.

It covers the management and funding of responses to Emergency Plant Pest (EPP) incidents, including the potential for Owner Reimbursement Costs for growers. It also formalises the role of plant industries' participation in decision making, as well as their contribution towards the costs related to approved responses.

Under the EPPRD an EPP is defined as a pest that meets one or more of the following criteria:

a) It is a known exotic Plant Pest the economic consequences of an occurrence of which would be economically or otherwise harmful for Australia, and for which it is considered to be in the regional and national interest to be free of the Plant Pest.
b) It is a variant form of an established Plant Pest which can be distinguished by appropriate investigative and diagnostic methods and which, if established in Australia, would have a regional and national impact.
c) It is a serious Plant Pest of unknown or uncertain origin which may, on the evidence available at the time, be an entirely new Plant Pest or one not listed in Schedule 13 and which if established in Australia is considered likely to have an adverse economic impact regionally and nationally.
d) It is a Plant Pest already found in Australia that:
    (i) is restricted to a defined area through the use of regulatory measures intended to prevent further spread of the pest out of the defined area or into an endangered area; and
    (ii) has been detected outside the defined area; and
    (iii) is not a native of Australia; and
    (iv) is not the subject of any instrument for management which is agreed to be effective risk mitigation and management at a national level; and
    (v) is considered likely to have an adverse economic impact such that an emergency response is required to prevent an incident of regional and national importance.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of the EPPRD.

6.2 PLANTPLAN

Underpinning the EPPRD is PLANTPLAN, the agreed technical response plan for an EPP incident. It provides nationally consistent guidelines for response procedures, outlining the phases of an incursion, as well as the key roles and responsibilities of industry and government during each of the phases.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of PLANTPLAN.

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8. APPENDICES

Appendix 1: Known hosts of vegetable leafminer

The vegetable leafminer (*Liriomyza sativae)* is highly polyphagous, with a wide host range across at least 15 families (Table 11). In order to facilitate trade and market access decisions, a thorough review was conducted as part of the Horticulture Innovation Australia funded Project MT16004 (R&E program for control, eradication and preparedness for vegetable leafminer), with a focus on evidence for lifecycle completion in the field. The results of this review, including commercial, ornamental and non-cultivated hosts of the vegetable leafminer (*L. sativae*), are presented in Table 12, Table 13, and Table 14 below. No guarantee can be made that these lists are exhaustive. Each record was scrutinized based on evidence provided showing completion of lifecycle within the field, and then included within the appropriate table. It is important to note that non-cultivated plants often serve as important reservoirs for *Liriomyza* pests (e.g. Schuster et al. 1991), and upon incursion into a new region, host ranges may expand to include native plant species. Moreover, host preferences may vary considerably across populations. For example, although chili (*Capsicum* spp.) is widely reported internationally as a favoured host of *Liriomyza sativae*, the population of *L. sativae* which has established within the Torres Strait (IPPC 2017) has very rarely been observed to attack chili (despite it being an abundant garden host in the region) (Elia Pirtle pers. comm).

Number of host records for the vegetable leafminer (*Liriomyza sativae*) within plant families.

Only records with either comprehensive or partial evidence of lifecycle completion in the field are included (see Table 12 and Table 13). Multiple records of the same species are included. Thus, the number of records does not indicate total number of affected species within each family. *Liriomyza sativae* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 11 Number of host records for the vegetable leafminer (Liriomyza sativae) within plant families.

Family	Number of records
Fabaceae	44
Asteraceae	26
Cucurbitaceae	22
Solanaceae	22
Brassicaceae	13
Lamiaceae	9
Malvaceae	9
Passifloraceae	7
Amaranthaceae	5
Apiaceae	4
Plantaginaceae	4
Amaryllidaceae	3
Verbenaceae	3

Family	Number of records
Araliaceae	2
Cleomaceae	2
Euphorbiaceae	2
Moringaceae	2
Ranunculaceae	2
Bignoniaceae	1
Caryophyllaceae	1
Convolvulaceae	1
Gentianaceae	1
Orchidaceae	1
Poaceae	1
Polemoniaceae	1
Polygonaceae	1
Rosaceae	1

Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*) has been provided within the scientific literature

This includes all records where the following conditions were met:

(1) Adults were reared from plant material infested with eggs or from larvae collected in the field; and
(2) Emerging adults were subsequently identified via morphology or molecular diagnostics; and
(3) Scientific names of hosts are reported

In addition, all records from Kenneth Spencer, the foremost authority on leafminer taxonomy, are included. Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included only if there are other records for species within this genus that met both conditions. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records confirming lifecycle completion for that same species that met both conditions. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report only partial confirmations of lifecycle completion are included in Table 13, while unverified records are included in Table 14. *Liriomyza sativae* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 12 Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the vegetable leafminer (Liriomyza sativae) has been provided within the scientific literature

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Spinach; Silverbeet	*Spinacia oleracea*		Amaranthaceae	Light mining (Oatman 1959)	Foba et al. (2015)	Oatman (1959); Stegmaier (1968); Andersen et al. (2008)
Commercial	Celery	*Apium graveolens*		Apiaceae	Leafminers damage leaves reducing marketability and quality (Palumbo 2016)	Zehnder and Trumble (1984)	Oatman (1959); Stegmaier (1968); Andersen et al. (2008)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Lettuce; Garden lettuce	*Lactuca sativa*		Asteraceae	Leaf mines make leaves unmarketable. Damage usually worse when planted near other hosts (Palumbo 2016)	Andersen et al. (2008); Reported affected in TS and CYP	Andersen et al. (2002); Parish et al. (2017)
Commercial	Leaf mustard; Mustard greens; Radish; Chinese mustard; Indian mustard; Mustard	*Brassica juncea*		Brassicaceae		Andersen et al. (2002); Andersen et al. (2008); Tran (2009)	Scheffer et al. (2006)A barcoding…)
Commercial	Gai lan; Cabbage; Broccoli; Caisin; Cabbage; Cauliflower; Field cabbage	*Brassica oleracea*		Brassicaceae	Broccoli and cauliflower can be attacked, impact most significant when plants are young (Palumbo 2016)	Andersen et al. (2008); Reported affected in TS and CYP	Stegmaier (1968); Parish et al. (2017)
Commercial	Kale	*Brassica oleracea*	*acephala*	Brassicaceae		Foba et al. (2015)	
Commercial	Cabbage	*Brassica oleracea*	*capitata*	Brassicaceae		Stegmaier (1966)	Scheffer et al. (2006)A barcoding…)
Commercial	Turnip; Rinsho; Chinese cabbage; Bokchoy	*Brassica rapa*		Brassicaceae	A common host in Vietnam (Andersen et al., 2008)	Stegmaier (1966); Reported affected in TS and CYP	Stegmaier (1968); Scheffer et al. (2006); Andersen et al. (2008)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Pak choi; Pechay; Chinese cabbage	*Brassica rapa*	*chinensis*	Brassicaceae		Andersen et al. (2008)	Andersen et al. (2002)
Commercial	Smooth leaf mustard; Mustard; Leaf mustard; Cabbage; Sweet crucifera; Chinese cabbage	*Brassica* sp./spp.		Brassicaceae		Stegmaier (1966); Spencer (1973); Andersen et al. (2008)	Oatman (1959); Stegmaier (1968); Andersen et al. (2008); Parish et al. (2017)
Commercial	Radish; Chinese radish (Daikon); Wild radish; Garden radish; White radish	*Raphanus sativus*		Brassicaceae	Light mining (Oatman 1959)	Andersen et al. (2008); Tran (2009)	Oatman (1959); Stegmaier (1968)
Commercial	Radish	*Raphanus* sp.		Brassicaceae		Reported affected in TS and CYP	Andersen et al. (2008)
Commercial	Wax gourd; White goard; Chinese melon; China squash; Ash gourd	*Benincasa hispida*		Cucurbitaceae		Rauf et al. (2000); Andersen et al. (2002); Andersen et al. (2008)	Andersen et al. (2008)
Commercial	Watermelon	*Citrullus lanatus*		Cucurbitaceae	Can cause seedling deaths	Rauf et al. (2000); Andersen et al.	

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
					(Palumbo 2016)	(2002); Andersen et al. (2008); Tran (2009); Foba et al. (2015); Reported affected in TS and CYP	
Commercial		*Citrullus lanatus*	*lanatus*	Cucurbitaceae			Scheffer et al. (2006)A barcoding…)
Commercial	Melon	*Cucumis* sp.		Cucurbitaceae		Spencer (1990)	Scheffer and Lewis (2005)
Commercial	Melon; Cantaloupe; Muskmelon; Honey melon; Cassaba melon	*Cucumis melo*		Cucurbitaceae	Damages leaves; can cause defoliation resulting in sunburnt fruit (Palumbo 2016); Heavy mining (Oatman 1959)	Stegmaier (1966); Spencer (1973); Scheffer and Lewis (2005); Andersen et al. (2008); Tran (2009)	Oatman (1959); Stegmaier (1968); Kox et al (2005); Guimarães et al. (2009)
Commercial	Cucumber	*Cucumis sativus*		Cucurbitaceae	Leaf mining and female puncturing reduces	Stegmaier (1966); Spencer (1973); Spencer (1983); Rauf et al. (2000);	Oatman (1959); Stegmaier (1968); Parish et al. (2017)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
					photosynthesis (Basji et al., 2011); Moderate mining (Oatman 1959)	Andersen et al. (2002); Scheffer and Lewis (2005); Andersen et al. (2008); Tran (2009); Nakamura et al. (2013); Reported affected in TS and CYP	
Commercial		*Cucumis sativus*	*sativus*	Cucurbitaceae			Scheffer et al. (2006)A barcoding…)
Commercial	Squash	*Cucurbita sp.*		Cucurbitaceae		Stegmaier (1966); Spencer (1990)	Stegmaier (1968); Scheffer et al. (2006)A barcoding…)
Commercial	Pumpkin; Winter squash; Squash	*Cucurbita maxima*		Cucurbitaceae	Light mining (Oatman 1959)	Andersen et al. (2002); Foba et al. (2015); Reported affected in TS and CYP	Oatman (1959); Stegmaier (1968); Andersen et al. (2008)
Commercial	Butternut squash; Pumpkin; Crookneck squash	*Cucurbita moschata*		Cucurbitaceae		Spencer (1973); Tran (2009)	
Commercial	Courgette; Zucchini; Pumpkin	*Cucurbita pepo*		Cucurbitaceae	The removal of leafminers using pesticides improved Californian	Stegmaier (1966); Spencer (1973); Scheffer and Lewis (2005); Foba et al. (2015); Reported	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
					squash yields by 32-62% (Sharma et al., 1980)	affected in TS and CYP	
Commercial		*Cucurbita pepo*	*pepo*	Cucurbitaceae			Scheffer et al. (2006)A barcoding…)
Commercial	Bottle gourd	*Lagenaria siceraria*		Cucurbitaceae		Andersen et al. (2008)	
Commercial	Angled luffa; Sing kwa; Towelsponge; Angled luffa	*Luffa acutangula*		Cucurbitaceae		Rauf et al. (2000); Andersen et al. (2008); Tran (2009)	Scheffer et al. (2006)A barcoding…)
Commercial	Sponge gourd; Ridge gourd	*Luffa aegyptiaca*		Cucurbitaceae		Andersen et al. (2008)	
Commercial	Bitter gourd; Balsam pear	*Momordica charantia*		Cucurbitaceae		Andersen et al. (2008); Tran (2009)	
Commercial		*Ricinus* sp.		Euphorbiaceae		Spencer (1973)	
Commercial		*Cajanus* sp.		Fabaceae		Spencer (1990)	
Commercial	Pidgeon pea	*Cajanus cajan*		Fabaceae		Stegmaier (1966); Spencer (1973)	Stegmaier (1968)
Commercial	Chickpea	*Cicer arietinum*		Fabaceae		Avalos et al. (2013); Medina et al. (2014)	
Commercial	Soybean; Soya bean	*Glycine max*		Fabaceae		Rauf et al. (2000)	Valladares (1984); Parish et al. (2017)
Commercial	Lentil; Lentil bean	*Lens culinaris*		Fabaceae		Scheffer and Lewis (2005)	
Commercial	Lupine	*Lupinus* sp./ spp.		Fabaceae	Heavy mining (Oatman 1959)	Stegmaier (1966); Spencer (1973)	Oatman (1959); Stegmaier (1968);

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
							Scheffer and Lewis (2005)
Commercial		*Medicago* sp.		Fabaceae		Spencer (1990)	
Commercial	Alfalfa; Lucerne	*Medicago sativa*		Fabaceae	Heavy mining (Oatman 1959)	Spencer (1973); Spencer (1990)	Oatman (1959); Stegmaier (1968); Valladares (1984); Salvo and Valladares (1997)
Commercial	Sieva; Lima bean; Lima bean	*Phaseolus lunatus*		Fabaceae		Stegmaier (1966); Spencer (1973)	Stegmaier (1968)
Commercial	Beans; Grean beans; Pole beans	*Phaseolus* sp.		Fabaceae		Stegmaier (1966)	Stegmaier (1968); Scheffer et al. (2006)A barcoding…)
Commercial	Green bean; French bean; Kidney bean; Bean; Common bean; Snap bean	*Phaseolus vulgaris*		Fabaceae	Leaves tend to be the only part affected. Leaf mines reduce photosynthetic capacity of plant (Johnson 1993)	Spencer (1983); Rauf et al. (2000); Andersen et al. (2002); Scheffer and Lewis (2005); Andersen et al. (2008); Tran (2009); Foba et al. (2015)	Stegmaier (1968); Kox et al (2005); Musundire et al. (2012); Parish et al. (2017)
Commercial	Snow pea	*Pisum* sp.		Fabaceae		Spencer (1990)	
Commercial	Garden pea; Pea; Snow pea; Sugar snap	*Pisum sativum*		Fabaceae		Andersen et al. (2008); Foba et al. (2015)	Oatman (1959); Stegmaier (1968)
Commercial		*Trifolium* sp.		Fabaceae		Spencer (1990)	
Commercial	Crimson clover	*Trifolium*		Fabaceae		Stegmaier (1966)	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
		incarnatum					
Commercial		*Vicia* sp.		Fabaceae		Spencer (1973)	
Commercial	Broad bean; Faba bean; Fava bean	*Vicia faba*		Fabaceae		Rauf et al. (2000); Scheffer and Lewis (2005); Andersen et al. (2008)	Musundire et al. (2012)
Commercial	Mungbean; Green gram	*Vigna radiata*		Fabaceae		Andersen et al. (2002); Tran (2009)	Andersen et al. (2008)
Commercial	Red bean; Black eyed pea; Cowpea	*Vigna sinensis*		Fabaceae		Stegmaier (1966); Spencer (1973); Rauf et al. (2000)	Stegmaier (1968)
Commercial	Walp Cowpea; Yard-long bean; Long bean; Snakebean; Cowpea	*Vigna unguiculata*		Fabaceae		Rauf et al. (2000); Andersen et al. (2002); Scheffer and Lewis (2005); Andersen et al. (2008); Tran (2009); Foba et al. (2015)	Scheffer et al. (2006)A barcoding…)
Commercial	Snake bean	*Vigna unguiculata*	*sesquipedalis*	Fabaceae		Pirtle et al. (in review); Reported affected in TS and CYP**	
Commercial	Basil; Sweet basil; Thai basil	*Ocimum basilicum*		Lamiaceae	Infested basil is frequently reported in Europe from imported material (Bragard	Pirtle et al. (in review); Reported affected in TS and CYP**	Andersen et al. (2008); Andersen and Hofsvang (2010)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
					et al 2020)		
Commercial		*Abelmoschus* sp.		Malvaceae		Spencer (1990)	
Commercial	Okra	*Abelmoschus esculentus*		Malvaceae		Andersen et al. (2008); Foba et al. (2015)	Scheffer et al. (2006)A barcoding…)
Commercial	Cotton	*Gossypium* sp.		Malvaceae		Spencer (1973)	Stegmaier (1968)
Commercial	Passionfruit	*Passiflora edulis*		Passifloraceae		Pirtle et al. (in review); Reported affected in TS and CYP**	
Commercial		*Passiflora* sp.		Passifloraceae		Stegmaier (1966)	
Commercial	Plantain	*Plantago* sp.		Plantaginaceae		Stegmaier (1966)	Stegmaier (1968)
Commercial	Sweet pepper	*Capsicum*		Solanaceae		Foba et al. (2015)	
Commercial	Chili	*Capsicum annum*		Solanaceae		Spencer (1983)	
Commercial	Pepper; Sweet pepper; Green pepper; Chili; Capsicum	*Capsicum annuum*		Solanaceae		Spencer (1973)	
Commercial	Sweet pepper; Pepper; Chilli	*Capsicum sp.*		Solanaceae		Pirtle et al. (in review)	Stegmaier (1968)
Commercial	Tomato	*Lycopersicum esculentum*		Solanaceae	Up to 70% losses reported in Vanuatu (Murphy and LeSalle 1999). Hawaiian tomato yield losses were	Spencer (1973); Spencer (1983); Zehnder and Trumble (1984); Rauf et al. (2000); Scheffer and Lewis (2005); Andersen et al.	Oatman (1959); Stegmaier (1968); Kox et al (2005); Scheffer and Lewis (2005); Scheffer et al. (2006); Musundire et al. (2012); Parish et al. (2017)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
					reported to be less than 10% until damage reached >8 mines/leaflet (Johnson 1993)	(2008); Tran (2009); Nakamura et al. (2013); Foba et al. (2015); Reported affected in TS and CYP**	
Commercial		*Solanum lycopersicum* (Syn. *Lycopersicum esculentum)*		Solanaceae		Stegmaier (1966)	
Commercial	Brinjal eggplant; Eggplant; common eggplant; Pickling eggplant	*Solanum melongena*		Solanaceae		Stegmaier (1966); Spencer (1973); Andersen et al. (2008); Tran (2009); Foba et al. (2015); Reported affected in TS and CYP	Oatman (1959); Stegmaier (1968); Scheffer et al. (2006); Andersen and Hofsvang (2010)
Commercial	Potato	*Solanum tuberosum*		Solanaceae		Spencer (1973); Foba et al. (2015)	Oatman (1959); Stegmaier (1968); Scheffer et al. (2006); Andersen et al. (2008)
Ornamental	Penniwort	*Hydrocotyle* sp.		Araliaceae		Spencer (1973)	
Ornamental		*Hydrocotyle umbellata*		Araliaceae		Stegmaier (1966)	

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Ornamental	Aster	*Aster sp.*		Asteraceae	Heavy mining (Oatman 1959)	Spencer (1973); Spencer (1990)	Oatman (1959); Stegmaier (1968)
Ornamental		*Calendula* sp.		Asteraceae		Spencer (1973)	
Ornamental	Pot marigold	*Calendula officinalis*		Asteraceae		Stegmaier (1966)	Stegmaier (1968)
Ornamental	Garland chrysanthemum	*Chrysanthemum coronarium*		Asteraceae		Tran (2009)	
Ornamental	Garland chrysanthemum; Chrysanthemum	*Chrysanthemum* sp.		Asteraceae		Scheffer and Lewis (2005); Andersen et al. (2008)	
Ornamental	Dahlia	*Dahlia* sp.*		Asteraceae		Spencer (1973)	Sooda et al. (2017)
Ornamental	Marigold	*Tagetes* sp.		Asteraceae	Heavy mining (Oatman 1959)	Reported affected in TS and CYP	Oatman (1959); Stegmaier (1968); Andersen et al. (2008)
Ornamental		*Zinnia* sp.*		Asteraceae	Heavy mining (Oatman 1959)	Spencer (1973)	Oatman (1959); Stegmaier (1968)
Ornamental	Yellow bells	*Tecoma stans*		Bignoniaceae		Reported affected in TS and CYP	
Ornamental		*Bauhinia sp.*		Fabaceae		Stegmaier (1966); Spencer (1973)	Stegmaier (1968)
Ornamental		*Indigofera* sp.		Fabaceae		Spencer (1973); Spencer (1990)	
Ornamental	Indigo	*Indigofera tinctoria*		Fabaceae		Reported affected in TS and CYP	
Ornamental		*Moringa* sp.		Moringaceae		Spencer (1973); Spencer (1990)	

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Ornamental	Passion flower	*Passiflora pallens*		Passifloraceae		Stegmaier (1966)	Stegmaier (1968)
Ornamental		*Phlox* sp.		Polemoniaceae		Spencer (1990)	
Ornamental		*Cestrum* sp.		Solanaceae		Spencer (1973); Spencer (1990)	
Ornamental		*Cestrum diurnum*		Solanaceae		Stegmaier (1966)	
Ornamental	Night blooming cestrum	*Cestrum nocturnum*		Solanaceae		Stegmaier (1966)	Stegmaier (1968)
Ornamental	Petunia	*Petunia* sp.		Solanaceae		Nakamura et al. (2013); Reported affected in TS and CYP	
Ornamental	Ornamental	unreported		Verbenaceae			Parish et al. (2017)
Non cultivated	Shepherd's needles; Spanish needles	*Bidens alba*		Asteraceae		Schuster et al. (1991)	
Non cultivated		*Galinsoga* sp.		Asteraceae		Spencer (1973); Spencer (1990)	
Non cultivated	Quickweed; Shaggy soldier	*Galinsoga ciliata*		Asteraceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated		*Lipochaeta* sp.		Asteraceae		Spencer (1973); Spencer (1990)	
Non cultivated		*Verbesina* sp.		Asteraceae		Spencer (1973)	
Non cultivated	Crownbeard	*Verbesina helianthoides*		Asteraceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated		*Verbesina* sp.		Asteraceae		Stegmaier (1966)	
Non cultivated	White	*Verbesina virginica*		Asteraceae		Stegmaier (1966)	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
	Crownbeard						
Non cultivated	Prickly spider flower	*Cleome aculeata*		Cleomaceae		Reported affected in TS and CYP	
Non cultivated	Tickweed	*Cleome viscosa*		Cleomaceae		Reported affected in TS and CYP	
Non cultivated		*Ceratosanthes* sp.		Cucurbitaceae		Spencer (1990)	
Non cultivated	Castor bean; Castor oil bush	*Ricinus communis*		Euphorbiaceae		Stegmaier (1966); Spencer (1983); Spencer (1990); Rauf et al. (2000); Reported affected in TS and CYP	Oatman (1959); Stegmaier (1968)
Non cultivated		*Cassia* sp. (now *Senna* sp.)		Fabaceae		Spencer (1973)	Andersen and Hofsvang (2010)
Non cultivated		*Cassia occidentalis* (now *Senna occidentalis*)		Fabaceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated		*Cassia tora* (now *Senna tora*)		Fabaceae		Stegmaier (1966)	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
Non cultivated	Streaked rattlepod	*Crotalaria pallida*		Fabaceae		Reported affected in TS and CYP**	
Non cultivated	Siratro	*Macroptilium atropurpureum*		Fabaceae		Reported affected in TS and CYP**	
Non cultivated		*Melilotus* sp.		Fabaceae		Spencer (1990)	
Non cultivated	White melilot	*Melilotus alba*		Fabaceae		Stegmaier (1966)	Oatman (1959); Stegmaier (1968)
Non cultivated		*Poissonia* sp.		Fabaceae		Spencer (1990)	
Non cultivated		*Poissonia hypoleuca*		Fabaceae		Spencer (1990)	
Non cultivated	Corkwood tree	*Sesbania grandiflora*		Fabaceae		Reported affected in TS and CYP	
Non cultivated	Cowpea	*Vigna repens*		Fabaceae		Stegmaier (1966); Spencer (1973)	Stegmaier (1968)
Non cultivated		*Annoda cristata*		Malvaceae		Stegmaier (1966)	
Non cultivated		*Anoda* sp.		Malvaceae		Spencer (1973)	
Non cultivated		*Sida* sp.		Malvaceae		Spencer (1973); Spencer (1990)	
Non cultivated	Sida	*Sida acuta*		Malvaceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated		*Passiflora* sp.		Passifloraceae		Spencer (1973)	
Non cultivated		*Passiflora foetida*		Passifloraceae		Pirtle et al. (in review); Reported affected in TS and CYP**	
Non cultivated	Corky stemmed passion flower	*Passiflora pallida*		Passifloraceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated		*Turnera ulmifolia*		Passifloraceae		Pirtle et al. (in review);	

Host type	Common names	Scientific name	Variety	Family	Notes	Records with comprehensive evidence	All other records (partial and unverified records)
						Reported affected in TS and CYP**	
Non cultivated	Plantain	*Plantago* sp.		Plantaginaceae		Spencer (1973)	
Non cultivated	Tolgaucha	*Datura* sp.		Solanaceae		Spencer (1973); Spencer (1990)	
Non cultivated	Cutleaf ground cherry; Mullaca; Wild gooseberry	*Physalis angulata*		Solanaceae		Pirtle et al. (in review); Reported affected in TS and CYP	Andersen et al. (2008)
Non cultivated	American black nightshade	*Solanum americanum*		Solanaceae		Schuster et al. (1991)	
Non cultivated	Black nightshade	*Solanum nigrum*		Solanaceae		Stegmaier (1966)	Stegmaier (1968)
Non cultivated	Turkey berry	*Solanum torvum*		Solanaceae		Tran (2009)	
Non cultivated	snakeweed	*Stachytarpheta jamaicensis*		Verbenaceae		Pirtle et al. (in review); Reported affected in TS and CYP**	

Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*) has been provided within the scientific literature

This includes all records where:

(1) Adults were reared from field collected plant material infested with larvae, but no subsequent identification via morphology or molecular diagnostics was reported; or
(2) Laboratory feeding and oviposition preference experiments were conducted that did not confirm lifecycle completion; or
(3) Identification via morphology or molecular diagnostics was reported, but collection methods in the field were not reported. Lifestyle completion cannot be confirmed in these instances due to the potential use of non-specific collection methods (e.g. sticky traps or sweep nets); or
(4) Laboratory cultures confirmed lifecycle completion, but evidence of lifecycle completion in the field was not provided; or
(5) Only common names of plant were given, but the scientific name could be confidently inferred; or
(6) The record reports presence of *L. sativae* on imported material detected at the border.

Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included here only if there are other records for species within this genus that met one of these conditions outlined above. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records for that same species that meet one of the conditions outlined above. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 12, while unverified records are included in Table 14. *Liriomyza sativae* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 13 Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the vegetable leafminer (Liriomyza sativae) has been provided within the scientific literature.

Host type	Common names	Scientific name	Variety	Family	Notes	All records (including any unverified records)
Commercial	Beet; Chard; Beetroot	*Beta vulgaris*		Amaranthaceae		Andersen et al. (2008)
Commercial	Leek; Leeks	*Allium ampeloprasum*		Amaryllidaceae		Andersen et al. (2008)
Commercial	Onion; Shallot;	*Allium cepa*		Amaryllidaceae	Mines can girdle	Herr and Johnson (1992);

Host type	Common names	Scientific name	Variety	Family	Notes	All records (including any unverified records)
	Multiplier onion				stems causing leaf death (Carolina et al., 1992)	Andersen et al. (2008)
Commercial	Garlic	*Allium sativum*		Amaryllidaceae		Andersen et al. (2008)
Commercial	Coriander	*Coriandrum sativum*		Apiaceae		Andersen and Hofsvang (2010)
Commercial	Carrot	*Daucus carota*		Apiaceae		Andersen et al. (2008)
Commercial		*Daucus carota*	*sativus*	Apiaceae		Scheffer et al. (2006)A barcoding…)
Commercial	Chinese cabbage; Petsai; Common yellow mustard	*Brassica campestris*		Brassicaceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Commercial	Nasturtium	*Nasturtium* sp.		Brassicaceae		Oatman (1959); Stegmaier (1968)
Commercial	Watermelon	*Citrullus vulgaris*		Cucurbitaceae	Heavy mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Commercial	Lima bean	*Phaseolus limensis*		Fabaceae		Oatman (1959); Stegmaier (1968); Herr and Johnson (1992); Kox et al (2005)
Commercial	Mung bean	*Phaseolus radiatus*		Fabaceae		Kox et al (2005); Kox et al. (2005)
Commercial	Strawberry clover	*Trifolium frageriferum*		Fabaceae		Oatman (1959); Stegmaier (1968)
Commercial	Alsike clover; White clover	*Trifolium hybridum*		Fabaceae		Oatman (1959); Stegmaier (1968)
Commercial	Red clover	*Trifolium pratense*		Fabaceae		Oatman (1959);

Host type	Common names	Scientific name	Variety	Family	Notes	All records (including any unverified records)
						Stegmaier (1968)
Commercial	White clover	*Trifolium repens*		Fabaceae		Oatman (1959); Stegmaier (1968)
Commercial		*Mentha* sp.		Lamiaceae		Andersen and Hofsvang (2010)
Commercial		*Ocimum americanum*		Lamiaceae		Andersen and Hofsvang (2010)
Commercial		*Ocimum canum*		Lamiaceae		Andersen and Hofsvang (2010)
Commercial		*Ocimum sanctum*		Lamiaceae		Andersen and Hofsvang (2010)
Commercial	Oregano	*Origanum vulgare*		Lamiaceae		Andersen et al. (2008)
Commercial	Upland cotton; Cotton	*Gossypium hirsutum*		Malvaceae	Moderate mining (Oatman 1959)	Oatman (1959); Stegmaier (1968); Andersen et al. (2002); Andersen et al. (2008)
Commercial	Barley	*Hordeum vulgare*		Poaceae	Light mining (Oatman 1959)	Oatman (1959)
Commercial	Bell pepper	*Capsicum frutescens*		Solanaceae		Oatman (1959); Stegmaier (1968)
Ornamental	Amaranth; Bayam	*Amaranthus* sp.		Amaranthaceae		Andersen and Hofsvang (2010)
Ornamental		*Amaranthus tricolour*		Amaranthaceae		Andersen and Hofsvang (2010)
Ornamental	Oxeye daisy	*Chrysanthemum leucanthemum*		Asteraceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Ornamental	Cineraria	*Felicia bergeriana*		Asteraceae	Moderate mining	Oatman (1959);

Host type	Common names	Scientific name	Variety	Family	Notes	All records (including any unverified records)
					(Oatman 1959)	Stegmaier (1968)
Ornamental	Sunflower; Common sunflower	*Helianthus annuus**		Asteraceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Ornamental	Gypsophila	*Gypsophila* sp.		Caryophyllaceae		Andersen and Hofsvang (2010)
Ornamental		*Lisianthus* sp.		Gentianaceae		Andersen and Hofsvang (2010)
Ornamental		*Dendrobium* sp.		Orchidaceae		Andersen and Hofsvang (2010)
Ornamental	Snapdragon	*Antirrhinum majus*		Plantaginaceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Ornamental	Columbine	*Aquilegia* sp.		Ranunculaceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Ornamental	Buttercup	*Ranunculus* sp.		Ranunculaceae	Light mining (Oatman 1959)	Oatman (1959); Stegmaier (1968)
Ornamental	Desert peach	*Prunus andersonii*		Rosaceae	Light mining (Oatman 1959)	Oatman (1959)
Non cultivated	Rough pigweed	*Amaranthus retroflexus*		Amaranthaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Goosefoot; Lamb's quarters; Fat hen	*Chenopodium album*		Amaranthaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Common groundsel	*Senecio vulgaris*		Asteraceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Milk thistle; Blessed milk	*Silybum marianum*		Asteraceae		Oatman (1959); Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Notes	All records (including any unverified records)
	thistle					
Non cultivated	Common sow thistle	*Sonchus oleraceus*		Asteraceae		Oatman (1959)
Non cultivated	Yellow melilot	*Melilotus indica*		Fabaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Garden balm	*Melissa officinalis*		Lamiaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Dwarf mallow	*Malva rotundifolia*		Malvaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Broad leaved plantain	*Plantago major*		Plantaginaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Knotweed	*Polygonum aviculare*		Polygonaceae		Oatman (1959); Stegmaier (1968)
Non cultivated	Tolgaucha	*Datura meteloides*		Solanaceae		Oatman (1959)
Non cultivated	Lance leafed ground cherry	*Physalis lanceifolia*		Solanaceae		Oatman (1959); Stegmaier (1968)

Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the vegetable leafminer (*Liriomyza sativae*).

This includes all records for which no evidence was presented within the record for lifecycle completion and no evidence was shown for confident species identification, or evidence could not suitably be accessed for verification. We are not disputing the veracity of these records - we simply did not have enough information available to verify if these hosts support lifecycle completion in the field. This includes all records where:

(1) Adults were collected by sweep netting the presumed host, but neither larval activity nor lifecycle completion was confirmed; or
(2) A scientific name could not reliably be assumed from a reported common name; or
(3) Collection and identification methodology were unclear or unreported; or
(4) The record could not be accessed or suitably translated to confirm collection and identification methodology.

For some unverified species within this table, there may be other congeneric species that have been either comprehensively or partially confirmed as a host plant, and thus these would appear within Table 12 or Table 13. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 12, while records that report only partial confirmations of lifecycle completion are included in Table 13. *Liriomyza sativae* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 14 Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the vegetable leafminer (Liriomyza sativae).

Host type	Common names	Scientific name	Variety	Family	Records
commercial	Okra	*Hibiscus esculentus*		Malvaceae	Stegmaier (1968)
ornamental	Pennywort	*Hydrocotyle unbellata*		Araliaceae	Stegmaier (1968)
ornamental	Dahlia	*Dahlia pinnata*		Asteraceae	Stegmaier (1968)
ornamental	Creek goldenrod	*Solidago elongata*		Asteraceae	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Records
ornamental	Giant goldenrod	*Solidago gigantea*		Asteraceae	Stegmaier (1968)
ornamental	Goldenrod	*Solidago sp.*		Asteraceae	Stegmaier (1968)
non cultivated	Black medic	*Medicago lupilina*		Fabaceae	Frick (1959); Stegmaier (1968)
non cultivated	Giant vetch	*Vicia gigantea*		Fabaceae	Frick (1959); Stegmaier (1968)
non cultivated	Anoda	*Anoda cristata*		Malvaceae	Stegmaier (1968)
non cultivated	Mallow	*Malva borealis*		Malvaceae	Stegmaier (1968)
non cultivated	Mallow	*Malva nicaeensis*		Malvaceae	Stegmaier (1968)
non cultivated	Tolgaucha	*Datura sp.*		Solanaceae	Stegmaier (1968)

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Appendix 2: Leaf mining damage caused by established species

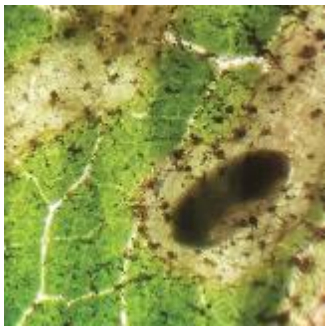
Table 15 provides a summary of established (native or naturalised) leaf mining species, their host overlap with vegetable leafminer and features that can be used to differentiate between vegetable leafminer and established leafminers.

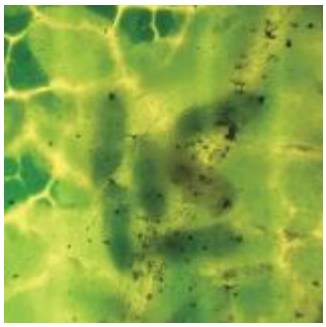
It should be noted that because host plant records are poor for many native or naturalised species, there could be many more affected hosts than included here. The host overlap between vegetable leafminer and several native leafminers underscores the importance of sample collection and molecular diagnostics.

Figure 10 provides some general guidelines for differentiating between dipteran and lepidopteran leafminer larvae.

Table 15. Leaf mining damage caused by Australian dipterans or lepidopterans on leaves of cultivated crops and common weeds of cultivated crops

LEAFMINER	DISTINGUISHING FEATURES FROM VEGETABLE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH VEGETABLE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Liriomyza brassicae	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from vegetable leafminer).	Brassicaceae Capparaceae Nasturtium (Tropaeolaceae) Peas (Fabaceae)	
Liriomyza chenopodii	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from vegetable leafminer).	Chenopodiaceae (incl. beet) Caryophyllaceae (incl. chickweed) Any native hosts are as yet unrecorded (*L. chenopodii* is a native species)	

LEAFMINER	DISTINGUISHING FEATURES FROM VEGETABLE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH VEGETABLE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Phytomyza syngenesiae	Pupation occurs inside the leaf mine.  Adults easily distinguishable from vegetable leafminer.	Asteraceae (esp. lettuces, daisies, sow thistle, Cineraria) Wild carrot (Umbelliferae) Peas (Fabaceae)	
Tropicomyia polyphtya	Larvae are upper surface epidermal feeders (deeper parenchymatous tissues is not eaten). As such, mines have a silvery, film like appearance. Pupation occurs inside the mine and pupa is noticeably flattened.  Adults outwardly distinguishable from vegetable leafminer.	Highly polyphagous Common hosts include plants in the Passifloraceae, Euphorbiaceae, Fabaceae, Rubiaceae, Ruaceae, and Solanaceae families, and more.	
Ophiomyia solanicola	Pupation occurs inside the leaf mine.  Adults easily distinguishable from vegetable leafminer.	Solanaceae (incl. egg plant)	

LEAFMINER	DISTINGUISHING FEATURES FROM VEGETABLE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH VEGETABLE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
Ophiomyia alysicarpi	Mint green pupae can be found inside mines.  Adults easily distinguishable from vegetable leafminer.	*Alysicarpus* sp. *Desmodium* sp.	
Ophiomyia cornuta	Adults easily distinguishable from vegetable leafminer.	*Scaevola* sp. *Goodenia* sp.	
Lepidopteran leafminer	Lepidopteran larvae can be distinguished from dipteran larvae on the spot, via a hand lens (Figure 10). For some lepidopteran species, when the larva is in its final stages, it forms a large blister at the end of the mine (see images on the right). vegetable leafminer larvae do not create such blisters.	Polyphagous (incl. <u>Fabaceae</u>, Eucalypts, native trees, etc) At least one lepidopteran leafminer is found in <u>*Macroptilium atropurpureum*</u> (a highly preferred host of vegetable leafminer)	 

LEAFMINER	DISTINGUISHING FEATURES FROM VEGETABLE LEAFMINER DAMAGE	HOSTS (THOSE SHARED WITH VEGETABLE LEAFMINER ARE UNDERLINED)	PHOTO OF LEAF MINE
			

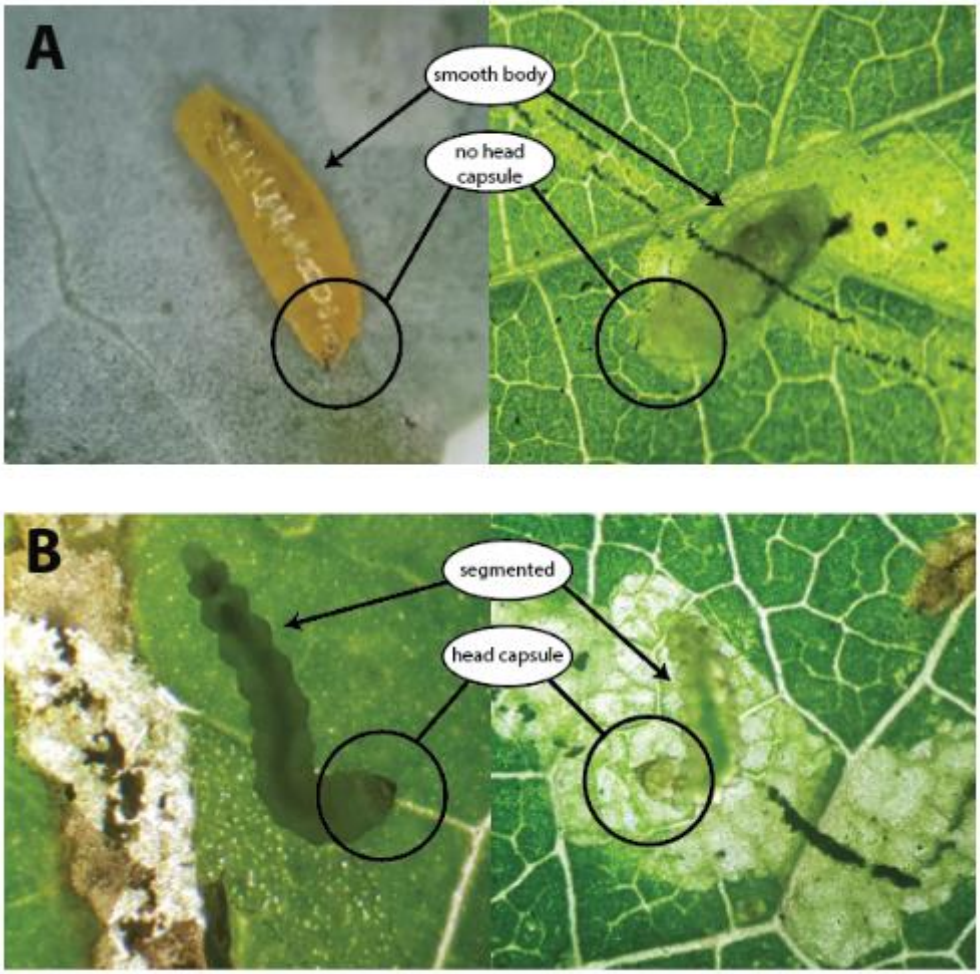


Figure 10. General guidelines for distinguishing dipteran vs lepidopteran leafminer larvae. Dipteran leafminer larvae (A) lack a head capsule and have a smooth body without clear segments. Lepidopteran leafminer larvae (B) have a distinct head capsule and are more visibly segmented. When attempting to distinguish a dipteran from lepidopteran leafminer larva on the spot, it is helpful to backlight the leaf as it is viewed under a hand lens, or to carefully excise the larva. Images: Elia Pirtle, Cesar Australia *Pty Ltd.*

Surveillance for *L. sativae* in Australia will be confounded by the presence of other leafmining insects already present in Australia, many of which create mines that are indistinguishable to a casual observer, and even in some cases indistinguishable to experts.

Table 16 lists genera that include hosts of *L. sativae* but do not appear to contain other agromyzids within Australia or Australasia. It is important to note that host plant records for non-pest leafminer flies are highly incomplete, and some of the genera included herein could host leafminer flies that have not yet been reported. Host records for weeds and native plants are less complete than for cultivated crops and ornamentals. Despite poor host records for native leafminers, we can be more confident that native leafminers are not currently a notable presence within the genera containing cultivated crops and ornamentals, where sampling effort is expected to be higher. As such, leafmining damage detected within the genera included in these tables should be regarded as highly suspicious.

Error! Reference source not found. indicates where known overlap exists between hosts of *L. huidobrensis, L. trifolii* or *L. sativae* and native or naturalised agromyzids in Australia and Australasia.

*Table 16 Host genera of L. huidobrensis, L. sativae or L trifolii (including only records with comprehensive or partial evidence for lifecycle completion in the field, see Appendix 1) without any reported overlap with native or naturalised agromyzids already present in Australasia (see **Error! Reference source not found.** for known overlap). For each host, preferences of the three exotic Liriomyza are indicated (LH = L. huidobrensis; LT = L. trifolii; LS = L. sativae).*

Genus	Family	Exotic *Liriomyza*	Host type
Allium	Amaryllidaceae	LH; LT; LS	commercial
Coriandrum	Apiaceae	LH; LT; LS	commercial
Eryngium	Apiaceae	LH	commercial
Petroselinum	Apiaceae	LT	commercial
Colocasia	Araceae	LH	commercial
Cichorium	Asteraceae	LH	commercial
Crassocephalum	Asteraceae	LH	commercial
Basella	Basellaceae	LH	commercial
Eruca	Brassicaceae	LH	commercial
Nasturtium	Brassicaceae	LH; LS	commercial
Spinacea	Chenopodiaceae	LH	commercial
Benincasa	Cucurbitaceae	LH; LT; LS	commercial
Coccinia	Cucurbitaceae	LT	commercial
Lagenaria	Cucurbitaceae	LH; LT; LS	commercial
Luffa	Cucurbitaceae	LH; LT; LS	commercial
Momordica	Cucurbitaceae	LH; LT; LS	commercial
Sechium	Cucurbitaceae	LH	commercial
Trichosanthes	Cucurbitaceae	LT	commercial
Arachis	Fabaceae	LT	commercial
Cicer	Fabaceae	LH; LT; LS	commercial
Cyamopsis	Fabaceae	LT	commercial
Lens	Fabaceae	LS	commercial
Lotus	Fabaceae	LH	commercial
Lupin	Fabaceae	LH	commercial
Medicago	Fabaceae	LH; LT; LS	commercial
Trigonella	Fabaceae	LH; LT	commercial
Vicia	Fabaceae	LH; LT; LS	commercial
Mentha	Lamiaceae	LH; LS	commercial

Genus	Family	Exotic *Liriomyza*	Host type
Origanum	Lamiaceae	LS	commercial
Linum	Linaceae	LH	commercial
Abelmoschus	Malvaceae	LH; LT; LS	commercial
Corchorus	Malvaceae	LT	commercial
Gossypium	Malvaceae	LT; LS	commercial
Hibiscus	Malvaceae	LH; LT	commercial
Zizania	Poaceae	LH	commercial
Lycium	Solanaceae	LH	commercial
Thurnbergia	Acanthaceae	LH	ornamental
Celosia	Amaranthaceae	LH; LT	ornamental
Bupleurum	Apiaceae	LH	ornamental
Centella	Apiaceae	LH	ornamental
Oenanthe	Apiaceae	LH	ornamental
Catharanthus	Apocynaceae	LH	ornamental
Asclepias	Asclepiadaceae	LT	ornamental
Chionodoxa	Asparagaceae	LH	ornamental
Hemerocallis	Asphodelaceae	LH	ornamental
Argyranthemum	Asteraceae	LH	ornamental
Calendula	Asteraceae	LH; LT; LS	ornamental
Centaurea	Asteraceae	LH	ornamental
Conoclinium	Asteraceae	LH	ornamental
Cosmos	Asteraceae	LH	ornamental
Dimorphotheca	Asteraceae	LT	ornamental
Echinacea	Asteraceae	LT	ornamental
Eupatorium	Asteraceae	LT	ornamental
Felicia	Asteraceae	LS	ornamental
Flaveria	Asteraceae	LT	ornamental
Kalimeris	Asteraceae	LH	ornamental
Osteospermum	Asteraceae	LH	ornamental
Pyrethrum	Asteraceae	LH	ornamental
Tanacetum	Asteraceae	LH	ornamental
Zinnia	Asteraceae	LH; LT; LS	ornamental
Impatiens	Balsaminaceae	LH	ornamental
Calceolaria	Calceolariaceae	LH	ornamental
Campanula	Campanulaceae	LH	ornamental
Platycodon	Campanulaceae	LH	ornamental
Trachelium	Campanulaceae	LH; LT	ornamental
Dianthus	Caryophyllaceae	LH; LT	ornamental
Gypsophila	Caryophyllaceae	LH; LT; LS	ornamental
Bauhinia	Fabaceae	LS	ornamental
Lathyrus	Fabaceae	LH	ornamental
Lupinus	Fabaceae	LH; LS	ornamental
Eustoma	Gentianaceae	LH; LT	ornamental
Exacum	Gentianaceae	LH	ornamental
Lisianthus	Gentianaceae	LH; LT; LS	ornamental
Freesia	Iridaceae	LH	ornamental
Gladiolus	Iridaceae	LH	ornamental
Salvia	Lamiaceae	LH; LT	ornamental

Genus	Family	Exotic *Liriomyza*	Host type
Lilium	Liliaceae	LH	ornamental
Torenia	Linderniaceae	LH	ornamental
Althaea	Malvaceae	LH	ornamental
Moringa	Moringaceae	LS	ornamental
Godetia	Onagraceae	LH	ornamental
Oenothera	Onagroideae	LH	ornamental
Dendrobium	Orchidaceae	LS	ornamental
Papaver	Papaveraceae	LH	ornamental
Antirrhinum	Plantaginaceae	LH; LT; LS	ornamental
Myosotis	Plumbaginaceae	LH	ornamental
Lagurus	Poaceae	LH	ornamental
Phlox	Polemoniaceae	LH; LT; LS	ornamental
Primula	Primulaceae	LH	ornamental
Aquilegia	Ranunculaceae	LS	ornamental
Delphinium	Ranunculaceae	LH	ornamental
Nigella	Ranunculaceae	LH	ornamental
Prunus	Rosaceae	LS	ornamental
Rosa	Rosaceae	LH; LT	ornamental
Diascia	Scrophulariaceae	LH	ornamental
Viola	Violaceae	LH	ornamental
Sagittaria	Alismataceae	LH	non cultivated
Alternanthera	Amaranthaceae	LT	non cultivated
Amaranthus	Amaranthaceae	LH; LT; LS	non cultivated
Deeringia	Amaranthaceae	LH	non cultivated
Gomphrena	Amaranthaceae	LH	non cultivated
Iresine	Amaranthaceae	LH	non cultivated
Alocasia	Araceae	LH	non cultivated
Hydrocotyle	Araliaceae	LH; LT; LS	non cultivated
Artemisia	Asteraceae	LH; LT	non cultivated
Baccharis	Asteraceae	LT	non cultivated
Carduus	Asteraceae	LH	non cultivated
Dichrocephala	Asteraceae	LH	non cultivated
Echinops	Asteraceae	LH	non cultivated
Eclipta	Asteraceae	LT	non cultivated
Emilia	Asteraceae	LH	non cultivated
Erechtites	Asteraceae	LH; LT	non cultivated
Galinsoga	Asteraceae	LH; LT; LS	non cultivated
Helipterum	Asteraceae	LH	non cultivated
Hemistepta	Asteraceae	LH	non cultivated
Hymenopappus	Asteraceae	LT	non cultivated
Lipochaeta	Asteraceae	LS	non cultivated
Melanthera	Asteraceae	LT	non cultivated
Parthenium	Asteraceae	LT	non cultivated
Synedrella	Asteraceae	LH; LT	non cultivated
Tridax	Asteraceae	LT	non cultivated
Verbesina	Asteraceae	LS	non cultivated
Xanthium	Asteraceae	LT	non cultivated
Cordia	Boranginaceae	LT	non cultivated

Genus	Family	Exotic *Liriomyza*	Host type
Capsella	Brassicaceae	LH	non cultivated
Rorippa	Brassicaceae	LH	non cultivated
Silene	Caryophillaceae	LH; LT	non cultivated
Malachium	Caryophyllaceae	LH	non cultivated
Spergula	Caryophyllaceae	LT	non cultivated
Commelina	Commelinaceae	LT	non cultivated
Calystegia	Convolvulaceae	LH	non cultivated
Convolvulus	Convolvulaceae	LT	non cultivated
Pharbitis	Convolvulaceae	LH	non cultivated
Ceratosanthes	Cucurbitaceae	LS	non cultivated
Melothria	Cucurbitaceae	LH	non cultivated
Melilotus	Fabaceae	LH; LS	non cultivated
Poissonia	Fabaceae	LS	non cultivated
Lamium	Lamiaceae	LH	non cultivated
Leonurus	Lamiaceae	LH	non cultivated
Melissa	Lamiaceae	LS	non cultivated
Anoda	Malvaceae	LS	non cultivated
Malva	Malvaceae	LH; LS	non cultivated
Sida	Malvaceae	LH; LS	non cultivated
Oxalis	Oxalidaceae	LH; LT	non cultivated
Piriqueta	Passifloraceae	LT	non cultivated
Linaria	Plantaginaceae	LT	non cultivated
Veronica	Plantaginaceae	LH	non cultivated
Limonium	Plumbaginaceae	LH	non cultivated
Polygala	Polygalaceae	LT	non cultivated
Rumex	Polygonaceae	LH	non cultivated
Cardiospermum	Sapindaceae	LT	non cultivated
Nemesia	Scrophulariaceae	LH	non cultivated
Datura	Solanaceae	LH; LT; LS	non cultivated
Nicotiana	Solanaceae	LH; LT	non cultivated
Kallstroemia	Zygophyllaceae	LT	non cultivated
Tribulus	Zygophyllaceae	LT	non cultivated

Table 17 Host genera of either L. huidobrensis, L. sativae or L trifolii with known overlap with native or naturalised agromyzids already present in Australasia. Some of these species are stem miner, which should be straightforward to distinguish from the leaf mining damage caused by L. huidobrensis, L. sativae or L trifolii. Records of L. sativae host plants within the Torres Strait Islands (TSI) and on the Cape York Peninsula (CYP) are included.

Host genus	Host family	Australasian Agromyzid genera	Sources
Beta	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977); Spencer (1990)
Chenopodium	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Spinacia	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Apium	Apiaceae	*Melanagromyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Daucus	Apiaceae	*Phytomyza*	Spencer (1973)
Asparagus	Asparagaceae	*Hexomyza*	Benavent Corai et al. (2005)
Ageratum	Asteraceae	*Melanagromyza*	Spencer (1977)
Arctium	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Aster	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Bellis	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Bidens	Asteraceae	*Melanagromyza*	Spencer (1977)
Callistephus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Carthamus	Asteraceae	*Melanagromyza*	Benavent Corai et al. (2005)
Chrysanthemum	Asteraceae	*Ophiomyia, Phytomyza, Melanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Cineraria	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cirsium	Asteraceae	*Phytomyza*	Spencer (1977)
Conyza	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Coreopsis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Craspedia	Asteraceae	*Liriomyza*	Benavent Corai et al. (2005)
Crepis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cynara	Asteraceae	*Phytomyza*	Spencer (1973)
Dahlia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Erigeron	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005); Spencer (1977)
Gaillardia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gerbera	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gnaphalium	Asteraceae	*Melanagromyza, Malanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Helianthus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Helichrysum	Asteraceae	*Liriomyza, Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Lactuca	Asteraceae	*Liriomyza*, Phytomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1973)
Leucanthemum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Picris	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Senecio	Asteraceae	*Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1977)
Solidago	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005)
Sonchus	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Tagetes	Asteraceae	*Liriomyza**	Present in TSI/CYP

Host genus	Host family	Australasian Agromyzid genera	Sources
Taraxacum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Tecoma	Bignoniaceae	*Liriomyza**	Present in TSI/CYP
Barbarea	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Brassica	Brassicaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Cardamine	Brassicaceae	*Liriomyza*	Spencer (1977)
Diplotaxis	Brassicaceae	*Liriomyza*	Spencer (1977)
Hirschfeldia	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Matthiola	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977)
Raphanus	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Silene	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Stellaria	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Cleome	Cleomaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP
Ipomoea	Convolvulaceae	*Melanagromyza*	Benavent Corai et al. (2005)
Citrullus	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucumis	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucurbita	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Euphorbia	Euphorbiaceae	*Tropicomyia*	Spencer (1977)
Ricinus	Euphorbiaceae	*Liriomyza**	Present in TSI/CYP
Cajanus	Fabaceae	*Melanagromyza, Ophiomyia*	Spencer (1973); Spencer (1977)
Canavalia	Fabaceae	*Ophiomyia*	Spencer (1973)
Cassia	Fabaceae	*Tropicomyia, Ophiomyia*	Spencer (1977); Spencer (1990)
Crotalaria	Fabaceae	*Liriomyza*, Ophiomyia*	Present in TSI/CYP; Spencer (1973); Spencer (1977)
Dolichos	Fabaceae	*Ophiomyia*	Spencer (1973); Spencer (1977)
Glycine	Fabaceae	*Melanagromyza, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Indigofera	Fabaceae	*Liriomyza*, Ophiomyia*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977); Spencer (1990)
Lablab	Fabaceae	*Ophiomyia*	Spencer (1973)
Macroptilium	Fabaceae	*Liriomyza*, Tropicomyia*	Present in TSI/CYP; Spencer (1977)
Millettia	Fabaceae	*Unknown*	Pirtle pers. comm.
unknown	Fabaceae	*Melanagromyza*	Spencer (1977)
Phaseolus	Fabaceae	*Ophiomyia, Tropicomyia, Melanagromyza, Ophiomyla*	Spencer (1973); Spencer (1977)
Pisum	Fabaceae	*Liriomyza, Phytomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Senna	Fabaceae	*Tropicomyia*	Spencer (1977)
Sesbania	Fabaceae	*Liriomyza**	Present in TSI/CYP
Trifolium	Fabaceae	*Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Vigna	Fabaceae	*Liriomyza*, Melanagromyza, Tropicomyia, Ophiomyia*	Present in TSI/CYP; Pirtle et al. (2020); Spencer (1973); Spencer (1977); Spencer 1977
Scaevola	Goodeniaceae	*Liriomyza, Ophiomyia*	Benavent Corai et al. (2005); Pirtle pers. comm.; Spencer (1977)

Host genus	Host family	Australasian Agromyzid genera	Sources
Hydrangea	Hydrangeaceae	*Tropicomyia*	Spencer (1977)
Ocimum	Lamiaceae	*Liriomyza**	Pirtle et al. (2020)
Stephania	Menispermaceae	*Tropicomyia*	Spencer (1977); Spencer (1990)
Passiflora	Passifloraceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977); Spencer (1990)
Turnera	Passifloraceae	*Liriomyza**	Pirtle et al. (2020)
Plantago	Plantaginaceae	*Liriomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977);
Hordeum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
unknown	Poaceae	*Agromyza, Cerodontha*	Spencer (1973)
Setaria	Poaceae	*Agromyza*	Benavent Corai et al. (2005)
Triticum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
Zea	Poaceae	*Pseudonapomyza*	Spencer (1973)
Portulaca	Portulacaceae	*Liriomyza*	Benavent Corai et al. (2005)
Ranunculus	Ranunculaceae	*Phytomyza, Napomyza*	Spencer (1977); Spencer (1990)
Passiflora	Resedaceae	*Tropicomyia*	Spencer (1990)
Coffea	Rubiaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Citrus	Rutaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Capsicum	Solanaceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977)
Cestrum	Solanaceae	*Tropicomyia*	Spencer (1977)
Lycopersicum	Solanaceae	*Liriomyza**	Present in TSI/CYP
Petunia	Solanaceae	*Liriomyza**	Present in TSI/CYP
Physalis	Solanaceae	*Liriomyza**	Present in TSI/CYP; Pirtle et al. (2020)
Solanum	Solanaceae	*Ophiomyia, Liriomyza**	Pirtle pers. comm.; Present in TSI/CYP; Spencer (1977)
Tropaeolum	Tropaeolaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Lantana	Verbenaceae	*Calycomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Stachytarpheta	Verbenaceae	*Liriomyza**	Pirtle et al. (2020)
Verbena	Verbenaceae	*Calycomyza*	Benavent Corai et al. (2005)

* Includes *Liriomyza sativae* within TSI/CYP

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Appendix 3: Permits for the control of *Liriomyza* spp. on different host crops

Table lists all commercial hosts of vegetable leafminer (refer to Appendix 1) and notes where suitable pesticides are available and approved by the APVMA for control on each host crop. Blank cells denote where suitable pesticides have not yet been approved or identified. Refer to the APVMA permit database (https://portal.apvma.gov.au/permits) for the most up to date information on current leafminer permits.

Table 18 Insecticides control options for different crop groups

Relevant Crop group	Crop/host plant	Pesticide	Permit number(s)/ status	Target pest (as per permit)	Expiration date	Withholding information/notes	Suitable for management or eradication[6]
Brassica vegetables	Bok choy; pak-choy; turnip	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Broccoli	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Broccoli	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

[6] Some pesticides will be more suitable for eradication than for the ongoing management of the pest. This is because either they will need to be used at high rates to be effective, there are potential resistance issues, or the pesticide is not likely to fit into existing IPM systems as well as alternative pesticides. Other chemicals provide good control of the pest and are also compatible with IPM systems and will be more suitable for the ongoing management of the pest. To highlight this, pesticides are noted for either eradication, or for management and eradication.

Brassica vegetables	Brussels sprouts	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Brussels sprouts	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cabbage	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cauliflower	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica	Cauliflower	Emamectin	PER87563	*Liriomyza* spp.	30 June 2024	3 days (harvest)	Management or

vegetables		benzoate		(including*: L. sativae*)		Do not graze or cut for stock food	eradication
Brassica vegetables	Indian mustard	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Bulb vegetables	Garlic	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Garlic	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	30 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae,*	31 December 2023	7 days (harvest) Do not graze or cut for	Management or eradication

				L trifolii, L. huidobrensis)		stock food	
Bulb vegetables	Shallots	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Shallots	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Cereal grains	Barley	No suitable pesticides identified					
Fruit trees (non-bearing)	Any trees that will not bear fruit for the next 12 months	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables	Angled luffa, sing-kwa	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:*	31 December 2023	1 day (harvest) Do not graze or cut for	Management or eradication

(Cucurbits – including melons)				*L. sativae, L trifolii, L. huidobrensis*)		stock food	
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits –	Cucumber	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae,*	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

including melons)				*L trifolii, L. huidobrensis*)			
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including	Pumpkin (e.g. butternut)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii,*	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

melons)				*L. huidobrensis*)			
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Wax gourd	Cyromazine	PER81867	*Liriomyza*	30 November	7 days (harvest)	Management or

vegetables (Cucurbits – including melons)				*sativae* and *L. huidobrensis*	2023	Do not graze or cut for stock food	eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than	Bell pepper	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

cucurbits)							
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Eggplant	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Eggplant	Spirotetramat	PER88640	*Liriomyza* spp.	31 May 2023	1 day (harvest)	Management

vegetables (other than cucurbits)				(including: *L. sativae, L trifolii, L. huidobrensis*)		Do not graze or cut for stock food	
Fruiting vegetables (other than cucurbits)	Okra*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Okra*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including: *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Okra*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including: *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than	Tomato	Spirotetramat	PER88640	*Liriomyza* spp. (including: *L. sativae,*	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management

cucurbits)				*L trifolii, L. huidobrensis*)			
Herbs	Basil	No suitable pesticides identified					
Herbs	Oregano	No suitable pesticides identified					
Herbs	Parsley[7]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management
Head lettuce	Lettuce (Head type only)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Lettuce	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Leafy vegetables (including brassica leafy vegetables)	Spinach	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables	Spinach	Cyromazine	PER81867	*Liriomyza sativae* and	30 November 2023	**Treated crop must be destroyed.**	**Eradication only**

[7] Note: Parsley is not a recorded host for *L. sativae* but may be a host of other *Liriomyza* spp. and is listed as one of the crops that the permit covers.

(including brassica leafy vegetables)				*L. huidobrensis*		Treated crops must not be made available for human consumption Do not graze or cut for stock food	
Leafy vegetables (including brassica leafy vegetables)	Spinach and silverbeet	Chlorantraniliprole	PER87631	*Liriomyza* spp. (including*:* *L. sativae, L brassicae, L. huidobrensis*)	30 June 2024	3 days (harvest) 7 days (grazing or cutting for stock food)	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	7 days (harvest) 7 days (grazing and hay)	Management
Legume vegetables	Faba bean, broad bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Faba bean, broad bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spinetoram	PER87878	*Liriomyza* spp.	28 February 2023	3 days (harvest); 14 days (grazing and hay) Do not allow dairy cattle to graze treated forage	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest); 3 days (grazing and hay)	Management
Legume vegetables	Red bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Red bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Not classified	Field mustard	No suitable					

		pesticides identified					
Not classified	Lucerne	No suitable pesticides identified					
Nursery stock (non-food)	Various non-food nursery plants (e.g. chrysanthemum etc.)	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Nursery stock (non-food)	Seedlings, tubes and plugs, potted colour, trees and shrubs, foliage plants, palms, grasses, fruiting plants (non-bearing), cut flowers and ornamentals.	Abamectin Azadirachtin Cyromazine Emamectin Chlorantraniliprole + thiamethoxam Cyantraniliprole Indoxacarb Spinetoram	PER88977	Leafminers (*Liriomyza* spp.) including Vegetable leafminer (*Liriomyza sativae*)	30 November, 2022	Not required when used as directed	Management or eradication
Oilseeds	Cotton	Spinetoram has been identified as suitable for the control of Vegetable leafminer and other *Liriomyza* spp. on cotton as overseas and Australian use patterns are comparable. However, no permit has been applied for this use to date					
Pulses	Pulses	Dimethoate	PER89184	*Liriomyza sativae, Liriomyza trifolii, Liriomyza huidobrensis*	31 March 2025	Harvest: Do not harvest for 14 days after application. Grazing: Do not graze or cut for stock food for 14 days after application.	Management
Root and tuber vegetables	Beets (beetroot, silver beet, sugar beets)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Beets (beetroot, silver beet, sugar beets)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Root and tuber vegetables	Carrot	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Carrot	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Potato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Potato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Potato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Radish	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Radish	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Celery	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem	Celery	Spirotetramat	PER88640	*Liriomyza* spp.	31 May 2023	3 days (harvest)	Management or

vegetables				(including*: L. sativae, L trifolii, L. huidobrensis*)		Do not graze or cut for stock food	eradication
Stalk and stem vegetables	Rhubarb[8]	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication

[8] Note: Rhubarb is not a recorded host for *L. sativae or L. huidobrensis* but may be a host of other *Liriomyza* spp. and is part of the 'Stalk and Stem Vegetable Crop Group' that the permit covers.

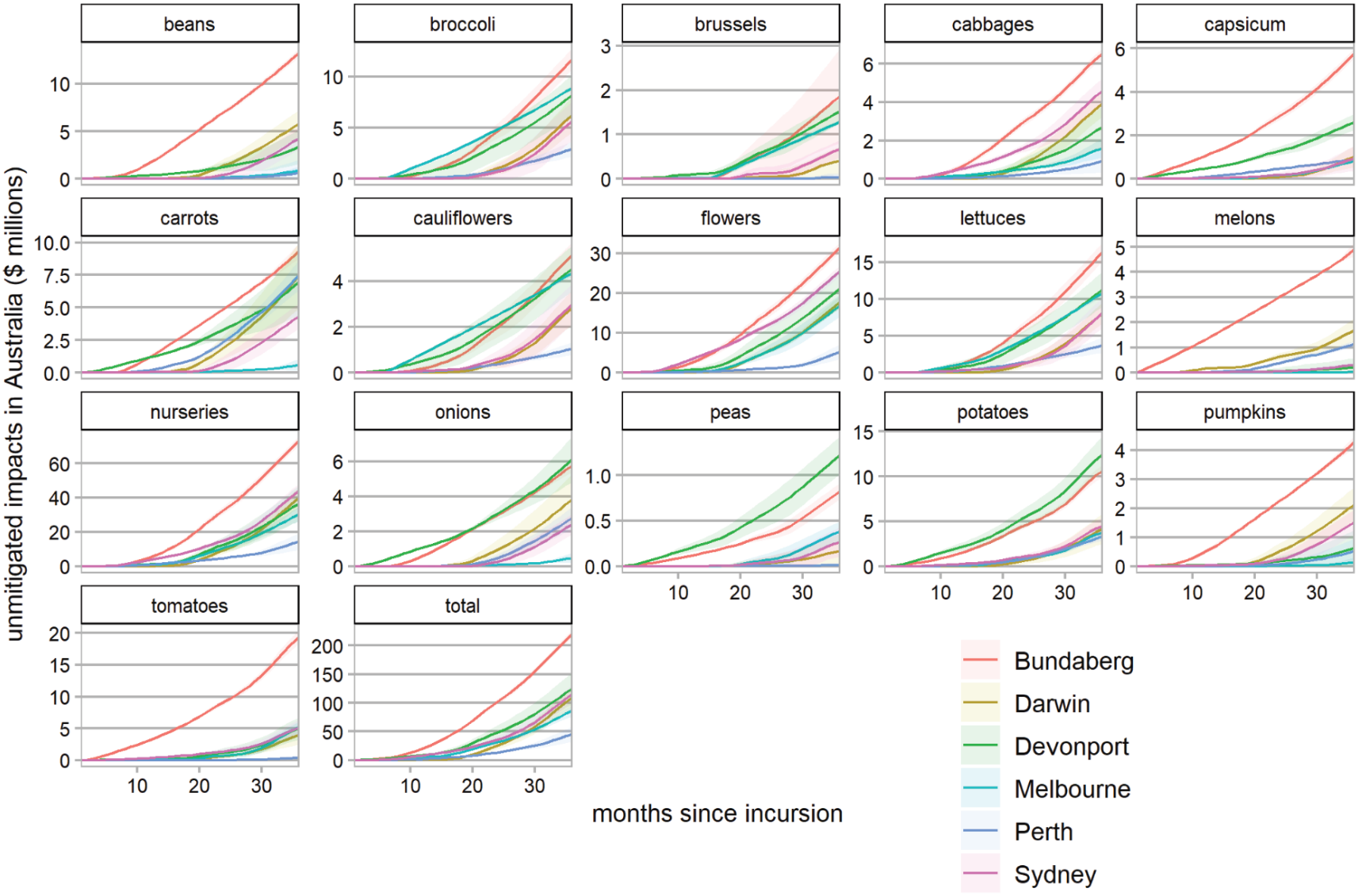
Appendix 4: Additional impact prediction outputs

Table 18 State level accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. sativae *at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preferences hosts may be overestimated while impacts of high preference hosts may be underestimated. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.*

INCURSION LOCATION															
	NSW		NT		QLD			SA	TAS	VIC		WA			
Crop	**Griffith**	**Sydney**	**Darwin**	**Katherine**	**Bowen**	**Bundaberg**	**Toowoomba**	**Adelaide**	**Devonport**	**Ballarat**	**Melbourne**	**Albany**	**Carnarvon**	**Geraldton**	**Perth**
Beans	0.00 (0.00)	0.11 (0.13)	0.07 (0.00)	0.00 (0.00)	10.57 (1.29)	12.51 (0.22)	11.75 (0.80)	0.00 (0.00)	1.24 (0.00)	1.07 (0.42)	0.67 (0.13)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.02 (0.00)
Broccoli	0.13 (0.00)	0.14 (0.01)	0.00 (0.00)	0.00 (0.00)	1.91 (0.84)	6.44 (0.21)	7.58 (0.05)	0.31 (0.03)	0.38 (0.00)	8.97 (0.73)	8.79 (0.08)	0.08 (0.05)	0.00 (0.00)	0.00 (0.00)	2.60 (0.17)
Brussels	0.00 (0.00)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.02)	0.12 (0.02)	0.14 (0.01)	5.34 (0.10)	0.43 (0.00)	1.37 (0.26)	1.28 (0.06)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Cabbages	0.03 (0.05)	2.71 (0.16)	0.00 (0.00)	0.00 (0.00)	1.22 (0.47)	3.73 (0.15)	4.42 (0.02)	0.24 (0.01)	0.12 (0.00)	1.05 (0.13)	0.97 (0.06)	0.01 (0.02)	0.00 (0.00)	0.00 (0.00)	0.55 (0.02)
Capsicum	0.17 (0.00)	0.17 (0.01)	0.00 (0.00)	0.00 (0.00)	9.67 (0.21)	4.73 (0.21)	2.35 (0.58)	1.82 (0.27)	1.47 (0.00)	1.03 (0.30)	0.77 (0.08)	0.06 (0.02)	0.00 (0.00)	0.00 (0.00)	0.87 (0.00)
Carrots	0.09 (0.01)	0.23 (0.02)	0.00 (0.00)	0.00 (0.00)	3.20 (1.02)	8.34 (0.16)	8.73 (0.19)	3.95 (0.22)	4.69 (0.00)	0.37 (0.07)	0.33 (0.02)	0.08 (0.11)	0.00 (0.00)	0.00 (0.00)	6.75 (0.36)
Cauliflowers	0.01 (0.02)	0.65 (0.08)	0.00 (0.00)	0.00 (0.00)	0.67 (0.27)	2.34 (0.10)	2.94 (0.03)	0.41 (0.04)	1.00 (0.00)	4.16 (0.37)	4.17 (0.00)	0.06 (0.03)	0.00 (0.00)	0.00 (0.00)	0.90 (0.02)
Flowers	0.41 (0.71)	18.27 (0.54)	0.21 (0.00)	0.00 (0.00)	1.62 (0.56)	5.97 (0.25)	5.21 (0.21)	1.92 (0.07)	0.80 (0.00)	11.37 (0.98)	10.46 (0.22)	0.22 (0.05)	0.00 (0.00)	0.00 (0.00)	2.09 (0.04)
Lettuces	0.01 (0.02)	1.35 (0.04)	0.00 (0.00)	0.00 (0.00)	2.44 (0.93)	8.50 (0.33)	9.96 (0.10)	0.84 (0.08)	0.01 (0.00)	11.06 (0.85)	10.41 (0.25)	0.06 (0.10)	0.00 (0.00)	0.00 (0.00)	3.24 (0.07)

	INCURSION LOCATION														
Melons	3.64 (0.00)	0.06 (0.01)	0.80 (0.01)	0.00 (0.00)	6.03 (0.08)	4.80 (0.26)	2.14 (1.48)	0.01 (0.01)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.02 (0.03)	0.00 (0.00)	0.00 (0.00)	1.10 (0.09)
Nurseries	3.14 (0.70)	25.63 (1.66)	0.37 (0.00)	0.00 (0.00)	8.05 (2.79)	26.56 (1.64)	26.77 (0.75)	5.53 (0.14)	0.79 (0.00)	25.17 (0.85)	22.72 (0.60)	0.14 (0.18)	0.00 (0.00)	0.00 (0.00)	9.43 (0.22)
Onions	0.34 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	1.82 (0.63)	5.26 (0.09)	5.81 (0.06)	4.22 (0.39)	4.75 (0.01)	0.44 (0.08)	0.36 (0.06)	0.01 (0.01)	0.00 (0.00)	0.00 (0.00)	2.41 (0.34)
Peas	0.00 (0.00)	0.03 (0.00)	0.00 (0.00)	0.00 (0.00)	0.02 (0.01)	0.47 (0.00)	0.17 (0.06)	0.00 (0.00)	0.74 (0.00)	0.50 (0.11)	0.38 (0.10)	0.02 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.00)
Potatoes	8.45 (0.04)	1.55 (0.11)	0.00 (0.00)	0.00 (0.00)	1.50 (0.51)	6.22 (0.38)	4.32 (0.48)	3.47 (0.18)	7.57 (0.01)	5.59 (0.57)	3.44 (0.47)	0.03 (0.03)	0.00 (0.00)	0.00 (0.00)	3.05 (0.22)
Pumpkins	2.38 (0.01)	0.20 (0.01)	0.00 (0.00)	0.00 (0.00)	3.14 (0.47)	3.98 (0.20)	4.03 (0.25)	0.00 (0.00)	0.13 (0.00)	0.02 (0.00)	0.02 (0.00)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.31 (0.03)
Tomatoes	1.00 (0.05)	2.02 (0.05)	0.01 (0.00)	0.00 (0.00)	18.53 (0.56)	13.73 (0.33)	7.12 (1.26)	0.77 (0.13)	0.35 (0.00)	5.90 (1.34)	4.57 (0.96)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.10 (0.02)
Total	**19.80 (1.58)**	**53.12 (2.47)**	**1.47 (0.02)**	**0.00 (0.00)**	**70.38 (10.21)**	**113.70 (3.15)**	**103.45 (5.38)**	**28.83 (0.83)**	**24.48 (0.02)**	**78.07 (4.69)**	**69.35 (1.67)**	**0.78 (0.51)**	**0.00 (0.00)**	**0.00 (0.00)**	**33.41 (1.28)**

Figure 11 Australia wide accumulating unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. sativae *at key entry and establishment points across Australia.*



Appendix 12: *Liriomyza trifolii* Continency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Rohan Burgess [1], Dr. Peter Ridland [2], Dr. Elia Pirtle [3]

[1] Plant Health Australia

[2] The University of Melbourne

[3] Cesar Australia

December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Liriomyza trifolii
Contingency Plan

Prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Rohan Burgess [1], Dr. Peter Ridland [2], Dr. Elia Pirtle [3]

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[3] Cesar Australia

December 7, 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

CONTINGENCY PLAN

AMERICAN SERPENTINE LEAFMINER (*LIRIOMYZA TRIFOLII*)



Central Science Laboratory, Harpenden, British Crown, Bugwood.org



Central Science Laboratory, Harpenden, British Crown, Bugwood.org

December 2020

This resource has been funded by Horticulture Innovation, through the Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*), using multiple research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

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1. Purpose and background of this contingency plan **6**
2. Pest details **7**
2.1 Background and impact of pest 7
2.2 Life cycle 7
2.3 Host range 8
2.4 Signs and symptoms 8
2.5 Dispersal 10
2.6 Current geographic distribution 10
2.7 Risk of establishment in Australia 14
2.8 Risk of spread within Australia 15
2.8.1 Risk pathways – international and domestic 15
2.8.2 Predicting spread potential 19
2.9 Risk of economic impact within Australia 20
3. Diagnostic information **23**
3.1 Diagnostic considerations 23
4. Surveillance and collection of samples **25**
4.1 Surveillance 25
4.1.1 Technical information for planning surveys 25
4.1.2 Delimiting surveys in the event of an incursion into new areas 28
4.1.3 Activities for general surveillance immediately following a detection 30
4.2 Collection and treatment of samples 30
4.2.1 If collecting for morphological diagnosis 30
4.2.2 If collecting for molecular diagnosis 31
5. Immediate response to detection **34**
5.1 Tracing 34
5.2 Quarantine and movement controls 34
5.3 Destruction strategy 34
5.3.1 Priorities and considerations 34
5.3.2 Decontamination of tools and equipment 35
5.3.3 Chemical control 35
5.3.4 Physical control options 40
5.3.5 Biological control 40
5.4 Decision support for eradication 41
6. Emergency Plant Pest Response Deed and PLANTPLAN **46**
6.1 Emergency Plant Pest Response Deed 46
6.2 PLANTPLAN 47
7. References **48**
8. Appendices **54**
Appendix 1: Known hosts of American serpentine leafminer 54
Number of host records for the American serpentine leafminer (*Liriomyza trifolii*) within plant families 54
Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*) has been

provided within the scientific literature....56
Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*) has been provided within the scientific literature....78
Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*)....84
References....90
Appendix 2: Leaf mining damage caused by established species....93
References....104
Appendix 3: Permits for the control of American serpentine leafminer and other *Liriomyza* spp. on different host crops....105
Appendix 4: Additional impact prediction outputs....105

1. PURPOSE AND BACKGROUND OF THIS CONTINGENCY PLAN

The American serpentine leafminer (*Liriomyza trifolii*) is an exotic leafminer that affects a wide range of horticultural industries, including the vegetable and melon industries. It is currently widespread overseas and is present in nearby countries such as Indonesia.

This Contingency Plan provides background information on American serpentine leafminer to assist in determining the requirements for the initial response to a detection of this species in Australia. Only key information for immediate response is provided in this document.

Additional information can be found in the following supporting material:

- Awareness material such as the fact sheets from Plant Health Australia (PHA), AUSVEG, and commonwealth, state and territory jurisdictions.
    - www.planthealthaustralia.com.au/pests/american-serpentine-leaf-miner/
    - www.agriculture.gov.au/pests-diseases-weeds/plant/leaf-miner
    - www.dpi.nsw.gov.au/biosecurity/plant/insect-pests-and-plant-diseases/exotic-leaf-miners
    - https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/
- Overseas websites with additional information
    - https://gd.eppo.int/taxon/LIRITR
    - www.cabi.org/cpc/datasheet/30965
    - https://cipotato.org/riskatlasforafrica/liriomyza-trifolii/
    - http://entnemdept.ufl.edu/creatures/veg/leaf/a_serpentine_leafminer.htm
    - http://ipm.ucanr.edu/PMG/r280300911.html
    - http://ipm.ucanr.edu/PMG/r783300911.html

2. PEST DETAILS

Preferred common name	American Serpentine Leafminer
Other common names	Serpentine leafminer; Broad bean leafminer; Californian leafminer; Celery leafminer; Chrysanthemum leafminer
Scientific name:	*Liriomyza trifolii* Burgess
Synonyms:	*Agromyza phaseolunata;* *Liriomyza alliivora;* *Liriomyza phaseolunata;* *Oscinis trifolii*
Taxonomic position:	Class: Insecta Order: Diptera Family: Agromyzidae Genus: *Liriomyza* Species: *Liriomyza trifolii*

2.1 Background and impact of pest

The Agromyzidae are a well-known group of small, morphologically similar flies whose larvae feed internally on plants, often as leaf and stem miners. Nearly all species are very host-specific, but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world.

The American serpentine leafminer is of concern as it has a wide host range including many horticulture crops in which it can cause significant yield losses and quality reductions (see Appendix 1). The American serpentine leafminer is widespread overseas being present in North and South America, Asia, Africa, Europe and some Pacific islands.

2.2 Life cycle

The lifecycle of the American serpentine leafminer consists of an egg inserted by the female just under the surface of a leaf of a host plant when temperatures are above 12°C (Capinera 2014). The egg hatches in 2-5 days (CABI 2018). The larvae then begin to feed within the leaf creating tunnels or mines that get larger as the larvae moult and mature. After passing through three larval stages in 4+ days the larva leaves the plant to form a puparium in the soil underneath the host plant (or occasionally inside the leaf on some hosts such as onions (CABI and EPPO date unknown)). After 7-14 days (at 20°C and 30°C) an adult emerges and begins to reproduce (Leibee 1984; CABI 2018). The times taken to complete each life stage vary depending on host and temperature. Leibee (1984) determined growth at a constant 25°C, and reported that it takes 19 days from egg deposition to emergence of the adult. Development rates become quicker as temperatures increase, with the

complete life cycle taking only 12 days on celery at 35°C (Leibee 1982). Similarly, development times become longer at low temperatures. American serpentine leafminer are reported to have a diapause (delay of development) at low temperatures allowing them to survive winter conditions.

2.3 Host range

American serpentine leafminer is a polyphagous pest of many agricultural and ornamental hosts (see Appendix 1It has been recorded from ~170 plant species representing 29 plant families. Its preferred hosts tend to be in the Asteraceae family (Spencer 1973).

Important agricultural hosts include sunflower, safflower, lettuce, celery, cucumber, melons, onion, potato, tomato, eggplant, legume vegetables and pulse crops and a wide range of ornamental species such as chrysanthemum.

A detailed host list is included in Appendix 1.

2.4 Signs and symptoms

Adult flies are small (less than 2 mm long with a wing length of 1.25-1.9 mm) with a shiny black mesonotum and yellow markings on the head and body (Figure 1).

Female flies use their ovipositor to puncture the leaves of the host plants causing wounds which serve as sites for feeding (by both males and females) or oviposition. Feeding punctures appear as white speckles between 0.13 and 0.15 mm in diameter. Oviposition punctures are usually smaller (0.05 mm) and are more uniformly round. However, the appearance of the punctures does not differ between *Liriomyza* species so cannot used to separate species.

Pale coloured leaf mines are created by the feeding larvae and are the most obvious symptom of infestations of *Liriomyza* spp. (Figure 2). The larval mine of American serpentine leafminer is on the upper surface of the leaf in the leaf mesophyll tissue, it is shallow, at first greenish, then later whitish in colour. The trails of frass are distinctive in being deposited in black strips alternately at either side of the mine (like the related Vegetable leafminer, *L. sativae*), but becomes more granular towards the end of the mine (unlike *L. sativae*) (Spencer 1973).

Factsheets with more information on what to look for in the field can be found at the following websites:

- www.planthealthaustralia.com.au/pests/american-serpentine-leaf-miner/
- http://entnemdept.ufl.edu/creatures/veg/leaf/a_serpentine_leafminer.htm
- http://ipm.ucanr.edu/PMG/r783300911.html
- https://ausveg.com.au/biosecurity-agrichemical/biosecurity/mt16004/



Figure 1 Adult American serpentine leafminer. Source: Central Science Laboratory, Harpenden, British Crown, Bugwood.org



Figure 2 Leaf mines on chrysanthemum. Source: Central Science Laboratory, Harpenden, British Crown, Bugwood.org

2.5 Dispersal

Typically, *Liriomyza* leafminers are considered to have invaded countries via the movement of infested plants (generally ornamentals). While fully formed mines should be readily visible, signs of early infestations are much less obvious and are easily overlooked.

Agromyzid flies are considered as "moderate fliers" (Yoshimoto and Gressitt 1964) that also have the potential to be spread over large distances through wind dispersal. Flight mill experiments on the related *L. sativae* have shown that leafminers can fly up to 8.22 km but that the average flight distances was<1.0 km at 18-36°C (Lei et al., 2002). Similar flight ability is expected for American serpentine leafminer.

Individual American serpentine leafminers within glasshouses have been shown to disperse on average 26 m but up to 100 m from original infestation points within 24 hours (Jones and Parella 1986) and in a field study were generally not found more than 50 m away from infested glasshouses (Ozawa et al 1999). Similarly, *Liriomyza huidobrensis* adults have been shown to disperse on average 46 m within 24 hours via a release recapture experiment (Fenoglio et al 2019). *Agromyza frontella*, a related agromyzid leafminer pest of lucerne with similar flight dynamics, was observed to spread at a rate of about 100 km per year (or averaging about 0.3 kms per day) during an incursion into the Midwestern United States (Venette et al. 1980). There is also some evidence of long distance dispersal of agromyzids via wind, with agromyzid species reportedly trapped as high as 900 min the air column (Hardy and Milne 1938; Glick 1960; White 1970), and over oceans (Cheng 1976, Yoshimoto and Gressit 1964).

However, windborne dispersal is likely to be a minor pathway of leafminer dispersal in Australia, and human-mediated pathways should be prioritised, particularly given the accumulation of research pointing to this as a major contributor to the movement of invasive species (Capinha et al. 2015; Wichmann et al. 2009). Such pathways include transport of contaminated produce, or "hitch-hiking" adults associated with the large volume of human movement.

The American serpentine leafminer has spread from North America to Central and South America, Europe, Africa and Asia through the movement of plant material (cuttings, cut flowers, etc) (Minkenberg and Van Lenteren 1986). This suggests that there is the potential for it to spread rapidly to new regions. Plant material should be considered as a potential pathway for the introduction and spread of this pest. The introduction of plant material from overseas is regulated, however illegal movement of plant material poses a potential risk for the entry of the pest into Australia.

2.6 Current geographic distribution

The American serpentine leafminer is thought to have originated in North America (Spencer 1973) and since spread to South America, Asia, Africa and Europe.

The current distribution of this pest is presented in Table 1. Note, American serpentine leafminer has also been found at various times in a number of European countries including; Czech Republic, Denmark, Finland, Hungary, Ireland, Netherlands, Poland, Slovenia, Sweden, United Kingdom, but has since been eradicated and is no longer found in those countries (EFSA Panel on Plant Health 2012).

Table 1. Countries where American serpentine leafminer is known to occur

COUNTRY/REGION	REFERENCE
American Samoa	CABI and EPPO (date unknown)
Angola	Černý and Von Tschirnhaus (2014)
Austria	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Bahamas	CABI and EPPO (date unknown); Spencer (1973)
Barbados	CABI and EPPO (date unknown); Spencer (1973)
Belgium	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Brazil	CABI and EPPO (date unknown); De Freitas Bueno et al., (2007)
Cambodia	Gao et al., (2017)
Canada	Broadbent et al., (1986); CABI and EPPO (date unknown); Parrella and Keil (1984); Černý and Von Tschirnhaus (2014); Spencer (1973)
China	Gao et al., (2011); Gao et al., (2017)
Colombia	CABI and EPPO (date unknown); Parrella and Keil (1984)
Costa Rica	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Spencer (1983)
Cuba	Altieri et al., (1999); CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014)
Cyprus	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); EFSA Panel on Plant Health (2012)
Dominican Republic	CABI and EPPO (date unknown); Serra et al., (2003)
Egypt	Abd-Rabou (2006); CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014)
Ethiopia	Abate (1988); CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014)
Federated States of Micronesia	Schreiner and Nafus (1986)
France	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
French Guiana	CABI and EPPO (date unknown)
Greece	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Guadeloupe	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Spencer et a., (1992)
Guam	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Schreiner and Nafus (1986)
Guatemala	CABI and EPPO (date unknown)
Guinea	Černý and Von Tschirnhaus (2014)

COUNTRY/REGION	REFERENCE
Guyana	CABI and EPPO (date unknown); Spencer (1973)
India	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Gao et al., (2017); Srinivasan et al., (1995)
Indonesia	Baliadi and Tengkano et al., (2010); Gao et al., (2017)
Iran	Doust (2010); Gao et al., (2017)
Israel	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Weintraub and Horowitz, (1995)
Italy	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Japan	Arakaki and Kinjo (1998); CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Gao et al., (2017)
Jordon	Al-Ghabeish and Allawi (2001); Gao et al., (2017)
Kenya	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); De Lima (1979)
Korea	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Han et al., (1996); Gao et al., (2017)
Lebanon	CABI and EPPO (date unknown); Gao et al., (2017)
Madagascar	Černý and Von Tschirnhaus (2014)
Malta	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Martinique	CABI and EPPO (date unknown); Etienne and Martinez (2013)
Mauritius	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Dove (1983)
Mexico	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Scheffer and Lewis (2006)
Micronesia	CABI and EPPO (date unknown); Suta and Esguerra (1993)
Nigeria	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Umeh et al., (2002)
Northern Mariana Islands	CABI and EPPO (date unknown); Schreiner and Nafus (1986)
Oman	Gao et al., (2017); Černý and Von Tschirnhaus (2014); Deadman et al., (2000)
Peru	CABI and EPPO (date unknown)
Philippines	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Gao et al., (2017); Scheffer et al., (2006)
Portugal	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)

COUNTRY/REGION	REFERENCE
Republic of Benin	Černý and Von Tschirnhaus (2014)
Republic of Cote d'Ivoire	Černý and Von Tschirnhaus (2014)
Réunion	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Vercambre (1980)
Romania	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Samoa	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014)
Saudi Arabia	Al-Khateeb and Al-Jabr (2004); Gao et al., (2017)
Senegal	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Diatte et al., (2018)
Serbia	Dobrosavljevic et al., (2017)
South Africa	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Daiber (1985)
Spain (including Canary Islands)	CABI and EPPO (date unknown); EFSA Panel on Plant Health (2012)
Sri Lanka	Niranjana et al., (2005)
Sudan	Černý and Von Tschirnhaus (2014)
Switzerland	CABI and EPPO (date unknown)
Taiwan	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Gao et al., (2017); Wang and Lin (1988)
Tanzania	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014)
Thailand	Gao et al., (2017)
Tonga	CABI and EPPO (date unknown)
Trinidad and Tobago	CABI and EPPO (date unknown)
Tunisia	Černý and Von Tschirnhaus (2014); CABI and EPPO (date unknown)
Turkey	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); Gao et al., (2017)
United Arab Emirates	Gao et al., (2017); Howarth (2006)
United States of America (including Hawaii)	CABI and EPPO (date unknown); Černý and Von Tschirnhaus (2014); De Freitas Bueno et al., (2007); Parrella and Keil (1984); Spencer (1973)
Venezuela	CABI and EPPO (date unknown); Spencer (1973)
Vietnam	Gao et al., (2017);
Yement	Černý and Von Tschirnhaus (2014); Gao et al., (2017)
Yugoslavia	CABI and EPPO (date unknown)

COUNTRY/REGION	REFERENCE
Zambia	Černý and Von Tschirnhaus (2014)
Zimbabwe	Černý and Von Tschirnhaus (2014)

2.7 Risk of establishment in Australia

CLIMEX models are provided in Jovicich (2009). These found that the American serpentine leafminer is most likely to establish along the northern, eastern, southern and southwestern coastline of Australia and Tasmania.

Refer to: https://ausveg.com.au/app/data/technical-insights/docs/VG06113.pdf for further details.

Cesar Australia has created a model of the potential geographic distribution of the American serpentine leafminer in Australia, the results of which are summarised below (Figure 3). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

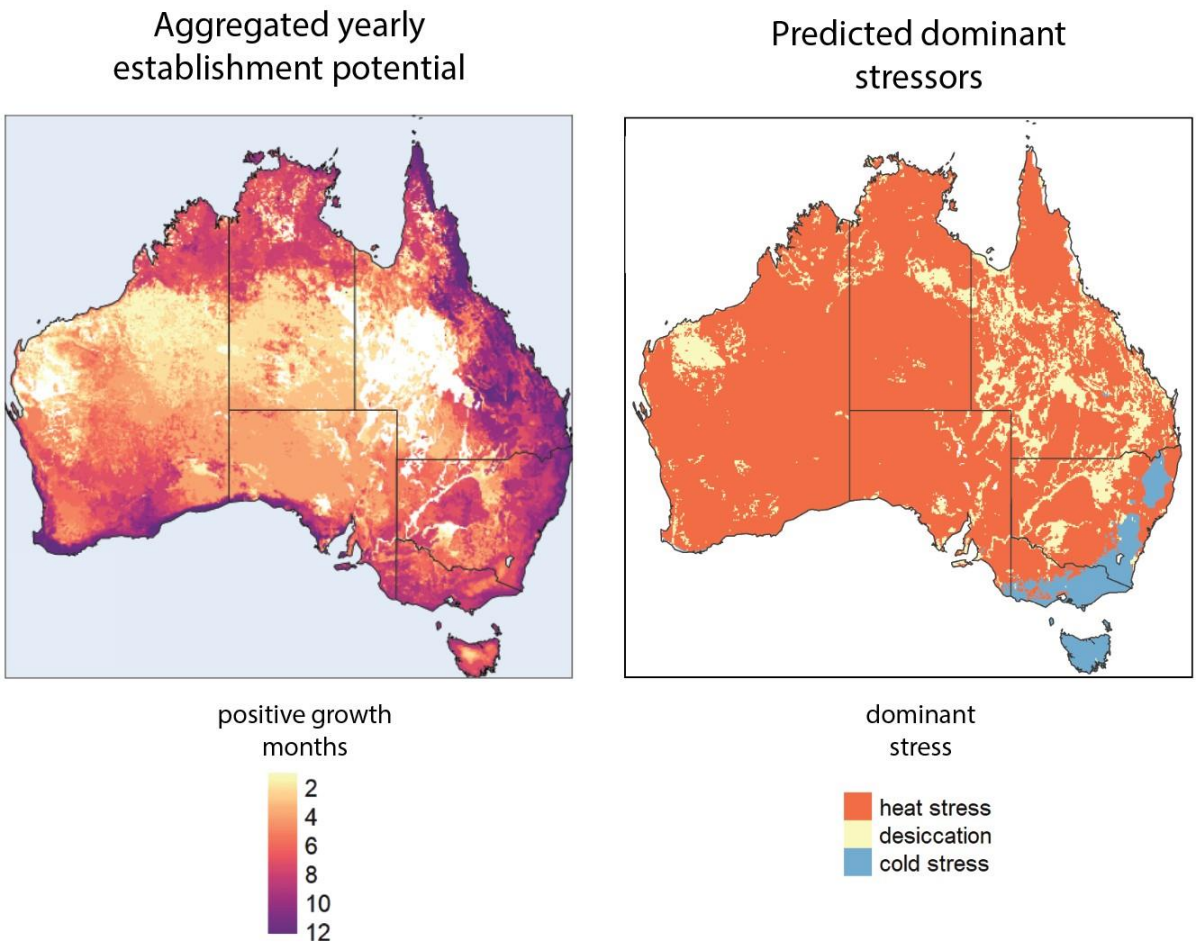


Figure 3 Aggregated yearly establishment potential of American serpentine leafminer as the number of months across the year with increasing population sizes) based on temperature and moisture constraints (left) and predicted dominant stressors based on the highest mortality rate from desiccation, cold, and heat stress across the year (right).

2.8 Risk of spread within Australia

2.8.1 Risk pathways – international and domestic

This species could potentially enter new areas by natural spread and by hitchhiking on goods, aircraft, vehicles, or through the movement of plant material (DAWR 2016).

Table 2 lists some of the potential international risk pathways and control measures in place. These are also be broadly applicable to other *Liriomyza* species.

Table 2 International risk pathways for the spread of the pest and control measures

RISK PATHWAY	DESCRIPTION	CONTROL MEASURES
Cut flowers on regulated imports	Infested cut flowers moved to residential dwellings that contain suitable hosts and the insect transfers to a host.	Border inspections
Infected plant material on passenger or mail pathways	Infested plant material moved to residential dwellings that contain suitable hosts and the insect transfers to a host.	Border inspections
Natural dispersal from Papua New Guinea (PNG)	Ongoing spread from PNG to Torres Strait and/or Far North Queensland via traditional movements, winds or flight.	Ongoing surveillance via the Northern Australia Quarantine Strategy Domestic quarantine
Infested plant material	Plant material or pupae in soil spread to new areas in baggage, vehicles or camping equipment.	Domestic quarantine

Additionally, as the larvae of *Liriomyza* spp. leave the plant to pupate, pupae may also be spread with crop debris or soil associated with infested areas. Table 3 lists some of the potential risk pathways and potential domestic control measures to reduce the spread of American serpentine leafminer. These will also be broadly applicable to other *Liriomyza* species.

Table 3. Risk pathways for the spread of the pest and potential domestic control measures

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Cut flowers (host plants)	Cut flowers that are a host of American serpentine leafminer (see Appendix 1) pose a significant risk. The life of cut flowers is long enough to allow the completion of the pest's lifecycle (CABI and EPPO, date unknown). Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Hitchhiking adults and pupae may be transported on cut flowers.	The application of systemic insecticides may be required before allowing cut flowers grown in an infested area to be moved outside the infested area. Further information on chemical control is provided in Section 5.3. 3. All cut flowers leaving infested areas will also need to be visually inspected for signs of leafminers before leaving the area.
Cut flowers (non-host plants)	Cut flowers that are not a host of American serpentine leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host cut flowers are transported with or spent time in close proximity with host cut flowers. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	To reduce the risk of spread, cut flowers that are not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with American serpentine leafminer.
Nursery Stock (containerized host plants)	Nursery stock that is a host plant for American serpentine leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Containerized nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae may also be present in the soil of containerized stock. Larvae emerge from the leaf and pupate in the soil under host plants meaning potting mix/soil can also potentially spread the pest. Larvae very occasionally attach to leaves or surrounding non-soil surfaces to pupate. Adults may potentially spread with containerized nursery stock as a hitchhiker.	Visual inspection of containerised nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae in the soil (see Figure 2, however pupae will be very hard to detect as they are mixed into the substrate) or attached to leaves and container surfaces. To reduce the risk of spread, containerised nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) should also be considered. Further information on chemical control is provided in Section 5.3.3.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Nursery Stock (bare rooted host plants)	Nursery stock that is a host plant for American serpentine leafminer (see Appendix 1) poses a significant risk for the spread of this pest. Bare rooted nursery stock can carry all life stages of the pest. Eggs may be present within leaves but are never visible. Larvae may be present and visible within leaves. Pupae are less likely to be present in bare rooted stock than containerized stock. However, upon emergence, if larvae are unable to find a substrate to bury into, they will attach to any surface available (including tray walls or plant leaves) meaning pupae can potentially still spread via bare rooted stock. Adults may potentially spread with bare rooted nursery stock as a hitchhiker.	Visual inspection of bare rooted nursery stock for the presence of adults resting on leaves, larvae creating leaf mines and pupae attached to leaves or surfaces surrounding the roots (see Figure 2). To reduce the risk of spread, bare rooted nursery stock that is a host plant for vegetable leafminer should not be moved from infested to non-infested areas without being treated with an appropriate systemic insecticide. Further information on chemical control is provided in Section 5.3.3.
Nursery Stock (non-host plants)	Nursery stock that is not a known host plant of American serpentine leafminer poses significantly less risk for the spread of this pest, but is still a potential pathway, particularly if the non-host stock spent time in close proximity with host stock. Eggs, larvae and pupae will not spread with non-host stock. Adults may potentially spread with non-host stock as a hitchhiker.	Visual inspection of nursery stock for the presence of adults resting on leaves. To reduce the risk of spread, nursery stock that is not a host plant for vegetable leafminer should be inspected and/or treated as a precaution if grown in an area known to be infested with American serpentine leafminer. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered as a precaution to limit the potential for spread.

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Non-commercially grown seedlings and plants	People and small businesses selling/trading seedlings at a local level (e.g. at farmers markets, school fetes, roadside stalls, etc.) pose similar threats to those posed by nursery stock, (i.e. plants can spread eggs, larvae, pupae and potentially adults via host plants to new areas). However, being less regulated this is a difficult pathway to control and therefore potentially poses a higher risk than commercial nursery businesses.	Should an incursion occur effort will be needed to identify those involved in the non-commercial trade of seedlings to reduce the risks that they pose. Similar to commercial nursery stock, host plants should not be moved from infested to non-infested areas without being inspected and treated with an appropriate systemic insecticide. Likewise, non-host crops grown in proximity to host plants should be inspected and/or treated as a precaution if grown in an area known to be infested with American serpentine leafminer, as there is a small risk that non-host crops could potentially spread adult leafminers as hitchhikers. Treatment of growing media using appropriate pesticides (eg Cyromazine-PER83506) may also be considered. Further information on chemical control is provided in Section 5.3.3.
Plant material including hay, plant debris and crop waste	Hay, plant debris and crop waste could potentially spread pupae and adults to new areas. Larvae are not expected to be able to survive long after plant debris and begins to wilt and rot (once it cannot feed on the plant, the larvae must vacate the leaf to begin pupation or die), however pupae will survive plant debris and crop waste. Adults may also hitchhike on hay, plant debris and crop waste.	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without being treated with an appropriate insecticide. Further information on chemical control is provided in Section 5.3.3.
Soil	Larvae emerge from the leaf and pupate in the soil under host plants (Capinera 2014). Soil therefore poses a potential risk for moving the pest to new areas.	Minimise potential spread by reducing the spread of soil off infested sites (e.g. by cleaning down machinery between properties).

RISK PATHWAY	DESCRIPTION	POTENTIAL DOMESTIC CONTROL MEASURES
Tools, equipment, and machinery	Tools, equipment, and machinery used on farm in the vicinity of host plants pose a low risk of spreading the pest, however, may transport pupae via soil deposits, or may transport adults.	Clean tools, equipment and machinery before moving off infested sites.
Transport vehicles	Vehicles in the vicinity of host plants could allow the pest to hitchhike (in cabins etc.) to new areas (DAWR 2016).	Clean vehicles before moving off infested sites.
Conveyances (includes crates, boxes, bins, pallets)	Crates, bins, pallets, etc. that are clean of soil are unlikely to spread leafminers.	As a precaution, clean conveyances before moving between sites to reduce any risk of spread.
Seed	There are no expected pathways for leafminer spread by the movement of seed.	Not applicable.
Fruit (without leaves)	There are no expected pathways for leafminer spread by the movement of fruit (without leaves). It is expected the process of harvesting and cleaning fruit will remove leaf and soil material that could spread larvae and pupae and would cause adults to disperse rather than hitchhike with fruit to new areas.	Not applicable.
Fresh leafy or green vegetables (including peas and beans)	Mines can occur on leaves of a wide range of green vegetables. There is therefore a risk that leafminer eggs or larvae (and potentially pupae and hitchhiking adults) could be spread to a new area via the movement of infested leafy vegetables. Mines do not occur on fruiting structures, with the exception that Liriomyza huidobrensis can form mines in the pods of snow peas and disrupt market access (Gitonga et al. 2010). However, it is not clear if this risk extends to Liriomyza trifolii.	Visual inspection for the presence of adults, larvae and pupae should be carried out. To reduce the potential risk of spread, plant material should not be moved from infested to non-infested areas without undertaking suitable treatments to reduce the risk of pest spread.

2.8.2 Predicting spread potential

Cesar Australia has created a model of the potential rate of area invasion by the American serpentine leafminer at entry points across Australia, the results of which are summarised below (Figure 4). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

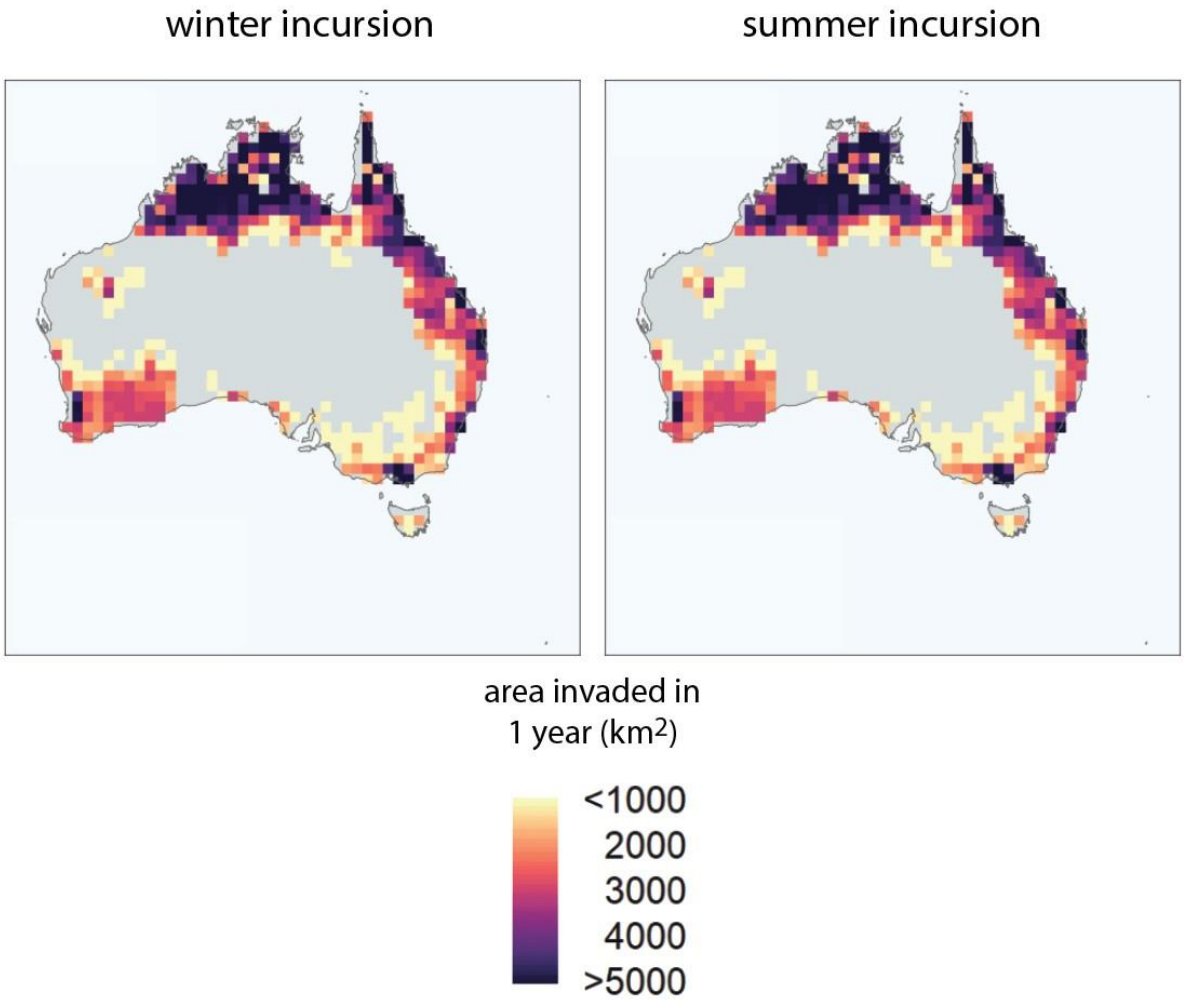


Figure 4 Rate of spread from locations across Australia. For each grid cell an incursion is simulated with the mean predicted area invaded by American serpentine leafminer in one year shown by the colour gradient. The mean predicted area is estimated from 10 replicated simulations. Incursions were commenced in July for a winter incursion and January for a summer incursion.

2.9 Risk of economic impact within Australia

Cesar Australia has created a model of the potential economic impacts of the American serpentine leafminer in Australia, the results of which are summarised below (Table 4, Table 5, and Figure 5). A copy of the relevant reports and/or published manuscripts are available from Hort Innovation or from Cesar Australia (http://cesaraustralia.com/contact-us/).

Table 4 Impact potential of American serpentine leafminer on affected commodities in terms of international collated reports of proportions crop value lost due to direct feeding damage.

Host	Proportion crop value lost			Sources
	Min	Max	Mean	
Beans	0.35	1.00	0.55	Schreiner et al 1986
Celery	0.01	1.00	0.27	Foster and Sanchez 1988
Tomatoes	0.28	1.00	0.32	Krishna Kumar 1998; Kotze et al 1996
Eggplant	0.01	1.00	0.07	Lim et al. 2007

Host	Proportion crop value lost			Sources
	Min	Max	Mean	
Watermelon	1.00	1.00	1.00	Palumbo 2020
Cotton	0.11	1.00	0.38	Nadagoudad et al. 2010
Cowpea	1.00	1.00	1.00	Jackai 1986

Table 5 Australia wide accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of American serpentine leafminer at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preferences hosts may be overestimated while impacts of high preference hosts may be underestimated. Predictions were made using ESIM framework and are the culmination of the establishment, spread and impact modules (with each level of risk building upon the previous). For impacts calculated at state levels, see Appendix 4, Table 20. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.

	Incursion location					
Crop	**Bundaberg**	**Darwin**	**Devonport**	**Melbourne**	**Perth**	**Sydney**
Beans	9.87 (0.57)	3.08 (1.39)	1.49 (0.35)	0.39 (0.09)	0.03 (0.04)	1.28 (0.92)
Broccoli	6.87 (1.46)	3.09 (1.65)	3.84 (2.94)	8.63 (0.27)	2.24 (0.46)	1.57 (1.77)
Brussels	0.79 (0.38)	0.09 (0.11)	0.54 (0.30)	0.82 (0.39)	0.00 (0.00)	0.28 (0.46)
Cabbages	4.27 (0.34)	1.88 (0.55)	1.01 (0.66)	1.19 (0.47)	0.60 (0.08)	2.63 (0.40)
Capsicum	4.20 (0.25)	0.25 (0.22)	1.63 (0.11)	0.35 (0.15)	0.68 (0.11)	0.20 (0.08)
Carrots	6.67 (0.69)	3.20 (1.30)	3.88 (0.11)	0.31 (0.05)	5.82 (1.17)	1.46 (0.67)
Cauliflowers	3.09 (0.77)	1.45 (0.84)	2.79 (1.62)	4.41 (0.15)	0.91 (0.12)	0.94 (1.00)
Flowers	21.08 (1.84)	9.65 (2.06)	9.81 (4.59)	10.67 (2.94)	2.26 (0.87)	18.49 (2.01)
Lettuces	9.03 (1.14)	3.58 (1.67)	4.09 (3.83)	10.00 (0.60)	3.27 (0.39)	2.75 (1.57)
Melons	4.62 (0.20)	1.12 (0.93)	0.01 (0.01)	0.01 (0.01)	0.80 (0.31)	0.09 (0.06)
Nurseries	48.08 (2.05)	19.13 (4.51)	12.74 (6.40)	20.38 (3.70)	8.89 (1.11)	25.65 (4.06)
Onions	3.92 (0.32)	1.63 (0.57)	3.83 (0.10)	0.20 (0.02)	1.61 (0.68)	0.66 (0.41)
Peas	0.53 (0.07)	0.09 (0.12)	0.82 (0.25)	0.21 (0.06)	0.01 (0.00)	0.07 (0.06)
Potatoes	6.69 (0.30)	1.51 (0.73)	6.56 (0.51)	1.81 (0.34)	2.41 (0.74)	1.57 (0.45)
Pumpkins	3.08 (0.17)	1.11 (0.62)	0.20 (0.04)	0.04 (0.03)	0.25 (0.11)	0.51 (0.31)
Tomatoes	13.30 (0.44)	1.38 (0.44)	1.16 (0.52)	1.28 (0.71)	0.13 (0.09)	2.26 (0.65)
T o t a l	**146.09 (6.47)**	**52.23 (13.38)**	**54.41 (19.81)**	**60.70 (8.28)**	**29.91 (5.58)**	**60.40 (11.92)**

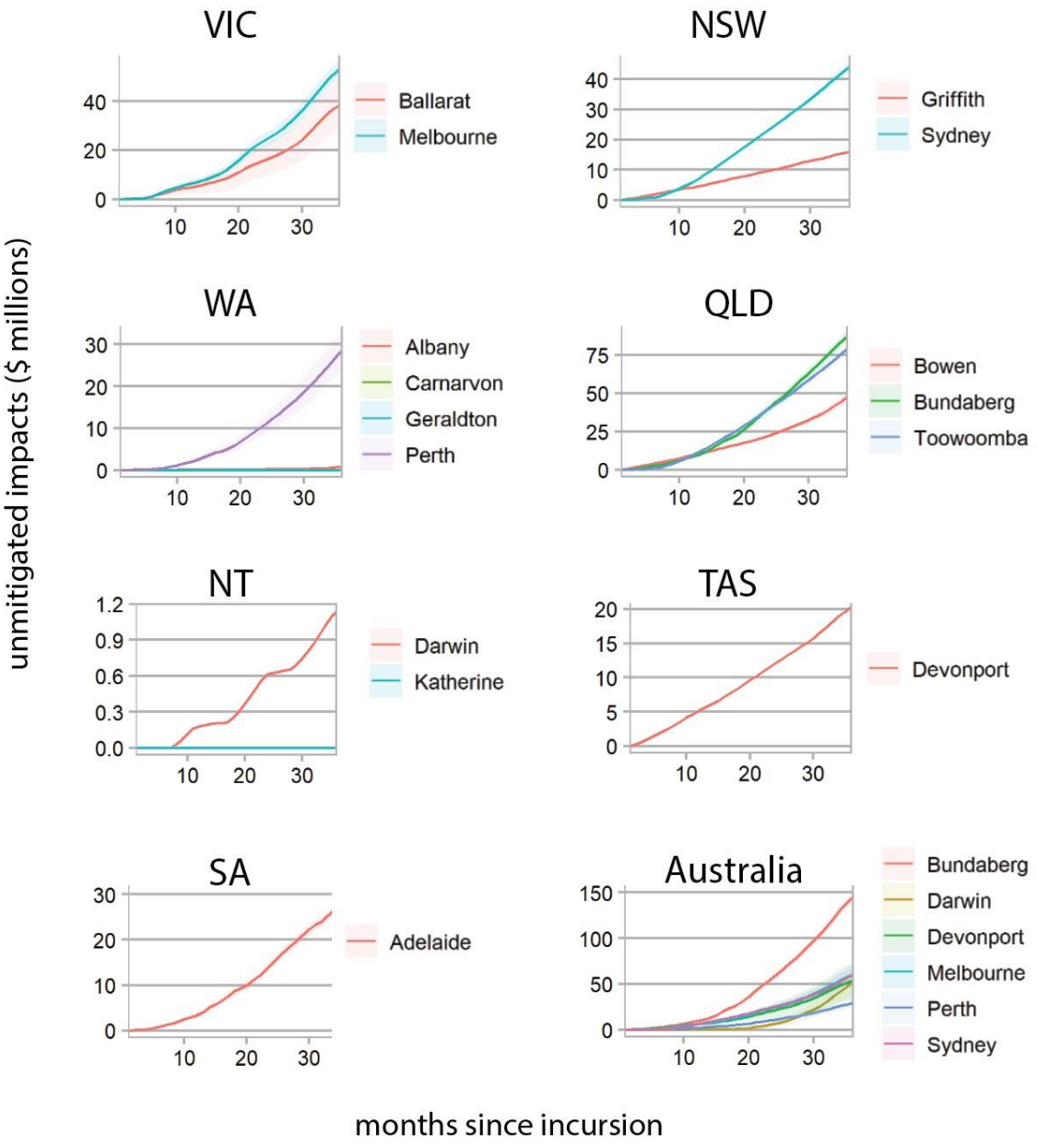


Figure 5 State level accumulating unmitigated impacts in millions of dollars across all host crops after 3 years resulting from a spring (September) incursion of American serpentine leafminer at key entry and establishment points. For state level impact accumulation by crop type, see Appendix 4 Figure 9 for additional information.

3. DIAGNOSTIC INFORMATION

Morphological diagnosis to a species level requires adult male flies. These can be collected by sweep netting, yellow sticky traps or collecting leaves with larvae in them and allowing the larvae to develop into adults. Alternatively, molecular tools can be used to identify larvae and adult flies.

Australia has a National Diagnostic Protocol covering American serpentine leafminer (SPHD 2016 – available from: http://www.plantbiosecuritydiagnostics.net.au/app/uploads/2018/11/NDP-27-American-serpentine-leaf-miner-Liriomyza-trifolii-V1.pdf). An International Plant Protection Convention (IPPC) Diagnostic Protocol (IPPC 2016) and an European and Mediterranean Plant Protection Organization (EPPO) diagnostic protocol for *Liriomyza* spp (EPPO 2005) are also available. These diagnostic protocols should be referred to for the diagnosis of suspected *Liriomyza* spp. It should be noted that these protocols do not include information on environmental DNA (eDNA). Additional information on the use of eDNA for vegetable leafminer diagnosis can be found at Pirtle et al., *in press*.

An information portal for polyphagous agromyzid leafminer identification is available at https://keys.lucidcentral.org/keys/v3/leafminers/index.htm, which includes species-specific fact sheets, and a pictorial Lucid3 key that allows users to make a preliminary identification of pest agromyzids and to be able to distinguish these species from non-pest endemic agromyzids. A factsheet for *Liriomyza sativae* is accessible here: https://keys.lucidcentral.org/keys/v3/leafminers/key/Polyphagous%20Agromyzid%20Leafminers/Media/Html/Liriomyza_trifolii.htm

3.1 Diagnostic considerations

Considerations when triaging samples of suspected American serpentine leafminer for diagnostics include:

- One of the greatest challenges in detection and subsequent management of *Liriomyza* species overseas has been the difficulty in correctly identifying the species (Reitz, Gao and Lei 2013), which can have considerable overlap in adult morphology, host range and damage symptoms left on a plant. In the presence of indigenous leafminer, it may be difficult to detect a new incursion, or determine the limits of an infestation (Powell 1981).
- There are also several difficult to distinguish Agromyzid species present within Australia that are morphologically indistinguishable from American serpentine leafminer to a non-expert (including *L. brassicae* and *L. chenopodii*). In many cases, specimens must be distinguished by expert taxonomists, or via molecular means.
- Within Australia, there at least 50 leafminer species (family Agromyzidae) (and presumably many more that have not yet been described) that create leaf mines that look similar to American serpentine leafminer and may appear on the same host plants. Some of the most common include *Liriomyza brassicae* (which shares several brassica hosts with American serpentine leafminer), *Liriomyza chenopodii* (which shares cultivated beets with American serpentine leafminer), and *Phytomyza syngenesiae (*which shares cultivated asters with American serpentine leafminer) (Appendix 2). Moreover, there are an unknown number of additional Dipteran and Lepidopteran species that share the leaf mining habit.
- Of these species that create similar leaf mining damage, some can be distinguished from American serpentine leafminer via their mine characteristics, and their pupal or adult forms. (see Appendix 2, IPCC 2016). However, the high degree of individual variation between each leaf mine means there can always be exceptions to these guidelines. For example, while American serpentine leafminer pupate outside the leaf, there are rare instances where a larvae will fail to emerge and complete pupation within the leaf mine.

- Due to the high degree of morphological overlap of adults and visual overlap of damage, sample collection (of flies of any life stage and/or leaf mines) will be paramount for identifying and delimiting potential American serpentine leafminer incursions.
- Surveyors should be familiar with the American serpentine leafminer host plant list (Appendix 1), as well as with commonly observed signs of indigenous leafminer damage and the extent of known overlap with indigenous Agromyzids for common host genera (Appendix 2) before conducting American serpentine leafminer surveillance. For example, damage caused by the closely related *L. brassicae* is essentially indistinguishable from American serpentine leafminer and is very common on brassica crops (Appendix 2). Crops such as tomato, cucumbers and melons, on the other hand, have little to no reported overlap with indigenous Agromyzids (Appendix 2). Mines detected on these crops during surveillance activities should be considered highly suspicious.

4. SURVEILLANCE AND COLLECTION OF SAMPLES

4.1 Surveillance

4.1.1 Technical information for planning surveys

Detection and delimiting surveys are required to determine the extent of outbreaks, ensure areas free of the pest retain market access, and ensure appropriate quarantine zones are established to contain outbreaks.

Initial surveillance priorities for American serpentine leafminer should consider the following:

- Visual surveillance for the leaf mines created by American serpentine leafminer is expected to be the most reliable way to detect new incursions. This is because the serpentine leaf mines created by leafminer larvae persist permanently on a leaf, extending the detection window beyond the lifespan of the fly itself. In comparison, sweep netting requires active populations, as do yellow sticky traps. While sticky traps can be left in place for extended periods to increase the chance of catching an adult, in open field settings, relying solely on sticky traps would be impractical due to the very large number of traps that would be needed to be placed and processed, to reach a statistically acceptable detection likelihood for small or isolated populations.
- In closed cropping systems, sticky traps become much more practical, but should still be accompanied by visual surveillance.
- Sticky traps will have a high level of bycatch (i.e. other insects will be caught in the trap), which can increase processing time for manual identifications. However, molecular diagnostic methods are also available for bulk invertebrate samples collected via a sticky trap (see Section 4.2.2.2).
- When undertaking visual surveillance in plant crops, the leaf mines are the most conspicuous symptoms to look for, though stippling from oviposition and feeding may also be apparent.
- Mines created by *Liriomyza* spp. are generally expected to be more common on the edges of fields and in the upper portions of the plant (Weintraub 2001).
- The American serpentine leafminer has an extensive list of weed hosts (see Section 2.3) and therefore surveillance in broadleaf weeds as well as cultivated crops should be undertaken.
- Larvae, pupae and adults can be identified using molecular methods (refer to Section 3).
- Only adult male leafminers can be identified using morphological methods.
    - Adults can be captured using yellow sticky traps, sweep netting (IPPC 2016), or rearing from infested material. The latter is the most practical and reliable method for collecting adult flies. Infested leaves can be placed into an insectarium to develop into adults (IPPC 2016). Techniques for rearing Agromyzids are described in Griffiths (1962) and Fisher et al. (2005).
- Collectors should be made aware of common signs of leafminers and note symptomatic plants when conducting surveillance for American serpentine leafminer.

- Australia is already home to several leafminer species, both native and naturalized, which can be confused for American serpentine leafminer. Surveyors should be made aware of the native and naturalized species they might expect to see in the particular region and crops they are surveying.
- Early detection surveillance will be most effective if timed when American serpentine leafminer population growth potential is greatest, such as during warm conditions. The seasonality of American serpentine leafminer activity at major centers is provided in Table 6.

Table 6 Major Australian growing regions showing risk seasonality for American serpentine leafminer activity across the year. Green means highest establishment risk (where the population growth potential was greater than 50% of the maximum predicted growth rate) yellow means lower establishment risk (where the population growth potential was between 1 and 50% of the maximum), and red means lowest establishment risk (where predicted growth rates are negative, meaning populations are predicted to decline).

State	Incursion point	Seasonality
		Jan Feb Mar Apr May Jun Jul Aug Sep Oct Nov Dec
Western Australia	Perth	
	Geraldton	
	Carnarvon	
	Bunbury	
	Albany	
Victoria	Thorpdale	
	Melbourne	
	Ballarat	
Tasmania	Smithton	
	Devonport	
South Australia	Mt. Gambier	
	Loxton	
	Adelaide	
Queensland	Toowoomba	
	Bundaberg	
	Bowen	
	Atherton	
Northern Territory	Katherine	
	Darwin	
New South Wales	Sydney	
	Griffith	

4.1.1.1 Surveillance tools

Yellow sticky traps and sweep netting can be used as supplementary methods to visual searches

during delimiting surveillance activities, particularly when suspicious damage has been observed, or for monitoring existing outbreaks.

- Sweep netting is expected to be most effective when employed near recently active leaf mines, and is less effective near old leaf mines (which tend to be darker coloured than fresh mines, see Figure 6). Windy conditions can also reduce the effectiveness of sweep netting as adults tend to remain low in foliage during windy conditions (Foster 1986).



Figure 6 Younger leafminer mines (on the left) are more white-coloured with more distinct black trails of frass. Frass becomes less distinct as mines age, and mines develop a brown colour (right). Younger mines are expected to give the best sweep netting success. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

- Agromyzid flies are attracted to the colour yellow (Chandler 1981), and can therefore be captured on yellow sticky traps. Yellow sticky traps are more effective for *Liriomyza* adults than other types of traps, such as funnel traps and yellow water pans (Chavez and Raman 1987). Vacuum sampling has also been shown to be a less effective means of sampling adult leafminer (Weintraub 2001).
- A great deal of effort overseas has been dedicated to improving the effectiveness of yellow sticky traps for *Liriomyza* adults (Heinz et al 1992) including the modifications of size, shape, adhesives, lures, height and orientation. For example, several studies report a strong effect of trap height on the number and species trapped (Trumble and Nakakihara 1983, Zehnder and Trumble 1984, Zoebisch and Schuster 1990), however these results do not always appear consistent and may be difficult to extrapolate across different crop types. Moreover, optimal height may vary considerably between *Liriomyza* species (Zehnder and Trumble 1984). General guidelines are:
- Some considerations when using yellow sticky traps for *Liriomyza* surveillance are provided below:
    - Traps should be placed (vertically oriented) at the edges of paddocks, preferably on the upwind side of paddocks, and near weed reservoirs if possible.
    - Optimal trap height may vary with crop type.
    - Traps should be placed in the afternoon, and left for a minimum of 24 hours, to ensure traps are active during the peak flight time for American serpentine leafminer
    - Traps may be left in place for up to one month, at which point they should be collected, to prevent excessive degradation of DNA and allow for molecular confirmation of American Serpentine leafminer caught on the trap. If traps are placed outdoors and exposed to the elements and/or high levels of bycatch they should be collected sooner than one month.

Experimental lures developed from the extracted volatiles of known plant hosts have been shown to be attractive to *Liriomyza*. However, none have yet been identified for American serpentine

leafminer.

4.1.2 Delimiting surveys in the event of an incursion into new areas

Following a new detection of American serpentine leafminer, the following guidelines are recommended for delimiting surveillance:

- Surveillance to delimit a detection of American serpentine leafminer should take into account tracing information as outlined in Section 5.1 to determine potential pathways for the movement of material from the site of the initial detection
- Surveillance should be a combination of:
    - Visual inspection in high risk areas (e.g. areas with suitable hosts)
    - Sweep nets to collect adults around infested host plants
    - Yellow sticky traps, particularly in closed cropping systems and around infested plants/areas
- Surveillance should be accompanied with awareness material, signs and personal visits to households and businesses within the surveillance zone and any buffer zones

The suggested surveillance procedure for American serpentine leafminer is as follows:

1. Delimiting surveillance should be conducted immediately upon detection of any incursions within a radius within 1 km of the incursion site, based on natural spread dynamics of agromyzid leafminer. The Surveillance zone may be increased as deemed necessary for the individual response
2. At each site, chose a crop that is a known host plant of American serpentine leafminer (refer to Section 2.3)
3. Choose a survey path that preferences the following areas if possible:
    - Paddocks and weeds near transport routes and unloading areas. These are preferred, because a major long-distance pathway of leafminer spread is human assisted movement
    - Paddock edges should be included in the survey path, with the 'incoming wind side' preferred, since short distance movements of leafminer can be driven by wind.
    - Weeds along paddocks should be included in the survey path.
4. Leafminer damage may be patchy at low densities.
5. At low densities, leaf mines have a relatively low detectability, despite their characteristic appearance. Slower search speeds will improve detection likelihood, however, when time is limited, search speed trades off with the total area covered (reducing the number of possible encounters). This trade-off is important in realistic paddock monitoring scenarios.
    - When time is not limited: To achieve a detection probability of 20%, 50%, and 80% respectively, when leaf mine density is about 1% of plants affected, the mean (and 95% CI) search time required per metre is 11.2 (4.8, 20.5), 34.8 (15.0, 63.8), and 80.8 (34.9, 148.2) seconds spend 1 minute searching per 1 m^2 of area (based on data collected during vegetable leafminer survey effort trials).

- When time is limited: The optimal search speed, balancing area coverage with sensitivity, when leaf mine density is about 1% of plants, is about 10 seconds per 1 m^2 of area. Further benefits are not achieved by increasing search speed beyond this level to achieve greater area coverage.
- (These recommendations are based on data collected during Vegetable leafminer (*L. sativae*) survey effort trials – other *Liriomyza* leafminers are expected to require similar effort)).

6. A survey plan should take into consideration any variables regarding the environment or surveyor that might reduce detectability and increase the effort spent accordingly. These variables might include:
    - *Plant type*
        i. *Leaf density:* leafminer damage may be harder to spot in plants that are very dense and leafy, than in plants with larger, sparser or flatter leaves.
        ii. *Plant age:* it may be easier to spot leaf mines in young plants with few leaves and more open space between plants than in larger mature, overlapping plants.
        iii. *Colour:* leafminer damage may be harder to spot against light coloured leaves than against darker leaves.
        iv. *Height*: Leafminer damage in plants that are at eye level may be easier to spot than crops that are low to the ground.
        v. *Time of day/direction of survey relative to the sun*: Survey success may be higher when surveyors are walking with their back to the sun, rather than walking into the sun.
    - *Residues on plants:* Chemical residue on plants may reduce the contrast between the mine and the leaf, or obscure mine patterns with background residue patterns.
    - *Damage from other pests/disease on plants:* damage caused by other pests or disease may obscure leaf mines, or lead to false positive detections.
    - *Surveyor experience:* Inexperienced surveyors may have a learning curve.
    - *Weather conditions:* Surveys during poor weather should be avoided when possible.
7. If suspicious damage is detected, samples should be collected (see Section 4.2), and yellow sticky traps should be placed around the affected area, in an attempt to capture adults and diagnose the fly responsible for the damage.
    a. If American serpentine leafminer are confirmed, visual surveillance along with sticky traps should be used to monitor the edges of the outbreak area.
8. If no suspicious damage is detected, the initial round of delimiting surveillance should be followed with a second round occurring after one week.
    a. This second round is necessary as leafminer eggs are undetectable in the leaf, and hatch after about 2-5 days. A crop that appears clean during the initial survey may still develop symptoms within a week.
    b. After the first revisit, any further revisits should occur on a monthly basis. One lifecycle of the American serpentine leafminer is estimated to take approximately 2 to 4 weeks, dependent on host and temperature.

4.1.3 Activities for general surveillance immediately following a detection

Given the wide host range of this pest and its noticeable impact it may be a suitable target for general surveillance programs that request submission of images and/or samples from the public.

To establish a general surveillance program, the following will be required:

- Use of online or app reporting tools such as MyPestGuide to promote the submission of reports of suspected detections.
- Factsheets to provide information on the pest, symptoms, impacts and reporting mechanisms.
- Media releases to describe the impact of the pest, surveillance programs and activities within the response program.
- Information for industry communication channels including articles for industry newsletters, magazines and websites, information for Twitter feeds and Facebook, and presentations for industry talks.
- A website to provide information for the public and for commercial businesses and links to other relevant sites.
- Release and promotion of information on details for physical sample collection and submission, as well as information on how to take and submit images of flies/leaf mines.
- Broader awareness campaigns should consider literature (brochures and factsheets) in several languages.
- A non-expert friendly surveillance and sample collection guide for vegetable leafminer, which also has relevance for the pea leafminer, as well as other useful information, can be found here.

4.2 Collection and treatment of samples

4.2.1 If collecting for morphological diagnosis

Of the four life stages (egg, larva, pupae and adult) only adult males are identifiable to a species level using morphological features.

4.2.1.1 Collection of adult male specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the foliage. However, the most practical and reliable method is the collection of leaves with mines containing pupae or mature larvae which can then be reared until adult flies emerge (refer to Section 4.2.1.3).

4.2.1.2 How to collect sticky traps for identification

Sticky traps should be left in the field for up to one month (but preferably 2 weeks or less), to allow morphological identifications. Upon collection, sticky traps should be folded lightly in half sticky side inwards (not pressed shut), and placed in a sealed, padded plastic bag. Traps should be sent within a week of collection (and kept in cool, dry place in the meantime, but not frozen).

4.2.1.3 How to collect plant samples for rearing

For rearing of adult flies, mined leaves containing pupae or mature larvae can be collected into an insectarium and kept in a constant temperature room for regular checking. Simple insectariums include sealed partially inflated ziplock bags, or sealed rigid plastic containers or jars. A piece of paper towel should be placed into the insectarium along with the infested leaves to regulate humidity. Once pupae emerge from the leaves, they should be transferred carefully with a wet paintbrush to a new rigid container, which contains a layer of damp sand but no leaf material. Pupae can be placed directly onto the damp sand. Techniques for rearing agromyzids are described in the IPCC diagnostic protocol for *Liriomyza* (IPPC 2016, available from here).

4.2.1.4 How to preserve plant samples for identification

Identifying the plant species affected by suspected American serpentine leafminer damage can aid diagnoses, particularly when surveying in weeds. To preserve a plant for identification, leaves can be stored between sheets of dry newspaper. However, if plant material with suspected leaf mines is pressed and dried, there will be no chance of rearing out adult flies from those leaf mines. Therefore, whenever plant identity is in question, infested material should be stored in an insectarium for rearing, and an accompanying pressed sample of uninfested leaves should be taken to facilitate identification of the host.

4.2.1.5 How to preserve leafminers

Adults caught in sweep nets or reared in insectariums can be placed in 70% ethanol and stored indefinitely for morphological purposes. For adults reared in insectariums, it is advised to collect these adults in the afternoon, as emergences tend to occur in the morning, and enough time should be given for wings to fully expand before collection.

4.2.1.6 How to transport leafminers

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.1.7 How to transport plant samples

Plant samples may be collected if the host needs to be identified. Queensland Herbarium (2016) provides information on how to collect and preserve herbarium specimens. Samples should then be taken to state herbaria for identification.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

4.2.2 If collecting for molecular diagnosis

All life stages can potentially be diagnosed using molecular methods, including empty leaf mines.

4.2.2.1 Collection of specimens

Adult flies can be collected with a vacuum sampler or sweep net. Adult flies are normally found on the foliage. However, as other life stages can be diagnosed using molecular methods leaves with mines containing pupae or mature larvae should also be collected for analysis. Rearing these life stages in an insectarium is not necessary for molecular diagnostics.

4.2.2.2 How to collect sticky traps

Upon collection, sticky traps should be either:

- Placed inside a plastic sleeve that has been cut along one long side and along the bottom, the trap and sleeve can then be sealed in a plastic zip lock bag.
- Folded in half, sticky side inwards, and placed in a sealed plastic bag.

Traps should be sent within a week of collection (and kept in a cool, dry place in the meantime, but never frozen).

4.2.2.3 How to collect plant samples

Plant samples with suspected American serpentine leafminer damage should be collected for molecular confirmation. If it is possible to discern a live larvae inside a leaf mine (which can be spotted through the leaf mine as a small yellow maggot, either by eye or with a hand lens, see Figure 7), these samples are always preferable. However, mined leaves that appear empty can also be collected. Empty mine samples that look youngest should be preferred, as they have the greatest chance of containing a live larva (see Figure 6).



Figure 7 A larva inside a leaf mine. Larvae are visible to the eye in later stages of development. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

4.2.2.4 How to preserve plant samples

Mined leaves containing pupae or mature larvae, or mined leaves that appear empty can all be placed straight into 100% Ethanol. Small mined leaves should be rolled and placed into the tube of absolute ethanol. For larger leaves, the mine should be cut out, with care taken to avoid cutting across the mine, and the cut out should be rolled and placed into the tube of absolute ethanol. These samples should be frozen and mailed within a week of collection for diagnostics.

Due to the very small amounts of DNA within leaf mines, the odds of sample contamination is much higher than when working with whole flies. Careful hygiene must be employed when collecting and preserving suspicious leaf mine samples. Mines from different host plants or different locations should

never be stored in the same container. Samples should either be immediately preserved into on site, or should be placed into unused, clean, sealed plastic bags on site and preserved within the day. If samples from multiple hosts or sites are being processed together at a field lab location, all equipment must be sterilised between each host/location (i.e. cleaning counter tops, sterilizing tweezers and other tools).

Leaf mines may also be preserved onto FTA cards, however DNA quality is somewhat lower than for preservation into 100% ethanol (Pirtle et al., *in press*). To preserve leaf mines onto FTA cards, remove the mined leaf from plant and push the leaf mine face down onto the FTA card. Smear the mine thoroughly on the FTA card, for about 30 seconds, and then close the card and place in small sealed plastic bag. Do not rub mines from different host plants/sites onto the same FTA card, and do not store FTA cards from different host plants/sites in the same sealed bag. Store the FTA cards in a cool, dark, dry place and send for analysis within a week of collection

4.2.2.5 How to preserve leafminers

Specimens required for molecular diagnostic work, including larvae, pupae or adults, should be killed and preserved in absolute ethanol, frozen at -80°C , or preserved on an FTA card.

4.2.2.6 How to transport leafminers and plant samples

Vials of ethanol should be sealed to avoid leakage and packed with cushioning material in a strong box.

For information on what can or cannot be sent by post refer to the Australia Post website: https://auspost.com.au/content/dam/auspost_corp/media/documents/dangerous-and-prohibited-goods-guide.pdf.

5. IMMEDIATE RESPONSE TO DETECTION

5.1 Tracing

Detection and delimiting surveys are required to delimit the extent of the outbreak, ensure areas free of the pest retain market access, and ensure that appropriate quarantine zones are established.

Extensive tracing (trace forward and trace back) may be feasible as *Liriomyza* spp. can be readily dispersed by the movement of infested plant material, soil and crop debris. The focus should be on high risk linkages including premises linked directly with the initial detection, particularly where movements of plant material (such as nursery plants and seedlings), or soil have occurred as these pathways may move American serpentine leafminers over long distances.

Further information on possible risk pathways are presented in Section 2.8 and in Table 3.

5.2 Quarantine and movement controls

If Restricted or Quarantine Areas are practical, no plant material should be moved from the infested to uninfested areas without first being inspected and appropriately treated. The size of the Restricted or Quarantine Area will be dependent on the type and scale of the incursion. However, a zone 1 km in radius should be considered for initial delimiting surveillance activities (Section 4.3). This zone can then be expanded as needed depending on the specific situation.

Voluntary movement control should be considered for urban/residential detections. Voluntary controls would involve negotiation with residents to undertake inspection and treatment of goods prior to movement from infested areas. Residents should be advised on measures to minimise the inadvertent transport of the pest from the infested area to unaffected areas. Voluntary compliance is likely to be implemented for urban areas using awareness campaigns to highlight high risk goods/situations and appropriate treatments.

5.3 Destruction strategy

5.3.1 Priorities and considerations

If eradication of an outbreak is considered feasible, the priority will involve the destruction of the pest. Adult leafminers may be present around host plants; eggs, larvae and pupae may be present in the leaves of host plants, and pupae may be present within the first 5 cm of soil under host plants.

Considerations include:

- The infested plants may need to be sprayed with a suitable insecticide (refer to Section 5.3.3), then bagged and then buried (or tilled/ploughed directly into the soil if a large area is affected).
- Other host plants in the immediate area should be inspected and treated as needed.
- Soil and plant media in greenhouses should be treated (heat or chemically) to destroy any pupae that may be present in the soil.

- Machinery and equipment used on site should be washed down to minimise the potential risk of spreading the pest between sites.

5.3.2 Decontamination of tools and equipment

Machinery, equipment, tools and clothes should be cleaned and disinfected between sites to minimise the potential to spread leafminers, and other pests and diseases that may be present on the site, to new areas.

For points to consider when decontaminating, refer to the PLANTPLAN Guideline document: Disinfection and Decontamination (PHA, 2015[1]). which includes guidelines for the destruction and decontamination of tools used when working with emergency plant pests.

Several state agriculture departments have also produced publications on cleaning down machinery and vehicles and should be referred to if decontamination is required, examples include:

- www.dpi.nsw.gov.au/ data/assets/pdf_file/0010/545554/procedure-decontamination-vehicles-and-equipment.pdf.
- www.daf.qld.gov.au/ data/assets/pdf_file/0011/58178/cleandown-procedures.pdf
- http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/weeds/weedstop-vehicle-hygiene-program/machinery-hygiene.

5.3.3 Chemical control

Overseas *Liriomyza* leafminers are most damaging when parasitoids are removed due to the use of non-selective insecticides (Cesar Australia 2018; Murphy and LaSalle 1999). For the ongoing management of leafminers it will therefore be important that chemicals are selected that are reasonably selective and have low toxicity towards key parasitoids.

The European and Mediterranean Plant Protection Organization (EPPO) provides guidance (EPPO 2009) on how to manage and eradicate Vegetable leafminer infestations in Europe, which are broadly applicable to other *Liriomyza* spp., such as the American serpentine leafminer. For Vegetable leafminer, the EPPO recommends that foliar sprays are applied at weekly intervals until control has been achieved. Importantly the EPPO suggests rotating chemicals and modes of action (where possible) each treatment (Table 7). In greenhouse environments the use of a suitable space treatment is also suggested (EPPO 2009). Based on this it would be ideal if pesticides from different mode of action groups are available for each host crop, as this would allow pesticides to be rotated rather than relying on a single product being continuously used to manage/treat the pest.

[1] www.planthealthaustralia.com.au/biosecurity/incursion-management/plantplan/

Table 7 An ideal chemical rotation schedule used for eradication of Liriomyza leafminers in Europe (refer to EPPO 2009)

WEEK	PESTICIDE USED TO TREAT INFESTED AREA
Week 1	Chemical A
Week 2	Chemical B
Week 3	Chemical C
Week 4 onwards	Repeat above sequence

Pesticides registered for the control of American serpentine leafminer overseas include the following actives:

- Abamectin
- Azadirachtin
- Chlorantraniliprole
- Cyantraniliprole
- Cyromazine
- Diazinon
- Emamectin benzoate
- Indoxacarb
- Permethrin
- Spinetoram
- Spinosad
- Spirotetramat

It should be noted that Australian Pesticides and Veterinary Medicines Authority (APVMA) permits will need to be put in place before these pesticides can be used for the control of American serpentine leafminers. Permits are being applied for as part of Horticulture Innovation Australia Project MT16004 (RD&E program for control, eradication and preparedness for Vegetable leafminer), and these are noted in Table 8.

As the American serpentine leafminer can affect many plant species (including agricultural and ornamental species) (refer to Section 2.3), it is important that all potential hosts are covered by at least one, but preferably multiple, pesticide permits. Appendix 3 provides a summary of the pesticides that can be used on each crop type and host and should be used as a guide when treating new American serpentine leafminer outbreaks, or if eradication is unsuccessful, managing the pest. As new permits are approved by the APVMA, they will be made publicly available on the APVMA permit database: https://portal.apvma.gov.au/permits.

Table 8 Insecticide permits that have been applied for or granted by the APVMA for the control of American serpentine leafminer in Australia (as of July 2020)

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[2]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Cyromazine (MOA 17)	Contact and translaminar activity	Not effective against adults but being an insect growth regulator is very effective against larval stages (Cesar Australia 2018b). Experiments on the American serpentine leafminer found that Cyromazine was very effective against first and second instar but less effective against late instar larvae (Yathom et al., 1986).	Nursery stock (non-food)	333 g in 20 L water/10 tonne potting mix 950 g/ha or 95 g/100 L applied in 1,000 L/ha.	PER83506	31 October 2022
			Fruit trees (non-bearing)			
Dimethoate (MOA 1B)	Contact and systemic activity	Dimethoate is reported to control *Liriomyza* spp. adults and larvae however resistance is known overseas (Tran and Takagi 2005)	Pulse crop (grains)	75ml (400 g a.i/L products)/100L water	PER89184	31/3/2025
Emamectin benzoate (MOA 6)	Contact and translaminar activity	Same mode of action as abamectin. Reported efficacy against larvae, adults and eggs (Cesar Australia 2018b)	Brassica vegetables (including broccoli, Brussels sprouts, cabbage, cauliflower)	250 to 300 g/ha (44 g a.i/kg products); or 650 to 780 mL/h (17 g a.i/L products) (suppression only)	PER87563	30 June 2024

[2] Refer to Appendix 4 for detailed information on chemical control options.

CHEMICAL	CHEMICAL ACTIVITY	LIFECYCLE STAGES CONTROLLED	CROPS COVERED[2]	PROPOSED RATE	PERMIT NUMBER	EXPIRATION DATE
Spinetoram (MOA 5)	Contact, systemic and translaminar activity	Spinetoram has shown efficacy against *L. sativae* adults and larvae (Shimokawatoko et al., 2012)	Vegetable legumes (peas and beans)	400 ml (48 g a.i)/ha	PER87878	28/2/2023
Spirotetramat (MOA 23)	Contact, systemic and translaminar activity	Affects the development of larvae and has little impact on adults (Ring 2019)	Capsicum	400ml (96 g a.i)/ha	PER88640	31/5/2023
			Celery			
			Chili			
			Eggplant			
			Green beans			
			Lettuce (head and leafy)			
			Parsley			
			Rhubarb			
			Snow & sugar snap peas			
			Tomato			

Research undertaken as part of the Hort Innovation project MT16004 has identified the suitability of the following as potential control options for *Liriomyza* spp.:

- Azadirachtin (a systemic insecticide that acts as an insect growth regulator, MOA unknown) on ornamental and floriculture plants.
- Abamectin (an insecticide with contact and translaminar activity, MOA 6) on fruiting vegetables, cucurbits, leafy vegetables, legume vegetables, bulb vegetables, cabbage, celery, and rhubarb.
- Cyantraniliprole (a systemic insecticide, MOA 28) on bulb vegetables, fruiting vegetables, cucurbits, melons, and potatoes (note residue data will be required to support this use pattern).
- Diazinon (a contact insecticide, MOA 1B) on beans, melons, beetroot, lettuce, parsnips, tomatoes, and potatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Dimethoate (a systemic insecticide, MOA 1B) on green beans, peas, tomatoes, capsicums, and potatoes. Note can have significant impact on beneficial insects and not fit well with IPM systems.
- Emamectin Benzoate (an insecticide with contact and translaminar activity, MOA 6) on brassicas, leafy vegetables, fruiting vegetables, cucurbits, and melons.

- Indoxocarb (an insecticide with contact and some translaminar activity, MOA 22A) on tomatoes.
- Permethrin (an insecticide with contact activity, MOA 3A) on tomatoes. Note can have significant impact on beneficial insects meaning it would likely be more suited to use in eradication programs than ongoing management of the pest.
- Pyrethrins (an insecticide with contact activity but very limited residual activity, MOA 3A) on brassicas, leafy vegetables, lettuce, fruiting vegetables, and legume vegetables.
- Spinetoram (an insecticide with contact, systemic and translaminar activity, MOA 5) on cotton.

Permits for these use patterns have not been applied to date. However, in the event of an incursion, emergency permits for these use patterns could be developed if required.

5.3.4 Physical control options

Destruction of host plants and deep ploughing of crop residues can assist with leafminer control as adult leafminers have difficulty emerging from pupae buried deeply in soil.

Removal of weed hosts of the leafminer and planting susceptible crops away from weed hosts can also assist with the ongoing management of the pest by removing potential leafminer reservoirs.

5.3.5 Biological control

Although not specifically for eradication, biological controls will assist in lowering leafminer populations and will be an important part of the management of American serpentine leafminers should eradication not be possible in the event of an outbreak.

Parasitoids often provide effective suppression of leafminers in the field when non-selective insecticides are not used (Cesar Australia 2018). History has also shown that invading *Liriomyza* populations are rapidly exploited by endemic Agromyzid parasitoids (Cesar Australia 2018). For these reasons it will be important that any pesticides used for the ongoing management of leafminers are compatible with Integrated Pest Management (IPM) systems.

Minkenberg and Lenteren (1986) identified 28 species of parasitoids that are reported to affect American serpentine leafminer (Table 9). Agromyzid parasitoids tend not to be very host-specific and Australia already has several endemic species that would likely be effective against the American serpentine leafminer. *Diglyphus isaea* is an example of a parasitoid that occurs in Australia and is mass reared overseas for the biological control of *Liriomyza* species including American serpentine leafminers.

The importation of biological control agents is not warranted at this time as the adventive parasitoids such as *D. isaea*, already occur in Australia and would likely be suitable for the management of leafminers assuming pesticides are selected appropriately and are compatible with IPM systems.

Table 9 Parasites and parasitoids of American serpentine leafminer noted in Minkenberg and Lenteren (1986)

PARASITE NAME	PARASITE NAME	PARASITE NAME	PARASITE NAME
Chorebus misella	*Diglyphus intermedus*	*Halticoptera circulus*	*Diglyphus crassinervis*
Dacnusa maculipes	*Diglyphus pulchripes*	*Halticoptera patellana*	*Ratzeburgiola incompleta*
Oenonogastra microrhopalae	*Diglyphus websteri*	*Diaulinopsis callichroma*	*Hemiptarsenus dropion*
Opius dimidiatus	*Chrysocharis parksi*	*Chrysocharis caribea*	*Pnigalio soemius*
Opius dissitus	*Chrysonotomyia formosa*	*Closterocerus purpureus*	*Chrysocharis pentheus*
Mirzagrammosoma lineaticeps	*Chrysonotomyia punctiventris*	*Hemiptarsenus semialbiclava*	*Dacnusa sibirica*
Diglyphus begini	*Closterocerus cinctipennis*	*Diglyphus isaea*	*Halticoptera crius*

5.4 Decision support for eradication

A summary of key activities associated with different detection scenarios for American serpentine leafminer is provided in Table 10. Differing physical detection scenarios have been chosen to highlight operational differences in urban compared with agricultural and horticultural environments.

A summary of factors to be considered for eradication or alternative action is provided in Table 11.

Table 10 Recommended responses and considerations for differing scenarios of detection of American serpentine leafminer

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Commercial glasshouse/ protected cropping setting	➢ Destruction of hosts within glasshouses could be considered to limit food sources or refuges (see Section 5.3.4). ➢ Chemical sprays to eradicate larvae and adults (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and in glasshouses managed by the same business as the infested glasshouse (use visual inspections, sweep netting and yellow sticky traps for	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around the Infected Premises (IP). ➢ Focus on known hosts. ➢ Surveillance at linked properties. ➢ See Section 4 for points to consider when establishing a surveillance program. Key points for glasshouses are: • Undertake visual surveillance of all host plants. • Consider using visual surveys, sweep nets and yellow sticky traps.
Open agricultural/ horticultural setting	➢ Spray infested area using appropriate insecticide (see Section 5.3.3). ➢ Surveillance within 1 km radius of the detection and survey jointly managed properties (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ No movement of equipment or produce without permit. ➢ Permits to include information on inspection, decontamination or destruction requirements.	➢ Minimum 1 km surveillance zone around each IP. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance awareness campaigns may be beneficial. ➢ Surveillance at linked properties should occur.

DETECTION SCENARIO	RECOMMENDED ERADICATION/ CONTROL TREATMENTS – SEE SECTION 5.3	RECOMMENDED MOVEMENT CONTROLS – SEE SECTION 5.2	RECOMMENDED SURVEILLANCE OPTIONS – SEE SECTION 4
Urban environment	➢ Chemical sprays to eradicate larvae and adults. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls in infested areas could be undertaken by permit or as voluntary movement controls (coupled with awareness campaign for urban areas).	➢ Surveillance on host plants within 1 km around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Surveillance to be undertaken in conjunction with awareness campaigns for households providing information on the pest and symptoms. ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ Surveillance at linked properties.
Open natural environment (e.g. detection on roadsides or national park)	➢ Treatment of detections in a 1km zone pending delimiting surveillance. ➢ Surveillance within 1 km radius of the detection (use visual inspections, sweep netting and yellow sticky traps for surveillance).	➢ Movement controls in infested areas could be undertaken by permit or as voluntary movement controls. ➢ Movement restrictions may be considered pending outcomes of delimiting surveillance.	➢ Surveillance on host plants within 1 km around IP (noting leaf mines, feeding and oviposition punctures are the most easily seen plant symptoms). ➢ Yellow sticky traps, visual inspections and sweep netting should be undertaken in the 1 km zone. ➢ General surveillance and awareness campaigns should be considered. ➢ Surveillance should be

Table 11 Summary of factors to be considered in determining whether eradication or alternative action will be taken for an incursion of a American serpentine leafminer

FACTORS TO CONSIDER REGARDING THE TECHNICAL FEASIBILITY OF ERADICATION
• **The population size and population structure associated with the initial detection.** • **The cost effectiveness of recommended control technique options.** - Multiple applications of several different pesticides over a period of time (weeks/months) may be required to successfully eradicate American serpentine leafminer populations. • **The ability to remove or destroy all American serpentine leafminers by the recommended control techniques.** - Permission to enter private premises for surveillance and treatment must be considered. - Determination of whether treatments can effectively eradicate populations within premises or environments. • **The recommended control techniques are publicly acceptable.** - Chemical treatments in residences or backyards may not be acceptable or will require negotiation with residents. • **Whether emergency containment measures can effectively be put in place to contain the outbreak.** • **Whether there are control methods, commonly employed for endemic pests, that may prevent the establishment of, or be impacted by the treatment, for American serpentine leafminer.** • **Legislative impediments to undertaking an eradication response.** - If not a prescribed pest, confirmatory diagnosis will be required under legislation in some jurisdictions before powers to enter premises and undertake treatments can be enacted. - Delimiting surveillance in urban environments may be restricted to front yards and environs rather than entry into premises (unless permission from the resident can be negotiated). - Emergency permits will be required for chemical treatments. Amendments to chemical labels will be required for longer term management of the pest if eradication is not possible. • **The ability to delimit the known area of infestation.** - Determination of linked properties in an urban detection may be unfeasible given the ability of adult leafminers to disperse at a local level. High risk linkages such as the recent purchase of seedlings, cut flowers or other commodities that could vector the pest should be investigated. • **The ability to identify and close the pathway for entry of the pest into Australia.** - Pathways into Australia such as on nursery plants from overseas are managed via Post Entry Quarantine. - Natural dispersal is a possible pathway from neighbouring countries into northern Australia. Northern Australia Quarantine Strategy (NAQS) surveys offer an early detection mechanism to detect natural introductions from the north. If an outbreak is detected the pathway will need to be identified and closed, where possible. • **The dispersal ability of the pest.** - American serpentine leafminer has the ability to naturally spread over short distances through flight or wind assisted dispersal. - Nursery material that is a host of American serpentine leafminer (including containerized plants, seedlings, bare rooted plants, etc.; see Appendix 1 for host list) and cut flowers that are host plants are the most likely means of spread to new areas. They may carry all life stages of the pest and will need to be managed to reduce the spread of the pest to new areas. Nursery material and cut flowers that are not a host of American serpentine leafminer are at significantly lower risk (but may still transport adults across short periods if they have been in close proximity with host plants).

- Plant material (potentially including fresh hay and crop debris) and soil could also facilitate the spread of the pest (via adults and pupae) to new areas. Tools, equipment, machinery and vehicles are lower risk but may still transport hitchhiking adults or pupae via soil deposits.
- American serpentine leafminer is considered unlikely to travel with harvested fruit (without leaves) as the process of harvesting or moving produce is likely to cause adults to disperse and provided soil and leaf material is absent it is unlikely to spread larvae and pupae. Leafminer are also not expected to travel via the movement of seed.
- Fresh green or leafy vegetables or fresh legume vegetables (e.g. snow peas) may allow the spread of eggs and larvae and potentially adults and pupae. Therefore, movement restrictions or mandatory treatments may be needed for some vegetable crops.
- See Table 3 for further details on possible dispersal pathways

- **The capability to detect *Liriomyza* leafminers at very low densities for the purpose of declaring freedom, and that all sites affected by the pest have or can be found.**
- Surveillance typically relies on visual inspection of host plants for mining activity as well as the use of yellow sticky traps and sweep netting.
- **The ability to put into place surveillance to confirm proof of freedom.**
- Surveillance options are available for American serpentine leafminer and include visual inspection of host plants for mines, yellow sticky traps and sweep netting.
- **Whether community consultation activities have or will be undertaken.**
- In an urban environment, community consultation will be critical to assist in securing public support for delimiting surveillance and an eradication program.
- Given the symptoms caused by this pest on a wide range of plants, general surveillance activities should be implemented comprising of awareness material, media releases, web-based reporting tools and the Exotic Plant Pest Hotline (1800 084 881).

6. EMERGENCY PLANT PEST RESPONSE DEED AND PLANTPLAN

The Emergency Plant Pest Response Deed (EPPRD) and PLANTPLAN should be referred to, in conjunction with this contingency plan when developing a Response Plan in the event of an American serpentine leafminer detection in Australia.

6.1 Emergency Plant Pest Response Deed

PHA is the custodian of the EPPRD. This is a formal legally binding agreement between PHA, the Australian Government, all state and territory governments and national plant industry body signatories.

It covers the management and funding of responses to Emergency Plant Pest (EPP) incidents, including the potential for Owner Reimbursement Costs (ORCs) for growers. It also formalises the role of plant industries' participation in decision making, as well as their contribution towards the costs related to approved responses.

Under the EPPRD an EPP is defined as a pest that meets one or more of the following criteria:

a) It is a known exotic Plant Pest the economic consequences of an occurrence of which would be economically or otherwise harmful for Australia, and for which it is considered to be in the regional and national interest to be free of the Plant Pest.
b) It is a variant form of an established Plant Pest which can be distinguished by appropriate investigative and diagnostic methods and which, if established in Australia, would have a regional and national impact.
c) It is a serious Plant Pest of unknown or uncertain origin which may, on the evidence available at the time, be an entirely new Plant Pest or one not listed in Schedule 13 and which if established in Australia is considered likely to have an adverse economic impact regionally and nationally.
d) It is a Plant Pest already found in Australia that:
    (i) is restricted to a defined area through the use of regulatory measures intended to prevent further spread of the pest out of the defined area or into an endangered area; and
    (ii) has been detected outside the defined area; and
    (iii) is not a native of Australia; and
    (iv) is not the subject of any instrument for management which is agreed to be effective risk mitigation and management at a national level; and
    (v) is considered likely to have an adverse economic impact such that an emergency response is required to prevent an incident of regional and national importance.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of the EPPRD.

6.2 PLANTPLAN

Underpinning the EPPRD is PLANTPLAN, the agreed technical response plan for an EPP incident. It provides nationally consistent guidelines for response procedures, outlining the phases of an incursion, as well as the key roles and responsibilities of industry and government during each of the phases.

See: www.planthealthaustralia.com.au/biosecurity/emergency-plant-pest-response-deed/ for more information and the most recent version of PLANTPLAN.

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Addendum:

Gitonga ZM, Chabi-Olaye A, Mithöfer D, Okello JJ & Ritho CN. 2010. Control of invasive *Liriomyza* leafminer species and compliance with food safety standards by small scale snow pea farmers in Kenya. *Crop Protection* 298: 1472-1477.

8. APPENDICES

Appendix 1: Known hosts of American serpentine leafminer

The American serpentine leafminer (*Liriomyza trifolii)* is highly polyphagous, with a wide host range across at least 15 families (Table 12). In order to facilitate trade and market access decisions, a thorough review was conducted as part of the Horticulture Innovation Australia funded Project MT16004 (R&E program for control, eradication and preparedness for vegetable leafminer), with a focus on evidence for lifecycle completion in the field. The results of this review, including commercial, ornamental and non-cultivated hosts of the American serpentine leafminer (*L. trifolii*), are presented in Table 13, Table 14 and Table 15 below. No guarantee can be made that these lists are exhaustive. Each record was scrutinized based on evidence provided showing completion of lifecycle within the field, and then included within the appropriate table. It is important to note that non-cultivated plants often serve as important reservoirs for *Liriomyza* pests (e.g. Schuster et al. 1991), and upon incursion into a new region, host ranges may expand to include native plant species. Moreover, host preferences may vary considerably across populations. For example, although chili (*Capsicum* spp.) is widely reported internationally as a favoured host of the closely related agromyzid *Liriomyza sativae*, the population of *L. sativae* which has established within the Torres Strait (IPPC 2017) has very rarely been observed to attack chili (despite it being an abundant garden host in the region) (Elia Pirtle pers. comm).

Number of host records for the American serpentine leafminer (*Liriomyza trifolii*) within plant families

Only records with either comprehensive or partial evidence of lifecycle completion in the field are included (see Table 13 and Table 14). Multiple records of the same species are included. Thus, the number of records does not indicate total number of affected species within each family. *Liriomyza trifolii* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 12 Number of host records for the American serpentine leafminer (Liriomyza trifolii) within plant families.

Family	Number of records
Asteraceae	77
Fabaceae	38
Solanaceae	26
Cucurbitaceae	22
Brassicaceae	15
Amaranthaceae	12
Caryophyllaceae	9
Malvaceae	8
Apiaceae	6
Amaryllidaceae	5
Lamiaceae	4
Oxalidaceae	3
Plantaginaceae	3
Gentianaceae	2
Convolvulaceae	2

Family	Number of records
Verbenaceae	2
Zygophyllaceae	2
Apocynaceae	1
Araliaceae	1
Boranginaceae	1
Campanulaceae	1
Cleomaceae	1
Commelinaceae	1
Euphorbiaceae	1
Polemoniaceae	1
Polygalaceae	1
Primulaceae	1
Ranunculaceae	1
Rosaceae	1
Sapindaceae	1
Tropaeolaceae	1
Turneraceae	1

Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*) has been provided within the scientific literature

This includes all records where the following conditions were met:

(1) Adults were reared from plant material infested with eggs or from larvae collected in the field; and

(2) Emerging adults were subsequently identified via morphology or molecular diagnostics; and

(3) Scientific names of hosts are reported

In addition, all records from Kenneth Spencer, the foremost authority on leafminer taxonomy, are included. Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included only if there are other records for species within this genus that met both conditions. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records confirming lifecycle completion for that same species that met both conditions. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report only partial confirmations of lifecycle completion are included in Table 14, while unverified records are included in Table 15. *Liriomyza trifolii* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 13 Commercial, ornamental and non-cultivated plants for which comprehensive evidence of lifecycle completion in the field for the American serpentine leafminer (Liriomyza trifolii) has been provided within the scientific literature.

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Beet; Chard; Beetroot	*Beta vulgaris*		Amaranthaceae	Stegmaier (1966); Spencer (1973); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Sugarbeet	*Beta vulgaris*	*saccharifera*	Amaranthaceae		CABI (2019)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Quinoa	*Chenopodium quinoa*		Amaranthaceae	Srinivasan et al., (1995)	
Commercial	Fat hen	*Chenopodium* spp.		Amaranthaceae		SPHD (2016)
Commercial	Spinach; Silverbeet	*Spinacia oleracea*		Amaranthaceae	Stegmaier (1966); Spencer (1973); Srinivasan et al., (1995)	Stegmaier (1968); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Onion; Shallot; Multiplier onion	*Allium cepa*		Amaryllidaceae	Stegmaier (1966); Spencer (1973); Spencer (1983)	Stegmaier (1968); Fagoonee and Toory (1984); Scheffer and Lewis (2005); Andersen et al. (2008); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Japanese bunching onion; Scallion; Welsh onion	*Allium fistulosum*		Amaryllidaceae	Chirinos et al. (2014)	EPPO (2020)
Commercial	Chive	*Allium schoenoprasum*		Amaryllidaceae	Spencer (1973)	SPHD (2016); Sooda et al. (2017)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Celery	*Apium graveolens*		Apiaceae	Stegmaier (1966); Spencer (1973)	Fagoonee and Toory (1984); Foster (1986); Altieri et al. (1999); Weintraub (1999); Scheffer and Lewis (2005); Andersen et al. (2008); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020); Andersen and Hofsvang (2010)
Commercial	Celery	*Apium graveolens*	*dulce*	Apiaceae		Stegmaier (1968); CABI (2019)
Commercial	Carrot	*Daucus carota*		Apiaceae	Stegmaier (1966); Spencer (1973)	Stegmaier (1968); Scheffer and Lewis (2005); Capinera (2014); SPHD (2016)
Commercial	Carrot	*Daucus carota*	*sativus*	Apiaceae		Scheffer et al. (2006)
Commercial	Carthamum; Safflower	*Carthamus tinctorius*		Asteraceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Lettuce; Garden lettuce	*Lactuca sativa*		Asteraceae	Stegmaier (1966); Spencer (1973); Masetti et al. (2006)	Stegmaier (1968); Fagoonee and Toory (1984); Olivera and Bordat (1996); Scheffer and Lewis (2005); Andersen et al. (2008); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial		*Lactuca* sp.		Asteraceae		Stegmaier (1968)
Commercial	Gai lan; Cabbage; Broccoli; Caisin; Cabbage; Cauliflower; Field cabbage	*Brassica oleracea*		Brassicaceae		Scheffer and Lewis (2005)
Commercial	Kale	*Brassica oleracea*	*acephala*	Brassicaceae	Foba et al. (2015)	
Commercial	Cauliflower	*Brassica oleracea*	*botrytis*	Brassicaceae		Fagoonee and Toory (1984)
Commercial	Cabbage	*Brassica oleracea*	*capitata*	Brassicaceae		Fagoonee and Toory (1984); Scheffer et al. (2006)A barcoding…)
Commercial	Brussels sprout	*Brassica oleracea*	*gemmiferae*	Brassicaceae		Fagoonee and Toory (1984)
Commercial		*Cleome gynandra*		Cleomaceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Wax gourd; White goard; Chinese melon; China squash; Ash gourd	*Benincasa hispida*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Watermelon	*Citrullus lanatus*		Cucurbitaceae	Foba et al. (2015)	Scheffer and Lewis (2005); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Watermelon	*Citrullus vulgaris*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Coccinia	*Coccinia indica*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Snake gourd	*Cucumis anguina*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Melon; Cantaloupe; Muskmelon; Honey melon; Cassaba melon	*Cucumis melo*		Cucurbitaceae	Stegmaier (1966); Spencer (1973)	Stegmaier (1968); Scheffer and Lewis (2005); Guimarães et al. (2009); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Cucumber	*Cucumis sativus*		Cucurbitaceae	Stegmaier (1966); Spencer (1973); Srinivasan et al., (1995)	Stegmaier (1968); Capinera (2014); Gao (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial		*Cucumis* sp./spp.		Cucurbitaceae	Stegmaier (1966)	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Butternut squash; Pumpkin; Crookneck squash	*Cucurbita moschata*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Courgette; Zucchini; Pumpkin	*Cucurbita pepo*		Cucurbitaceae	Stegmaier (1966); Spencer (1973); Foba et al. (2015)	Stegmaier (1968); Olivera and Bordat (1996); Scheffer and Lewis (2005); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Squash	*Cucurbita* sp./spp.		Cucurbitaceae	Stegmaier (1966)	Stegmaier (1968)
Commercial	Sponge gourd; Ridge gourd	*Luffa aegyptiaca*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Towel gourd	*Luffa cylindrica*		Cucurbitaceae	Gao (2014)	
Commercial	Bitter gourd; Balsam pear	*Momordica charantia*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Smooth gourd	*Trichosanthes dioica*		Cucurbitaceae	Srinivasan et al., (1995)	
Commercial	Pidgeon pea	*Cajanus cajan*		Fabaceae	Srinivasan et al., (1995)	Fagoonee and Toory (1984)
Commercial	Sword bean	*Canavalia gladiata*		Fabaceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Sunn hemp	*Crotalaria juncea*		Fabaceae	Srinivasan et al., (1995)	SPHD (2016)
Commercial	Cluster beans	*Cyamopsis tetragonolobus*		Fabaceae	Srinivasan et al., (1995)	
Commercial	Creeper bean	*Dolichos lablab*		Fabaceae	Srinivasan et al., (1995)	
Commercial	Soybean; Soya bean	*Glycine max*		Fabaceae	Srinivasan et al., (1995)	Fagoonee and Toory (1984); SPHD (2016)
Commercial	Sweet Dolichos; Field bean	*Lablab purpureus*		Fabaceae	Srinivasan et al., (1995)	
Commercial	Mung bean	*Phaseolus aureus*		Fabaceae	Stegmaier (1966)	Stegmaier (1968)
Commercial	Sieva; Lima bean; Lima bean	*Phaseolus lunatus*		Fabaceae	Spencer (1973)	Fagoonee and Toory (1984); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	String bean	*Phaseolus* sp./spp.		Fabaceae	Stegmaier (1966)	Stegmaier (1968); Scheffer et al. (2006); Scheffer and Lewis (2005)
Commercial		*Phaseolus tribulus*		Fabaceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Green bean; French bean; Kidney bean; Bean; Common bean; Snap bean	*Phaseolus vulgaris*		Fabaceae	Spencer (1983); de souza (1986); Srinivasan et al., (1995); Masetti et al. (2006); Foba et al. (2015)	Fagoonee and Toory (1984); Olivera and Bordat (1996); Altieri et al. (1999); Kox et al (2005); Andersen et al. (2008); Musundire et al. (2012); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Garden pea; Pea; Snow pea; Sugar snap	*Pisum sativum*		Fabaceae	Spencer (1973); Srinivasan et al., (1995); Foba et al. (2015)	Stegmaier (1968); Fagoonee and Toory (1984); Scheffer and Lewis (2005); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial		*Pisum* sp./spp.		Fabaceae	Stegmaier (1966)	Stegmaier (1968); Scheffer et al. (2006)
Commercial	White clover	*Trifolium repens*		Fabaceae	Stegmaier (1966); Spencer (1973)	Stegmaier (1968); Andrade et al. (1989); SPHD (2016)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Fenugreek	*Trigonella foenum-graecum*		Fabaceae	Srinivasan et al., (1995)	
Commercial	Fenugreek	*Trigonella* spp.		Fabaceae		SPHD (2016)
Commercial	Black gram	*Vigna mungo*		Fabaceae	Srinivasan et al., (1995)	
Commercial	Mungbean; Green gram	*Vigna radiata*		Fabaceae	Srinivasan et al., (1995)	Andersen et al. (2008)
Commercial	Red bean; Black eyed pea; Cowpea	*Vigna sinensis*		Fabaceae	Stegmaier (1966); Gao (2014)	Stegmaier (1968); Fagoonee and Toory (1984); SPHD (2016)
Commercial	Bean, gram	*Vigna* sp./spp.		Fabaceae		Stegmaier (1968); CABI (2019); EPPO (2020)
Commercial	Walp Cowpea; Yard-long bean; Long bean; Snakebean; Cowpea	*Vigna unguiculata*		Fabaceae	Srinivasan et al., (1995); Foba et al. (2015)	
Commercial	Okra	*Abelmoschus esculentus*		Malvaceae	Stegmaier (1966); Spencer (1973); Srinivasan et al., (1995); Foba et al. (2015)	SPHD (2016)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial		*Corchorus olitorius*		Malvaceae	Srinivasan et al., (1995)	
Commercial	Cotton	*Gossypium arboreum*		Malvaceae	Srinivasan et al., (1995)	
Commercial	Cotton	*Gossypium barbadense*		Malvaceae	Srinivasan et al., (1995)	
Commercial	Upland cotton; Cotton	*Gossypium hirsutum*		Malvaceae	Srinivasan et al., (1995)	SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Pepper; Sweet pepper; Green pepper; Chili; Capsicum	*Capsicum annuum*		Solanaceae	Spencer (1973); Srinivasan et al., (1995)	Kox et al (2005); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Bell pepper	*Capsicum frutescens*		Solanaceae	Srinivasan et al., (1995)	Altieri et al. (1999)
Commercial	Pepper	*Capsicum sp./spp.*		Solanaceae	Stegmaier (1966)	Stegmaier (1968); Scheffer and Lewis (2005)
Commercial	Tomato	*Lycopersicum esculentum*		Solanaceae	Stegmaier (1966); Spencer (1973); Spencer (1983); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984); Olivera and Bordat (1996); Kox et al (2005); Scheffer et al. (2006); Capinera (2014); SPHD (2016);

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
						CABI (2019); EPPO (2020)
Commercial	Tomato	*Solanum lycopersicum*		Solanaceae	Foba et al. (2015)	Altieri et al. (1999); Macdonald et al. (2002); Hanafi (2005); Scheffer et al. (2006); Andersen et al. (2008); Musundire et al. (2011); Musundire et al. (2012)
Commercial	Brinjal eggplant; Eggplant; common eggplant; Pickling eggplant	*Solanum melongena*		Solanaceae	Stegmaier (1966); Spencer (1973); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984); Capinera (2014); SPHD (2016)
Commercial		*Solanum* sp.		Solanaceae		Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Commercial	Potato	*Solanum tuberosum*		Solanaceae	Stegmaier (1966); Spencer (1973); de souza (1986); Srinivasan et al., (1995); Foba et al. (2015)	Stegmaier (1968); Fagoonee and Toory (1984); Macdonald et al. (2002); Scheffer and Lewis (2005); De Freitas Bueno et al., (2007); Andersen et al. (2008); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Cock's comb	*Celosia argentea*		Amaranthaceae	Srinivasan et al., (1995)	
Ornamental	Aster	*Aster* sp./spp.		Asteraceae	Stegmaier (1966)	Stegmaier (1968); SPHD (2016); CABI (2019); EPPO (2020); Andersen and Hofsvang (2010)
Ornamental	Pot marigold	*Calendula officinalis*		Asteraceae	Srinivasan et al., (1995)	
Ornamental	Chinese aster	*Callistephus chinensis*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968); SPHD (2016)
Ornamental	Garland chrysanthemum; Chrysanthemum	*Chrysanthemum sp.*		Asteraceae	Nakamura et al. (2013)	Bartlett and Powell (1981); Andersen et al. (2008); Musundire et al. (2011); Andersen and Hofsvang (2010)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Ornamental	Chrysanthemum	*Chrysanthemum sp.*	*Flamenca*	Asteraceae	Spencer (1983)	
Ornamental	Chrysanthemum	*Chrysanthemum spp.*		Asteraceae	Spencer (1983)	Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Dahlia	*Dahlia* sp./spp.		Asteraceae	Stegmaier (1966)	Fagoonee and Toory (1984); Stegmaier (1968); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Dahlia	*Dahlia variabilis*		Asteraceae	Srinivasan et al., (1995)	
Ornamental	Blue mistflower; Mistflower	*Eupatorium coelestinum*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Ornamental		*Flaveria* sp.		Asteraceae		Stegmaier (1968); Srinivasan et al., (1995)
Ornamental	Clustered yellowtops	*Flaveria trinervia*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Ornamental	Gaillardia; Maroon blanketflower	*Gaillardia amblyodon*		Asteraceae	Srinivasan et al., (1995)	SPHD (2016)
Ornamental	Common blanketflower	*Gaillardia aristata*		Asteraceae	Stegmaier (1966)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Ornamental	Gerbera; Barberton daisy; Transvaal daisy	*Gerbera jamesonii*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968)
Ornamental	Gerbera	*Gerbera* sp./spp.		Asteraceae	Stegmaier (1966); Nakamura et al. (2013)	Stegmaier (1968); Fagoonee and Toory (1984); Kox et al (2005); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Sunflower; Common sunflower	*Helianthus annuus*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984); SPHD (2016)
Ornamental	Ragwort	*Senecio sp./spp.*		Asteraceae	Stegmaier (1966)	SPHD (2016)
Ornamental	Marigold; Aztec marigold	*Tagetes erecta*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968); SPHD (2016)
Ornamental	Marigold; French marigold	*Tagetes patula*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); Capinera (2014); SPHD (2016)
Ornamental	Marigold	*Tagetes spp.*		Asteraceae		Stegmaier (1968); Fagoonee and Toory (1984)
Ornamental		*Zinnia sp./spp.*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); Fagoonee and Toory

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
						(1983); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Indian pink; China pink	*Dianthus chinensis*		Caryophyllaceae	Srinivasan et al., (1995)	SPHD (2016)
Ornamental	Dianthus	*Dianthus* spp.		Caryophyllaceae		SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Gypsophila; Bbaby's breath	*Gypsophila* sp./spp.		Caryophyllaceae	Stegmaier (1966)	Stegmaier (1968); Andersen and Hofsvang (2010); Capinera (2014); SPHD (2016); CABI (2019); EPPO (2020)
Ornamental	Salvia; Azure blue sage	*Salvia azurea*		Lamiaceae	Srinivasan et al., (1995)	SPHD (2016)
Ornamental	Yellow hibiscus	*Hibiscus panduraeformis*		Malvaceae	Srinivasan et al., (1995)	
Ornamental	Indian hemp	*Hibiscus subdarifa*		Malvaceae	Srinivasan et al., (1995)	
Ornamental		*Gerbera sp.*		Primulaceae		Bartlett and Powell (1981)
Ornamental	Petunia; Large white petunia	*Petunia axillaris*		Solanaceae	Srinivasan et al., (1995)	
Ornamental	Petunia	*Petunia* sp./spp.		Solanaceae	Stegmaier (1966)	Stegmaier (1968)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated		*Alternanthera ficoidea*		Amaranthaceae	Srinivasan et al., (1995)	
Non Cultivated		*Amaranthus spinosus*		Amaranthaceae	Srinivasan et al., (1995)	
Non Cultivated		*Amaranthus* spp.		Amaranthaceae		Fagoonee and Toory (1984)
Non Cultivated	Elephant-head amaranth	*Amaranthus tricolor*		Amaranthaceae	Srinivasan et al., (1995)	
Non Cultivated	Goosefoot; Lamb's quarters; Fat hen	*Chenopodium album*		Amaranthaceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Mugwort	*Artemisia vulgaris*		Asteraceae	Masetti et al. (2004)	SPHD (2016)
Non Cultivated	Sea myrtle; Groundsel tree	*Baccharis halimifolia*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); Hudson and Stiling (1997); SPHD (2016)
Non Cultivated	Groundsel tree	*Baccharis* sp.		Asteraceae		Stegmaier (1968)
Non Cultivated	Shepherd's needles; Spanish needles	*Bidens alba*		Asteraceae	Schuster et al. (1991)	Capinera (2014); CABI (2019); EPPO (2020)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated	Black jack; Spanish needle; hairy beggarticks	*Bidens pilosa*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984); SPHD (2016)
Non Cultivated	Bidens	*Bidens* sp.		Asteraceae		Stegmaier (1968); Scheffer and Lewis (2005)
Non Cultivated	Distaff thistle	*Carthamus* spp.		Asteraceae		SPHD (2016)
Non Cultivated		*Cirsium arvense*		Asteraceae	Masetti et al. (2004)	
Non Cultivated	American burnweed; Pilewort; Fireweed	*Erechtites hieracifolia*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); Capinera (2014)
Non Cultivated	Fireweed	*Erechtites* sp.		Asteraceae		Stegmaier (1968)
Non Cultivated	Blue mistflower; Mistflower	*Eupatorium coelestinum*		Asteraceae		SPHD (2016)
Non Cultivated	Late boneset	*Eupatorium serotinum*		Asteraceae	Stegmaier (1966)	SPHD (2016)
Non Cultivated	Quickweed; Shaggy soldier	*Galinsoga ciliata*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); SPHD (2016)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated	Cudweed	*Gnaphalium spathalium*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Carolina woollywhite	*Hymenopappus scabiosaeus*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Canada lettuce; Canada wild lettuce	*Lactuca canadensis*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated		*Lactuca serriola*		Asteraceae	Masetti et al. (2004)	
Non Cultivated	Melanthera	*Melanthera deltoidea*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated		*Parthenium hysterophorus*		Asteraceae	Srinivasan et al., (1995)	
Non Cultivated		*Picris echioides*		Asteraceae	Masetti et al. (2004)	
Non Cultivated		*Picris hieracioides*		Asteraceae	Masetti et al. (2004)	
Non Cultivated	Butterweed	*Senecio glabellus*		Asteraceae	Stegmaier (1966)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated	Perennial sow thistle	*Sonchus arvensis*		Asteraceae	Srinivasan et al., (1995)	SPHD (2016)
Non Cultivated	Spiny sow thistle; Prickly sow thistle	*Sonchus asper*		Asteraceae	Stegmaier (1966); Masetti et al. (2004)	Stegmaier (1968)
Non Cultivated	Common sow thistle	*Sonchus oleraceus*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995); Masetti et al. (2004)	Fagoonee and Toory (1984); SPHD (2016)
Non Cultivated	Cinderella weed; Nodeweed	*Synedrella nodiflora*		Asteraceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968)
Non Cultivated	Dandelion	*Taraxacum* sp./spp.		Asteraceae	Masetti et al. (2004)	SPHD (2016)
Non Cultivated	Coat buttons; Tridax	*Tridax procumbens*		Asteraceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Cocklebur	*Xanthium sp./spp.*		Asteraceae	Stegmaier (1966)	Stegmaier (1968); SPHD (2016)
Non Cultivated		*Commelina benghalensis*		Commelinaceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated		*Convolvulus arvensis*		Convolvulaceae	Srinivasan et al., (1995)	
Non Cultivated		*Blastania garcinia*		Cucurbitaceae	Srinivasan et al., (1995)	
Non Cultivated		*Momordica tuberosa*		Cucurbitaceae	Srinivasan et al., (1995)	
Non Cultivated	Castor bean; Castor oil bush	*Ricinus communis*		Euphorbiaceae	Srinivasan et al., (1995)	SPHD (2016)
Non Cultivated	Desert cassia	*Cassia biflora*		Fabaceae	Srinivasan et al., (1995)	SPHD (2016)
Non Cultivated	Rattlepod; Rattlebox	*Crotalaria incana*		Fabaceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Black medic	*Medicago lupulina*		Fabaceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Pongamia	*Pongamia pinnata*		Fabaceae	Srinivasan et al., (1995)	
Non Cultivated	Corkwood tree	*Sesbania grandiflora*		Fabaceae	Srinivasan et al., (1995)	
Non Cultivated	Berseem	*Trifolium alexandricum*		Fabaceae	Srinivasan et al., (1995)	
Non Cultivated	Carolina Pirqueta	*Piriqueta cistoides*	*caroliniana*	Passifloraceae	Stegmaier (1968)	
Non Cultivated		*Polygala persicariifolia*		Polygalaceae	Srinivasan et al., (1995)	

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated		*Cardiospermum halicacabum*		Sapindaceae	Srinivasan et al., (1995)	
Non Cultivated		*Datura spp.*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Manyflower tobacoo	*Nicotiana acuminata*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Wild tobacco	*Nicotiana goodspeedi*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Wild tobacco	*Nicotiana paniculata*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Wild tobacco	*Nicotiana simusslands*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Wild tobacco	*Nicotiana velutina*		Solanaceae	Srinivasan et al., (1995)	
Non Cultivated	Ground cherry	*Physalis* sp./spp.		Solanaceae	Stegmaier (1966)	Stegmaier (1968); SPHD (2016)
Non Cultivated	American black nightshade	*Solanum americanum*		Solanaceae	Schuster et al. (1991)	Capinera (2014)
Non Cultivated	Black nightshade	*Solanum nigrum*		Solanaceae	Stegmaier (1966); Srinivasan et al., (1995)	Stegmaier (1968); Fagoonee and Toory (1984)

Host type	Common names	Scientific name	Variety	Family	Records with comprehensive evidence	All other records (partial and unverified records)
Non Cultivated	Big caltrop; Caltrop	*Kallstroemia maxima*		Zygophyllaceae	Stegmaier (1966)	Stegmaier (1968)
Non Cultivated	Bindii; Caltrop	*Tribulus terrestris*		Zygophyllaceae	Stegmaier (1966)	Stegmaier (1968); SPHD (2016)

Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*) has been provided within the scientific literature

This includes all records where:

(1) Adults were reared from field collected plant material infested with larvae, but no subsequent identification via morphology or molecular diagnostics was reported; or

(2) Laboratory feeding and oviposition preference experiments were conducted that did not confirm lifecycle completion; or

(3) Identification via morphology or molecular diagnostics was reported, but collection methods in the field were not reported. Lifestyle completion cannot be confirmed in these instances due to the potential use of non-specific collection methods (e.g. sticky traps or sweep nets); or

(4) Laboratory cultures confirmed lifecycle completion, but evidence of lifecycle completion in the field was not provided; or

(5) Only common names of plant were given, but the scientific name could be confidently inferred; or

(6) The record reports presence of *L. trifolii* on imported material detected at the border.

Records for plants identified only to the genus level (sp. or spp.) that did not meet the above conditions are included here only if there are other records for species within this genus that met one of these conditions outlined above. Records for specific varieties within a species that did not meet the above conditions are included only if there are other records for that same species that meet one of the conditions outlined above. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 13, while unverified records are included in Table 15. *Liriomyza trifolii* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 14 Commercial, ornamental and non-cultivated plants for which only partial evidence of lifecycle completion in the field for the American serpentine leafminer (Liriomyza trifolii) has been provided within the scientific literature

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Leek	*Allium porrum*		Amaryllidaceae	Fagoonee and Toory (1984)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Garlic	*Allium sativum*		Amaryllidaceae	Fagoonee and Toory (1984); Ortega et al. (2014); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Coriander	*Coriandrum sativum*		Apiaceae	Fagoonee and Toory (1984)
Commercial	Parsley	*Petroselinum crispum*		Apiaceae	Fagoonee and Toory (1984)
Commercial		*Anthemis nobilis*		Asteraceae	Andrade et al. (1989)
Commercial	Turnip	*Brassica campestris*	*rapa*	Brassicaceae	Fagoonee and Toory (1984)
Commercial	Chinese cabbage	*Brassica chinensis*	*chinensis*	Brassicaceae	Fagoonee and Toory (1984)
Commercial	Leaf mustard; Mustard greens; Radish; Chinese mustard; Indian mustard; Mustard	*Brassica juncea*		Brassicaceae	Fagoonee and Toory (1984); Scheffer et al. (2006); Scheffer and Lewis (2005); Andersen et al. (2008)
Commercial	Turnip; Rinsho; Chinese cabbage; Bokchoy	*Brassica rapa*		Brassicaceae	Scheffer et al. (2006)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Pak choi; Pechay; Chinese cabbage	*Brassica rapa*	*chinensis*	Brassicaceae	Scheffer and Lewis (2005); Andersen et al. (2008); SPHD (2016); CABI (2019); EPPO (2020)
Commercial	Chinese mustard	*Brassica rapa*	*pekinensis*	Brassicaceae	Andersen et al. (2008)
Commercial	Radish; Chinese radish (Daikon); Wild radish; Garden radish; White radish	*Raphanus sativus*		Brassicaceae	Fagoonee and Toory (1984)
Commercial	White radish	*Raphanus sativus*	*longipinnatus*	Brassicaceae	Andersen et al. (2008)
Commercial	Sweet potato	*Ipomoea batatas*		Convolvulaceae	Fagoonee and Toory (1984)
Commercial	Pumpkin; Winter squash; Squash	*Cucurbita maxima*		Cucurbitaceae	Fagoonee and Toory (1984)
Commercial	Bottle gourd	*Lagenaria siceraria*		Cucurbitaceae	Fagoonee and Toory (1984)
Commercial	Snake gourd	*Lagenaria vulgaris*		Cucurbitaceae	Fagoonee and Toory (1984)
Commercial	Groundnut; Peanut	*Arachis hypogaea*		Fabaceae	Fagoonee and Toory (1984); SPHD (2016)
Commercial	Chickpea	*Cicer arietinum*		Fabaceae	Fagoonee and Toory (1984)
Commercial	Alfalfa; Lucerne	*Medicago sativa*		Fabaceae	Stegmaier (1968); Fagoonee and Toory (1984); SPHD (2016); CABI (2019); EPPO (2020)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Commercial	Fenugreek	*Trigonella foingraecum*		Fabaceae	Fagoonee and Toory (1984)
Commercial	Broad bean; Faba bean; Fava bean	*Vicia faba*		Fabaceae	Fagoonee and Toory (1984); Shahein and El-Maghraby (1988); Musundire et al. (2012)
Commercial	Broad bean; Vetch	*Vicia* spp.		Fabaceae	Fagoonee and Toory (1984); SPHD (2016)
Commercial	Basil; Sweet basil; Thai basil	*Ocimum basilicum*		Lamiaceae	Andersen et al. (2008)
Commercial		*Ocimum americanum*		Lamiaceae	Andersen and Hofsvang (2010)
Commercial	Okra	*Hibiscus esculentus*		Malvaceae	Stegmaier (1968); Fagoonee and Toory (1984)
Ornamental		*Asclepias* sp.		Apocynaceae	Andersen and Hofsvang (2010)
Ornamental	Daisy	*Bellis perennis*		Asteraceae	Fagoonee and Toory (1984)
Ornamental	Chrysanthemum; Florists' chrysanthemum	*Chrysanthemum morifolium*		Asteraceae	Fagoonee and Toory (1984); Andrade et al. (1989); CABI (2019)
Ornamental	Coreopsis	*Coreopsis sp.*		Asteraceae	Scheffer and Lewis (2005)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental		*Dendranthema sp.*		Asteraceae	Miller et al. (1985); Andersen and Hofsvang (2010)
Ornamental		*Dimorphotheca aurantiaca*		Asteraceae	Kox et al (2005)
Ornamental		*Echinacea* sp.		Asteraceae	Andersen and Hofsvang (2010)
Ornamental		*Leucanthemum* sp.		Asteraceae	Kox et al (2005)
Ornamental		*Solidago hybrida*		Asteraceae	Andersen and Hofsvang (2010)
Ornamental	Goldenrod	*Solidago sp.*		Asteraceae	Andersen and Hofsvang (2010)
Ornamental	Marigold	*Tagetes indica*		Asteraceae	Fagoonee and Toory (1984)
Ornamental		*Trachelium* sp.		Campanulaceae	Andersen and Hofsvang (2010)
Ornamental	Gypsophila; Baby's breath	*Gypsophila paniculata*		Caryophyllaceae	Andersen and Hofsvang (2010)
Ornamental		*Eustoma sp.*		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Lisianthus sp.*		Gentianaceae	Andersen and Hofsvang (2010)
Ornamental		*Salvia splendens*		Lamiaceae	Fagoonee and Toory (1984)
Ornamental	Snapdragon	*Antirrhinum majus*		Plantaginaceae	Fagoonee and Toory (1984)
Ornamental	Phlox	*Phlox drummondii*		Polemoniaceae	Fagoonee and Toory (1984)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Ornamental	Buttercup	*Ranunculus sp.*		Ranunculaceae	Andersen and Hofsvang (2010)
Ornamental		*Rosa sp.*		Rosaceae	Andersen and Hofsvang (2010)
Ornamental	Nasturtium	*Tropaeolum* sp.		Tropaeolaceae	Fagoonee and Toory (1984)
Ornamental		*Lantana camara*		Verbenaceae	Fagoonee and Toory (1984)
Ornamental	Vervain	*Verbena officinalis*		Verbenaceae	Fagoonee and Toory (1984)
Non cultivated	Rough pigweed	*Amaranthus retroflexus*		Amaranthaceae	Fagoonee and Toory (1984)
Non cultivated		*Hydrocotyle bonariensis*		Araliaceae	Fagoonee and Toory (1984)
Non cultivated	Chick weed; Goatweed	*Ageratum conyzoides*		Asteraceae	Fagoonee and Toory (1984)
Non cultivated		*Eclipta prostrata*		Asteraceae	Fagoonee and Toory (1984)
Non cultivated		*Erigeron canadensis*		Asteraceae	Fagoonee and Toory (1984)
Non cultivated		*Galinsoga caracasana*		Asteraceae	Andrade et al. (1989)

Host type	Common names	Scientific name	Variety	Family	All records (including any unverified records)
Non cultivated		*Cordia myxa*		Boraginaceae	Fagoonee and Toory (1984)
Non cultivated		*Silene gallica*		Caryophyllaceae	Andrade et al. (1989)
Non cultivated		*Spergula arvensis*		Caryophyllaceae	Andrade et al. (1989)
Non cultivated		*Oxalis debilis*	*corymbosa*	Oxalidaceae	Fagoonee and Toory (1984)
Non cultivated		*Oxalis latifolia*		Oxalidaceae	Fagoonee and Toory (1984)
Non cultivated		*Oxalis repens*		Oxalidaceae	Fagoonee and Toory (1984)
Non cultivated	Blue toadflax	*Linaria canadensis*		Plantaginaceae	Fagoonee and Toory (1984)
Non cultivated		*Plantago lanceolata*		Plantaginaceae	Fagoonee and Toory (1984)
Non cultivated		*Solanum auriculatum*		Solanaceae	Fagoonee and Toory (1984)
Non cultivated		*Solanum indicum*		Solanaceae	Fagoonee and Toory (1984)

Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the American serpentine leafminer (*Liriomyza trifolii*)

This includes all records for which no evidence was presented within the record for lifecycle completion and no evidence was shown for confident species identification, or evidence could not suitably be accessed for verification. We are not disputing the veracity of these records - we simply did not have enough information available to verify if these hosts support lifecycle completion in the field. This includes all records where:

(1) Adults were collected by sweep netting the presumed host, but neither larval activity nor lifecycle completion was confirmed; or

(2) A scientific name could not reliably be assumed from a reported common name; or

(3) Collection and identification methodology were unclear or unreported; or

(4) The record could not be accessed or suitably translated to confirm collection and identification methodology.

For some unverified species within this table, there may be other congeneric species that have been either comprehensively or partially confirmed as a host plant, and thus these would appear within Table 13 and Table 14. Scientific names in the table appear as they were originally cited. A number of these names are now recognised as synonyms, however for brevity, we are not reporting the currently accepted names for these taxa (refer to WorldFloraOnline.org, Australian Plant Census, or Catalogue of Life for current taxonomic decisions). All common names associated with each scientific name, across all reports, are included. Additionally, many plants labelled as commercial and ornamental have escaped cultivation and naturalised in Australia. Records that report comprehensive confirmations of lifecycle completion are included in Table 13, while records that report only partial confirmations of lifecycle completion are included in Table 14. *Liriomyza trifolii* is highly polyphagous and no guarantee can be made that this list is exhaustive.

Table 15 Commercial, ornamental and non-cultivated plants with unverified records of lifecycle completion in the field for the American serpentine leafminer (Liriomyza trifolii)

Host type	Common names	Scientific name	Variety	Family	Records
commercial	Leek; Leeks	*Allium ampeloprasum*		Amaryllidaceae	CABI (2019); EPPO (2020)
commercial	Malabar spinach	*Basella*		Basellaceae	SPHD (2016)
commercial	Angled luffa; Sing kwa; Towelsponge; Angled luffa	*Luffa acutangula*		Cucurbitaceae	CABI (2019)
commercial	Ground bean	*Macrotyloma* sp./spp.		Fabaceae	CABI (2019); SPHD (2016)
commercial	Runner bean	*Phaseolus coccineus*		Fabaceae	EPPO (2020)
commercial	Passionfruit	*Passiflora* sp.		Passifloraceae	Kahinga et al. (2017)

Host type	Common names	Scientific name	Variety	Family	Records
commercial	Oats	*Avena sativa*		Poaceae	SPHD (2016); CABI (2019)
commercial	Barley	*Hordeum spp.*		Poaceae	SPHD (2016); CABI (2019)
commercial	Arabica coffee	*Coffea arabica*		Rubiceae	CABI (2019)
commercial	Robusta coffee	*Coffea canephora*		Rubiceae	CABI (2019)
commercial		*Capsicum chinense*		Solanaceae	Holhuin-Pena et al (2019); EPPO (2020)
ornamental		*Alstroemeria* sp.		Alstroemeriaceae	Bartlett and Powell (1981)
ornamental		*Argyranthemum frutescens*		Asteraceae	EPPO (2020)
ornamental	Knapweed	*Centaurea* spp.		Asteraceae	SPHD (2016); CABI (2019)
ornamental		*Chrysanthemum indicum*		Asteraceae	CABI (2019)
ornamental		*Dahlia hybrids*		Asteraceae	EPPO (2020)
ornamental		*Dendranthema indicum*		Asteraceae	EPPO (2020)
ornamental		*Dendranthema x grandiflorum*		Asteraceae	EPPO (2020)

Host type	Common names	Scientific name	Variety	Family	Records
ornamental	Gazania; Treasure-flower	*Gazania* spp.		Asteraceae	SPHD (2016); CABI (2019)
ornamental		*Leucanthemum vulgare*		Asteraceae	EPPO (2020)
ornamental		*Leucanthemum x superbum*		Asteraceae	EPPO (2020)
ornamental		*Pericallis x hybrida*		Asteraceae	EPPO (2020)
ornamental	Butterweed	*Senecio cruentus*		Asteraceae	Stegmaier (1968)
ornamental		*Tanacetum parthenium*		Asteraceae	EPPO (2020)
ornamental		*Tanacetum vulgare*		Asteraceae	EPPO (2020)
ornamental	Carnation	*Dianthus caryophyllus*		Caryophyllaceae	EPPO (2020)
ornamental	Gypsophila; Baby's breath	*Gypsophila paniculata*		Caryophyllaceae	EPPO (2020)
ornamental	Vetchling	*Lathyrus* spp.		Fabaceae	SPHD (2016); CABI (2019); EPPO (2020)
ornamental	Sword lily	*Gladiolus hybridus*		Iridaceae	CABI (2019)
ornamental	Gladioli	*Gladiolus* spp.		Iridaceae	SPHD (2016)

Host type	Common names	Scientific name	Variety	Family	Records
ornamental	Shell flower	*Moluccella* sp./spp.		Lamiaceae	SPHD (2016); CABI (2019)
ornamental	Primrose	*Primula* sp./spp.		Primulaceae	Bartlett and Powell (1981); SPHD (2016); CABI (2019)
ornamental	Hawthorn	*Crataegus* spp.		Rosaceae	SPHD (2016); CABI (2019)
ornamental	Garden nasturtium	*Tropaeolum majus*		Tropaeolaceae	EPPO (2020)
non cultivated	Lilly of the Incas; Inca lily	*Alstroemeria* spp.		Alstroemeriaceae	Malipatil and Wainer (2005); SPHD (2016); CABI (2019)
non cultivated	Rag weed	*Ambrosia* spp.		Asteraceae	Malipatil and Wainer (2005); SPHD (2016); CABI (2019)
non cultivated		*Symphyotrichum novi-belgii*		Asteraceae	EPPO (2020)
non cultivated	Tree marigold	*Tithonia* spp.		Asteraceae	SPHD (2016); CABI (2019)
non cultivated	Yellow salsify; Goat's-beard	*Tragopogon* spp.		Asteraceae	SPHD (2016); CABI (2019)
non cultivated	Malabar spinach	*Basella* spp.		Basellaceae	SPHD (2016); CABI (2019)
non cultivated	Melilots; Sweet clover	*Melilotus* sp./spp.		Fabaceae	SPHD (2016); CABI (2019)
non cultivated	Mallow	*Malva* sp./spp.		Malvaceae	SPHD (2016); CABI (2019)

Host type	Common names	Scientific name	Variety	Family	Records
non cultivated	Cestrums; Jassamine	*Cestrum* spp.		Solanaceae	SPHD (2016); CABI (2019)
non cultivated	Bulrush; Reedmace	*Typha* spp.		Typhaceae	SPHD (2016); CABI (2019)

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Appendix 2: Leaf mining damage caused by established species

Table 16 provides a summary of established (native or naturalised) leaf mining species, their host overlap with American serpentine leafminer and features that can be used to differentiate between American serpentine leafminer and established leafminers.

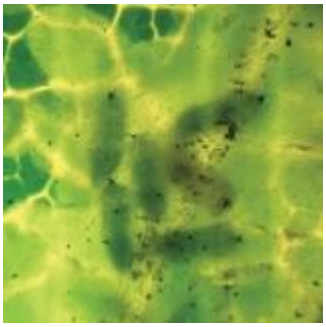
It should be noted that because host plant records are poor for many native or naturalised species, there could be many more affected hosts than included here. The host overlap between American serpentine leafminer and several native leafminer underscores the importance of sample collection and molecular diagnostics.

Figure 8 provides some general guidelines for differentiating between Dipteran and Lepidopteran leafminer larvae.

Table 16 Leaf mining damage caused by Australian dipterans or lepidopterans on leaves of cultivated crops and common weeds of cultivated crops

LEAFMINER	DISTINGUISHING FEATURES FROM L. *TRIFOLII* DAMAGE	HOSTS (THOSE SHARED WITH L. *TRIFOLII* ARE UNDERLINED)	PHOTO OF LEAF MINE
Liriomyza brassicae	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from American serpentine leafminer)	Brassicaceae Capparaceae Tropaeolaceae Fabaceae	
Liriomyza chenopodii	None (all life stages are morphologically indistinguishable in appearance, without dissection, and behaviour from American serpentine leafminer)	Amaranthaceae Caryophyllaceae Any native hosts are as yet unrecorded (*L. chenopodii* is a native species)	

LEAFMINER	DISTINGUISHING FEATURES FROM L. *TRIFOLII* DAMAGE	HOSTS (THOSE SHARED WITH L. *TRIFOLII* ARE UNDERLINED)	PHOTO OF LEAF MINE
Phytomyza syngenesiae	Pupation occurs inside the leaf mine.  Adults easily distinguishable from American serpentine leafminer	Apiaceae Asteraceae Fabaceae	
Tropicomyia polyphtya	Larvae are upper surface epidermal feeders (deeper parenchymatous tissues is not eaten). As such, mines have a silvery, film like appearance. Pupation occurs inside the mine and pupa is noticeably flattened.  Adults easily distinguishable from American serpentine leafminer	Highly polyphagous Common hosts include plants in the Passifloraceae, Euphorbiaceae, Fabaceae, Rubiaceae, Ruaceae, and Solanaceae	
Ophiomyia solanicola	Pupation occurs inside the leaf mine.  Adults easily distinguishable from American serpentine leafminer	Solanaceae (incl. egg plant)	

LEAFMINER	DISTINGUISHING FEATURES FROM L. *TRIFOLII* DAMAGE	HOSTS (THOSE SHARED WITH L. *TRIFOLII* ARE UNDERLINED)	PHOTO OF LEAF MINE
Ophiomyia alysicarpi	Mint green pupae can be found inside mines  Adults easily distinguishable from American serpentine leafminer	*Alysicarpus* sp. *Desmodium* sp.	
Ophiomyia cornuta	Adults easily distinguishable from American serpentine leafminer	*Scaevola* sp. *Goodenia* sp.	
Lepidopteran leafminer	Lepidopteran larvae can be distinguished from dipteran larvae on the spot, via a hand lens (Figure 10). For some lepidopteran species, when the larva is in its final stages, it forms a large blister at the end of the mine (see images on the right). American serpentine leafminer larvae do not create such blisters.	Polyphagous (incl. <u>Fabaceae</u>, Eucalypts, native trees, etc)	 

LEAFMINER	DISTINGUISHING FEATURES FROM L. *TRIFOLII* DAMAGE	HOSTS (THOSE SHARED WITH L. *TRIFOLII* ARE UNDERLINED)	PHOTO OF LEAF MINE
			

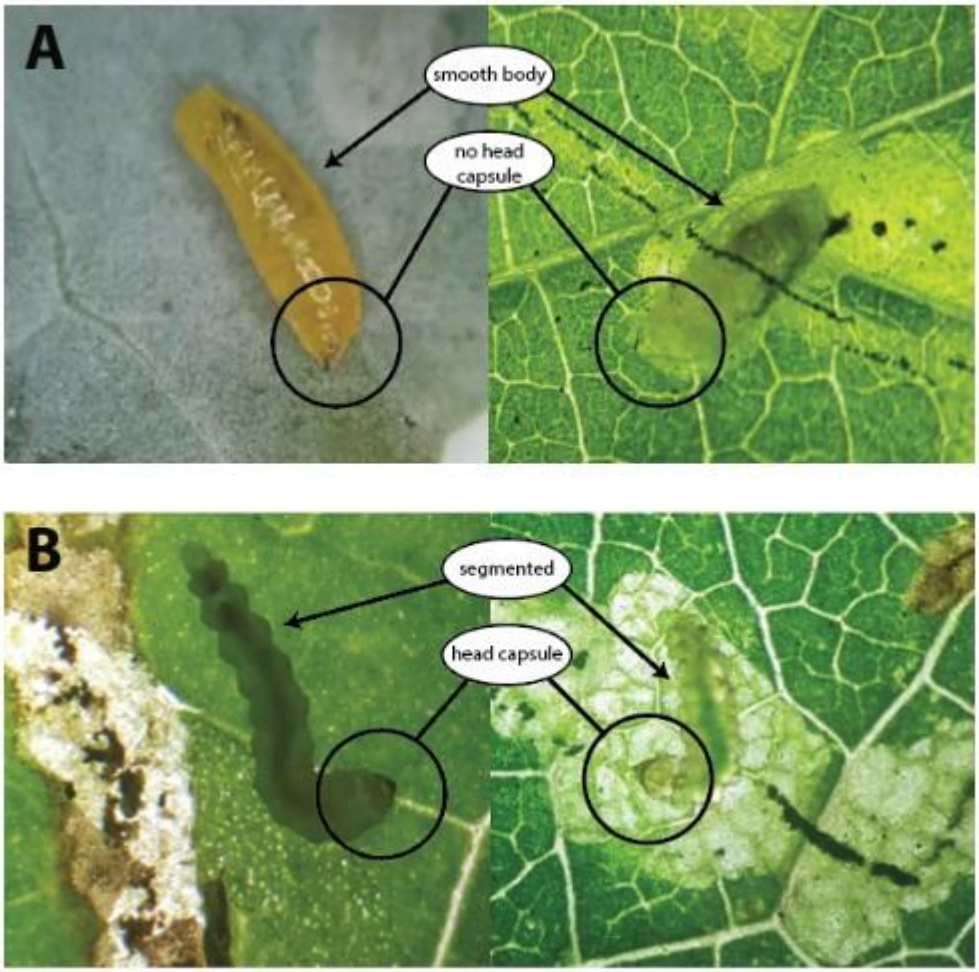


Figure 8 General guidelines for distinguishing dipteran vs lepidopteran leafminer larvae. Dipteran leafminer larvae (A) lack a head capsule and have a smooth body without clear segments. Lepidopteran leafminer larvae (B) have a distinct head capsule and are more visibly segmented. When attempting to distinguish a dipteran from lepidopteran leafminer larva on the spot, it is helpful to backlight the leaf as it is viewed under a hand lens, or to carefully excise the larva. Images: Elia Pirtle, Cesar Australia *Pty Ltd*

Surveillance for *L. trifolii* in Australia will be confounded by the presence of other leafmining insects already present in Australia, many of which create mines that are indistinguishable to a casual observer, and even in some cases indistinguishable to experts.

Table 17 lists genera that include hosts of *L. trifolii* but do not appear to contain of other agromyzids within Australasia. It is important to note that host plant records for non-pest leafminer flies are highly incomplete, and some of the genera included herein could host leafminer flies that have not yet been reported. Host records for weeds and native plants are expected to be more incomplete than for cultivated crops and ornamentals, and despite poor host records for native leafminers. We can be more confident that native leafminers are not currently a notable presence within the genera containing cultivated crops and ornamentals, where sampling effort is expected to be higher. As such, leafmining damage detected within the genera included in these tables should be regarded as highly suspicious.

Table 18 indicates where known overlap exists between hosts of *L. huidobrensis, L. trifolii* or *L. sativae* and native or naturalised agromyzids in Australia and Australasia.

Table 17 Host genera of L. huidobrensis, L. sativae or L trifolii (including only records with comprehensive or partial evidence for lifecycle completion in the field, see Appendix 1) without any reported overlap with native or naturalised agromyzids already present in Australia or Australasia (see Table 18 for known overlap). For each host, preferences of the three exotic Liriomyza are indicated (LH = L. huidobrensis; LT = L. trifolii; LS = L. sativae).

Genus	Family	Exotic *Liriomyza*	Host type
Allium	Amaryllidaceae	LH; LT; LS	commercial
Coriandrum	Apiaceae	LH; LT; LS	commercial
Eryngium	Apiaceae	LH	commercial
Petroselinum	Apiaceae	LT	commercial
Colocasia	Araceae	LH	commercial
Cichorium	Asteraceae	LH	commercial
Crassocephalum	Asteraceae	LH	commercial
Basella	Basellaceae	LH	commercial
Eruca	Brassicaceae	LH	commercial
Spinacea	Chenopodiaceae	LH	commercial
Benincasa	Cucurbitaceae	LH; LT; LS	commercial
Coccinia	Cucurbitaceae	LT	commercial
Lagenaria	Cucurbitaceae	LH; LT; LS	commercial
Luffa	Cucurbitaceae	LH; LT; LS	commercial
Momordica	Cucurbitaceae	LH; LT; LS	commercial
Sechium	Cucurbitaceae	LH	commercial
Trichosanthes	Cucurbitaceae	LT	commercial
Arachis	Fabaceae	LT	commercial
Cicer	Fabaceae	LH; LT; LS	commercial
Cyamopsis	Fabaceae	LT	commercial
Lens	Fabaceae	LS	commercial
Lotus	Fabaceae	LH	commercial
Lupin	Fabaceae	LH	commercial
Medicago	Fabaceae	LH; LT; LS	commercial
Trigonella	Fabaceae	LH; LT	commercial
Vicia	Fabaceae	LH; LT; LS	commercial
Mentha	Lamiaceae	LH; LS	commercial
Origanum	Lamiaceae	LS	commercial
Linum	Linaceae	LH	commercial

Genus	Family	Exotic *Liriomyza*	Host type
Abelmoschus	Malvaceae	LH; LT; LS	commercial
Corchorus	Malvaceae	LT	commercial
Gossypium	Malvaceae	LT; LS	commercial
Hibiscus	Malvaceae	LH; LT	commercial
Zizania	Poaceae	LH	commercial
Lycium	Solanaceae	LH	commercial
Thurnbergia	Acanthaceae	LH	ornamental
Celosia	Amaranthaceae	LH; LT	ornamental
Bupleurum	Apiaceae	LH	ornamental
Centella	Apiaceae	LH	ornamental
Oenanthe	Apiaceae	LH	ornamental
Catharanthus	Apocynaceae	LH	ornamental
Asclepias	Asclepiadaceae	LT	ornamental
Chionodoxa	Asparagaceae	LH	ornamental
Hemerocallis	Asphodelaceae	LH	ornamental
Argyranthemum	Asteraceae	LH	ornamental
Calendula	Asteraceae	LH; LT; LS	ornamental
Centaurea	Asteraceae	LH	ornamental
Conoclinium	Asteraceae	LH	ornamental
Cosmos	Asteraceae	LH	ornamental
Dimorphotheca	Asteraceae	LT	ornamental
Echinacea	Asteraceae	LT	ornamental
Eupatorium	Asteraceae	LT	ornamental
Felicia	Asteraceae	LS	ornamental
Flaveria	Asteraceae	LT	ornamental
Kalimeris	Asteraceae	LH	ornamental
Osteospermum	Asteraceae	LH	ornamental
Pyrethrum	Asteraceae	LH	ornamental
Tanacetum	Asteraceae	LH	ornamental
Zinnia	Asteraceae	LH; LT; LS	ornamental
Impatiens	Balsaminaceae	LH	ornamental
Nasturtium	Brassicaceae	LH; LS	ornamental
Calceolaria	Calceolariaceae	LH	ornamental
Campanula	Campanulaceae	LH	ornamental
Platycodon	Campanulaceae	LH	ornamental
Trachelium	Campanulaceae	LH; LT	ornamental
Dianthus	Caryophyllaceae	LH; LT	ornamental
Gypsophila	Caryophyllaceae	LH; LT; LS	ornamental
Bauhinia	Fabaceae	LS	ornamental
Lathyrus	Fabaceae	LH	ornamental
Lupinus	Fabaceae	LH; LS	ornamental
Eustoma	Gentianaceae	LH; LT	ornamental
Exacum	Gentianaceae	LH	ornamental
Lisianthus	Gentianaceae	LH; LT; LS	ornamental
Freesia	Iridaceae	LH	ornamental
Gladiolus	Iridaceae	LH	ornamental
Salvia	Lamiaceae	LH; LT	ornamental
Lilium	Liliaceae	LH	ornamental

Genus	Family	Exotic *Liriomyza*	Host type
Torenia	Linderniaceae	LH	ornamental
Althaea	Malvaceae	LH	ornamental
Moringa	Moringaceae	LS	ornamental
Godetia	Onagraceae	LH	ornamental
Oenothera	Onagroideae	LH	ornamental
Dendrobium	Orchidaceae	LS	ornamental
Papaver	Papaveraceae	LH	ornamental
Antirrhinum	Plantaginaceae	LH; LT; LS	ornamental
Myosotis	Plumbaginaceae	LH	ornamental
Lagurus	Poaceae	LH	ornamental
Phlox	Polemoniaceae	LH; LT; LS	ornamental
Primula	Primulaceae	LH	ornamental
Aquilegia	Ranunculaceae	LS	ornamental
Delphinium	Ranunculaceae	LH	ornamental
Nigella	Ranunculaceae	LH	ornamental
Prunus	Rosaceae	LS	ornamental
Rosa	Rosaceae	LH; LT	ornamental
Diascia	Scrophulariaceae	LH	ornamental
Viola	Violaceae	LH	ornamental
Sagittaria	Alismataceae	LH	non cultivated
Alternanthera	Amaranthaceae	LT	non cultivated
Amaranthus	Amaranthaceae	LH; LT; LS	non cultivated
Deeringia	Amaranthaceae	LH	non cultivated
Gomphrena	Amaranthaceae	LH	non cultivated
Iresine	Amaranthaceae	LH	non cultivated
Alocasia	Araceae	LH	non cultivated
Hydrocotyle	Araliaceae	LH; LT; LS	non cultivated
Artemisia	Asteraceae	LH; LT	non cultivated
Baccharis	Asteraceae	LT	non cultivated
Carduus	Asteraceae	LH	non cultivated
Dichrocephala	Asteraceae	LH	non cultivated
Echinops	Asteraceae	LH	non cultivated
Eclipta	Asteraceae	LT	non cultivated
Emilia	Asteraceae	LH	non cultivated
Erechtites	Asteraceae	LH; LT	non cultivated
Galinsoga	Asteraceae	LH; LT; LS	non cultivated
Helipterum	Asteraceae	LH	non cultivated
Hemistepta	Asteraceae	LH	non cultivated
Hymenopappus	Asteraceae	LT	non cultivated
Lipochaeta	Asteraceae	LS	non cultivated
Melanthera	Asteraceae	LT	non cultivated
Parthenium	Asteraceae	LT	non cultivated
Synedrella	Asteraceae	LH; LT	non cultivated
Tridax	Asteraceae	LT	non cultivated
Verbesina	Asteraceae	LS	non cultivated
Xanthium	Asteraceae	LT	non cultivated
Cordia	Boraginaceae	LT	non cultivated
Capsella	Brassicaceae	LH	non cultivated

Genus	Family	Exotic *Liriomyza*	Host type
Rorippa	Brassicaceae	LH	non cultivated
Silene	Caryophillaceae	LH; LT	non cultivated
Malachium	Caryophyllaceae	LH	non cultivated
Spergula	Caryophyllaceae	LT	non cultivated
Commelina	Commelinaceae	LT	non cultivated
Calystegia	Convolvulaceae	LH	non cultivated
Convolvulus	Convolvulaceae	LT	non cultivated
Pharbitis	Convolvulaceae	LH	non cultivated
Ceratosanthes	Cucurbitaceae	LS	non cultivated
Melothria	Cucurbitaceae	LH	non cultivated
Melilotus	Fabaceae	LH; LS	non cultivated
Poissonia	Fabaceae	LS	non cultivated
Lamium	Lamiaceae	LH	non cultivated
Leonurus	Lamiaceae	LH	non cultivated
Melissa	Lamiaceae	LS	non cultivated
Anoda	Malvaceae	LS	non cultivated
Malva	Malvaceae	LH; LS	non cultivated
Sida	Malvaceae	LH; LS	non cultivated
Oxalis	Oxalidaceae	LH; LT	non cultivated
Piriqueta	Passifloraceae	LT	non cultivated
Linaria	Plantaginaceae	LT	non cultivated
Veronica	Plantaginaceae	LH	non cultivated
Limonium	Plumbaginaceae	LH	non cultivated
Polygala	Polygalaceae	LT	non cultivated
Rumex	Polygonaceae	LH	non cultivated
Cardiospermum	Sapindaceae	LT	non cultivated
Nemesia	Scrophulariaceae	LH	non cultivated
Datura	Solanaceae	LH; LT; LS	non cultivated
Nicotiana	Solanaceae	LH; LT	non cultivated
Kallstroemia	Zygophyllaceae	LT	non cultivated
Tribulus	Zygophyllaceae	LT	non cultivated

Table 18 Host genera of either L. huidobrensis, L. sativae or L trifolii with known overlap with native or naturalised agromyzids already present in Australia or Australasia. Some of these species are stem miner, which should be straightforward to distinguish from the leaf mining damage caused by L. huidobrensis, L. sativae or L trifolii. Records of L. sativae host plants within the Torres Strait Islands (TSI) and on the Cape York Peninsula (CYP) are included.

Host genus	Host family	Australasian Agromyzid genera	Sources
Beta	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977); Spencer (1990)
Chenopodium	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Spinacia	Amaranthaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Apium	Apiaceae	*Melanagromyza*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Daucus	Apiaceae	*Phytomyza*	Spencer (1973)
Asparagus	Asparagaceae	*Hexomyza*	Benavent Corai et al. (2005)
Ageratum	Asteraceae	*Melanagromyza*	Spencer (1977)
Arctium	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Aster	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Bellis	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Bidens	Asteraceae	*Melanagromyza*	Spencer (1977)
Callistephus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Carthamus	Asteraceae	*Melanagromyza*	Benavent Corai et al. (2005)
Chrysanthemum	Asteraceae	*Ophiomyia, Phytomyza, Melanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Cineraria	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cirsium	Asteraceae	*Phytomyza*	Spencer (1977)
Conyza	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Coreopsis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Craspedia	Asteraceae	*Liriomyza*	Benavent Corai et al. (2005)
Crepis	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Cynara	Asteraceae	*Phytomyza*	Spencer (1973)
Dahlia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Erigeron	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005); Spencer (1977)
Gaillardia	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gerbera	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Gnaphalium	Asteraceae	*Melanagromyza, Malanagromyza*	Benavent Corai et al. (2005); Spencer (1977)
Helianthus	Asteraceae	*Calycomyza, Phytomyza*	Benavent Corai et al. (2005)
Helichrysum	Asteraceae	*Liriomyza, Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Lactuca	Asteraceae	*Liriomyza*, Phytomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1973)
Leucanthemum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Picris	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Senecio	Asteraceae	*Melanagromyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1977)
Solidago	Asteraceae	*Calycomyza*	Benavent Corai et al. (2005)
Sonchus	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005); Spencer (1977)
Tagetes	Asteraceae	*Liriomyza**	Present in TSI/CYP

Host genus	Host family	Australasian Agromyzid genera	Sources
Taraxacum	Asteraceae	*Phytomyza*	Benavent Corai et al. (2005)
Tecoma	Bignoniaceae	*Liriomyza**	Present in TSI/CYP
Barbarea	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Brassica	Brassicaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Cardamine	Brassicaceae	*Liriomyza*	Spencer (1977)
Diplotaxis	Brassicaceae	*Liriomyza*	Spencer (1977)
Hirschfeldia	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005)
Matthiola	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977)
Raphanus	Brassicaceae	*Liriomyza*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977)
Silene	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Stellaria	Caryophyllaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Cleome	Cleomaceae	*Liriomyza**	Benavent Corai et al. (2005); Present in TSI/CYP
Ipomoea	Convolvulaceae	*Melanagromyza*	Benavent Corai et al. (2005)
Citrullus	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucumis	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Cucurbita	Cucurbitaceae	*Liriomyza**	Present in TSI/CYP
Euphorbia	Euphorbiaceae	*Tropicomyia*	Spencer (1977)
Ricinus	Euphorbiaceae	*Liriomyza**	Present in TSI/CYP
Cajanus	Fabaceae	*Melanagromyza, Ophiomyia*	Spencer (1973); Spencer (1977)
Canavalia	Fabaceae	*Ophiomyia*	Spencer (1973)
Cassia	Fabaceae	*Tropicomyia, Ophiomyia*	Spencer (1977); Spencer (1990)
Crotalaria	Fabaceae	*Liriomyza*, Ophiomyia*	Present in TSI/CYP; Spencer (1973); Spencer (1977)
Dolichos	Fabaceae	*Ophiomyia*	Spencer (1973); Spencer (1977)
Glycine	Fabaceae	*Melanagromyza, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Indigofera	Fabaceae	*Liriomyza*, Ophiomyia*	Benavent Corai et al. (2005); Present in TSI/CYP; Spencer (1977); Spencer (1990)
Lablab	Fabaceae	*Ophiomyia*	Spencer (1973)
Macroptilium	Fabaceae	*Liriomyza*, Tropicomyia*	Present in TSI/CYP; Spencer (1977)
Millettia	Fabaceae	*Unknown*	Pirtle pers. comm.
unknown	Fabaceae	*Melanagromyza*	Spencer (1977)
Phaseolus	Fabaceae	*Ophiomyia, Tropicomyia, Melanagromyza, Ophiomyla*	Spencer (1973); Spencer (1977)
Pisum	Fabaceae	*Liriomyza, Phytomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1973); Spencer (1977)
Senna	Fabaceae	*Tropicomyia*	Spencer (1977)
Sesbania	Fabaceae	*Liriomyza**	Present in TSI/CYP
Trifolium	Fabaceae	*Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Vigna	Fabaceae	*Liriomyza*, Melanagromyza, Tropicomyia, Ophiomyia*	Present in TSI/CYP; Pirtle et al. (2020); Spencer (1973); Spencer (1977); Spencer 1977
Scaevola	Goodeniaceae	*Liriomyza, Ophiomyia*	Benavent Corai et al. (2005); Pirtle pers.

Host genus	Host family	Australasian Agromyzid genera	Sources
			comm.; Spencer (1977)
Hydrangea	Hydrangeaceae	*Tropicomyia*	Spencer (1977)
Ocimum	Lamiaceae	*Liriomyza**	Pirtle et al. (2020)
Stephania	Menispermaceae	*Tropicomyia*	Spencer (1977); Spencer (1990)
Passiflora	Passifloraceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977); Spencer (1990)
Turnera	Passifloraceae	*Liriomyza**	Pirtle et al. (2020)
Plantago	Plantaginaceae	*Liriomyza, Phytomyza*	Benavent Corai et al. (2005); Spencer (1977);
Hordeum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
unknown	Poaceae	*Agromyza, Cerodontha*	Spencer (1973)
Setaria	Poaceae	*Agromyza*	Benavent Corai et al. (2005)
Triticum	Poaceae	*Ceradontha, Pseudonapomyza*	Benavent Corai et al. (2005); Spencer (1973)
Zea	Poaceae	*Pseudonapomyza*	Spencer (1973)
Portulaca	Portulacaceae	*Liriomyza*	Benavent Corai et al. (2005)
Ranunculus	Ranunculaceae	*Phytomyza, Napomyza*	Spencer (1977); Spencer (1990)
Passiflora	Resedaceae	*Tropicomyia*	Spencer (1990)
Coffea	Rubiaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Citrus	Rutaceae	*Tropicomyia*	Spencer (1973); Spencer (1977)
Capsicum	Solanaceae	*Tropicomyia, Liriomyza**	Pirtle et al. (2020); Spencer (1977)
Cestrum	Solanaceae	*Tropicomyia*	Spencer (1977)
Lycopersicum	Solanaceae	*Liriomyza**	Present in TSI/CYP
Petunia	Solanaceae	*Liriomyza**	Present in TSI/CYP
Physalis	Solanaceae	*Liriomyza**	Present in TSI/CYP; Pirtle et al. (2020)
Solanum	Solanaceae	*Ophiomyia, Liriomyza**	Pirtle pers. comm.; Present in TSI/CYP; Spencer (1977)
Tropaeolum	Tropaeolaceae	*Liriomyza*	Benavent Corai et al. (2005); Spencer (1977); Spencer (1990)
Lantana	Verbenaceae	*Calycomyza, Tropicomyia, Ophiomyia*	Benavent Corai et al. (2005); Spencer (1977)
Stachytarpheta	Verbenaceae	*Liriomyza**	Pirtle et al. (2020)
Verbena	Verbenaceae	*Calycomyza*	Benavent Corai et al. (2005)

* Includes *Liriomyza sativae* within TSI/CYP

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Appendix 3: Permits for the control of American serpentine leafminer and other *Liriomyza* spp. on different host crops

Table 19 lists the commercial hosts of American serpentine leafminer (refer to Appendix 1) and notes where suitable pesticides are available and approved by the APVMA for control on each host crop. Blank cells denote where suitable pesticides have not yet been approved or identified. Refer to the APVMA permit database (https://portal.apvma.gov.au/permits) or the most up to date information on current leafminer permits.

Table 19 Insecticides control options for different crop groups

Relevant Crop group	Crop/host plant	Pesticide	Permit number(s)/ status	Target pest (as per permit)	Expiration date	Withholding information/notes	Suitable for management or eradication[3]
Brassica vegetables	Bok choy; pak-choy; turnip	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Broccoli	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Broccoli	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

[3] Some pesticides will be more suitable for eradication than for the ongoing management of the pest. This is because either they will need to be used at high rates to be effective, there are potential resistance issues, or the pesticide is not likely to fit into existing IPM systems as well as alternative pesticides. Other chemicals provide good control of the pest and are also compatible with IPM systems and will be more suitable for the ongoing management of the pest. To highlight this, pesticides are noted for either eradication, or for management and eradication.

Brassica vegetables	Brussels sprouts	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Brussels sprouts	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cabbage	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cabbage	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L. sativae*)	30 June 2024	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Brassica vegetables	Cauliflower	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Brassica vegetables	Cauliflower	Emamectin benzoate	PER87563	*Liriomyza* spp. (including*: L.*	30 June 2024	3 days (harvest) Do not graze or cut for	Management or eradication

				sativae)		stock food	
Brassica vegetables	Indian mustard	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Bulb vegetables	Garlic	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Garlic	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Leek	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	30 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Onions	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Bulb vegetables	Shallots	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Shallots	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Bulb vegetables	Spring onion	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Cereal grains	Barley	No suitable pesticides identified					
Fruit trees (non-bearing)	Any trees that will not bear fruit for the next 12 months	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Angled luffa, sing-kwa	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Cucumber	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Cucumber	Cyromazine	PER81867	*Liriomyza*	30 November	7 days (harvest)	Management or

vegetables (Cucurbits – including melons)				*sativae* and *L. huidobrensis*	2023	Do not graze or cut for stock food	eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Melon (including honeydew, rock melon)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. butternut)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits –	Bottle gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

including melons)							
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Pumpkin (e.g. Queensland blue)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:* *L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Smooth luffa, sponge gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting	Watermelon	Abamectin	PER81876	*Liriomyza*	30 April 2024	7 days (harvest)	Management or

vegetables (Cucurbits – including melons)				*sativae* and *L. huidobrensis*		Do not graze or cut for stock food	eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Watermelon	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Wax gourd	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (Cucurbits – including melons)	Zucchini	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (Cucurbits – including melons)	Zucchini	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Bell pepper	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables	Capsicum, chili*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*:*	31 December 2023	1 day (harvest) Do not graze or cut for	Management or eradication

(other than cucurbits)				*L. sativae, L trifolii, L. huidobrensis*)		stock food	
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Capsicum, chili*	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Eggplant	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Eggplant	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Fruiting vegetables (other than cucurbits)	Okra*	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication

Fruiting vegetables (other than cucurbits)	Okra*	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Okra*	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	1 day (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Fruiting vegetables (other than cucurbits)	Tomato	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Herbs	Basil	No suitable pesticides identified					
Herbs	Oregano	No suitable pesticides identified					

Herbs	Parsley[4]	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management
Head lettuce	Lettuce (Head type only)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Lettuce	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	1 day (harvest) Do not graze or cut for stock food	Management
Leafy vegetables (including brassica leafy vegetables)	Spinach	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Leafy vegetables (including brassica leafy vegetables)	Spinach	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	**Treated crop must be destroyed.** Treated crops must not be made available for human consumption Do not graze or cut for stock food	**Eradication only**
Leafy vegetables	Spinach and silverbeet	Chlorantraniliprole	PER87631	*Liriomyza* spp. (including*:*	30 June 2024	3 days (harvest) 7 days (grazing or	Management or eradication

[4] Note: Parsley is not a recorded host for *L. sativae* but may be a host of other *Liriomyza* spp. and is listed as one of the crops that the permit covers.

(including brassica leafy vegetables)				*L. sativae, L brassicae, L. huidobrensis*)		cutting for stock food)	
Legume vegetables	Common bean; French beans, kidney beans	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Common bean; French beans, kidney beans	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	7 days (harvest) 7 days (grazing and hay)	Management
Legume vegetables	Faba bean, broad bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Faba bean, broad bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Mung bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Pea, snow peas, sugar snap peas,	Spinetoram	PER87878	*Liriomyza* spp.	28 February 2023	3 days (harvest); 14 days (grazing and	Management or eradication

	field pea					hay) Do not allow dairy cattle to graze treated forage	
Legume vegetables	Pea, snow peas, sugar snap peas, field pea	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest); 3 days (grazing and hay)	Management
Legume vegetables	Red bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Red bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Legume vegetables	Snake bean	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Not classified	Field mustard	No suitable pesticides identified					
Not classified	Lucerne	No suitable pesticides identified					
Nursery stock (non-food)	Various non-food nursery plants (e.g. chrysanthemum etc.)	Cyromazine	PER83506	Larvae of leafminers	31 October 2022	Do not ship within 7 days of treatment	Management or eradication
Nursery stock	Seedlings, tubes	Abamectin	PER88977	Leafminers	30	Not required when	Management or

(non-food)	and plugs, potted colour, trees and shrubs, foliage plants, palms, grasses, fruiting plants (non-bearing), cut flowers and ornamentals.	Azadirachtin Cyromazine Emamectin Chlorantraniliprole + thiamethoxam Cyantraniliprole Indoxacarb Spinetoram		(*Liriomyza* spp.) including Vegetable leafminer (*Liriomyza sativae*)	November, 2022	used as directed	eradication
Oilseeds	Cotton	Spinetoram has been identified as suitable for the control of Vegetable leafminer and other *Liriomyza* spp. on cotton as overseas and Australian use patterns are comparable. However, no permit has been applied for this use to date					
Pulses	Pulses	Dimethoate	PER89184	*Liriomyza sativae, Liriomyza trifolii, Liriomyza huidobrensis*	31 March 2025	Harvest: Do not harvest for 14 days after application. Grazing: Do not graze or cut for stock food for 14 days after application.	Management
Root and tuber vegetables	Beets (beetroot, silver beet, sugar beets)	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Beets (beetroot, silver beet, sugar beets)	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Carrot	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Carrot	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber	Potato	Abamectin	PER81876	*Liriomyza sativae* and	30 April 2024	14 days (harvest) Do not graze or cut for	Management or eradication

vegetables				*L. huidobrensis*		stock food	
Root and tuber vegetables	Potato	Cyantraniliprole	PER90387	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 December 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Potato	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Root and tuber vegetables	Radish	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	14 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Radish	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication
Stalk and stem vegetables	Celery	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Celery	Spirotetramat	PER88640	*Liriomyza* spp. (including*: L. sativae, L trifolii, L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[5]	Abamectin	PER81876	*Liriomyza sativae* and *L. huidobrensis*	30 April 2024	7 days (harvest); do not graze	Management or eradication

[5] Note: Rhubarb is not a recorded host for *L. sativae or L. huidobrensis* but may be a host of other *Liriomyza* spp. and is part of the 'Stalk and Stem Vegetable Crop Group' that the permit covers.

Stalk and stem vegetables	Rhubarb[3]	Cyromazine	PER81867	*Liriomyza sativae* and *L. huidobrensis*	30 November 2023	7 days (harvest) Do not graze or cut for stock food	Management or eradication
Stalk and stem vegetables	Rhubarb[3]	Spirotetramat	PER88640	*Liriomyza* spp. (including*:* *L. sativae,* *L trifolii,* *L. huidobrensis*)	31 May 2023	3 days (harvest) Do not graze or cut for stock food	Management or eradication

Appendix 4: Additional impact prediction outputs

Table 20 State level accumulated unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. trifolii *at key entry points across Australia. Simulations were replicated 10 times with means and standard deviations shown in parentheses. The proportion crop impact was fixed at 10% in order to explore variability due to incursion location and the size and distribution of different industries. Thus, host preferences of the pests do not influence the predicted crop impacts, and impacts of low preference hosts may be overestimated while impacts of high preference hosts may be underestimated. All cells with a value of 0.00 represent unmitigated impacts less than $10,000.*

	INCURSION LOCATION														
	NSW		**NT**		**QLD**			**SA**	**TAS**	**VIC**		**WA**			
Crop	**Griffith**	**Sydney**	**Darwin**	**Katherine**	**Bowen**	**Bundaberg**	**Toowoomba**	**Adelaide**	**Devonport**	**Ballarat**	**Melbourne**	**Albany**	**Carnarvon**	**Geraldton**	**Perth**
Beans	0.00 (0.00)	0.05 (0.00)	0.06 (0.00)	0.00 (0.00)	7.56 (0.66)	9.62 (0.56)	9.23 (0.31)	0.00 (0.00)	1.18 (0.00)	0.31 (0.15)	0.39 (0.09)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.02 (0.00)
Broccoli	0.11 (0.00)	0.10 (0.00)	0.00 (0.00)	0.00 (0.00)	0.34 (0.30)	4.33 (0.41)	6.34 (0.22)	0.38 (0.05)	0.38 (0.00)	6.86 (1.80)	8.62 (0.27)	0.07 (0.02)	0.00 (0.00)	0.00 (0.00)	2.24 (0.46)
Brussels sprouts	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.05 (0.02)	0.07 (0.03)	5.01 (0.05)	0.31 (0.00)	0.38 (0.23)	0.82 (0.39)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)
Cabbages	0.00 (0.00)	2.19 (0.11)	0.00 (0.00)	0.00 (0.00)	0.23 (0.19)	2.66 (0.26)	3.91 (0.04)	0.26 (0.01)	0.12 (0.00)	0.70 (0.16)	0.87 (0.03)	0.00 (0.01)	0.00 (0.00)	0.00 (0.00)	0.58 (0.07)
Capsicum	0.14 (0.00)	0.14 (0.01)	0.00 (0.00)	0.00 (0.00)	9.24 (0.03)	3.87 (0.16)	0.76 (0.21)	2.59 (0.40)	1.47 (0.00)	0.19 (0.14)	0.33 (0.15)	0.08 (0.05)	0.00 (0.00)	0.00 (0.00)	0.68 (0.11)
Carrots	0.07 (0.00)	0.18 (0.03)	0.00 (0.00)	0.00 (0.00)	1.05 (0.87)	6.14 (0.46)	7.12 (0.60)	3.45 (0.20)	3.75 (0.00)	0.21 (0.09)	0.29 (0.03)	0.04 (0.06)	0.00 (0.00)	0.00 (0.01)	5.75 (1.17)
Cauliflowers	0.00 (0.00)	0.40 (0.03)	0.00 (0.00)	0.00 (0.00)	0.08 (0.09)	1.56 (0.15)	2.45 (0.07)	0.56 (0.06)	0.97 (0.00)	3.56 (1.00)	4.35 (0.13)	0.05 (0.01)	0.00 (0.00)	0.00 (0.00)	0.90 (0.11)
Flowers	0.00 (0.00)	16.57 (0.62)	0.18 (0.00)	0.00 (0.00)	0.51 (0.31)	4.91 (0.16)	4.17 (0.24)	2.04 (0.14)	0.63 (0.00)	4.18 (2.23)	7.59 (1.07)	0.24 (0.08)	0.00 (0.00)	0.00 (0.01)	1.84 (0.23)
Lettuces	0.00 (0.00)	1.18 (0.06)	0.00 (0.00)	0.00 (0.00)	0.38 (0.32)	5.96 (0.52)	8.30 (0.09)	0.93 (0.08)	0.01 (0.00)	7.38 (2.31)	9.85 (0.45)	0.04 (0.06)	0.00 (0.00)	0.00 (0.01)	3.26 (0.39)
Melons	3.03 (0.00)	0.04 (0.00)	0.61 (0.02)	0.00 (0.00)	4.55 (0.03)	4.50 (0.04)	0.73 (0.16)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.80 (0.30)

	INCURSION LOCATION														
Nurseries	2.39 (0.00)	20.32 (1.26)	0.28 (0.00)	0.00 (0.00)	3.45 (1.50)	19.88 (0.99)	21.13 (1.37)	5.85 (0.10)	0.70 (0.02)	10.83 (3.42)	17.10 (1.64)	0.18 (0.27)	0.00 (0.00)	0.01 (0.04)	8.31 (0.58)
Onions	0.29 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.49 (0.34)	3.77 (0.26)	4.86 (0.25)	3.92 (0.41)	3.74 (0.00)	0.11 (0.06)	0.19 (0.02)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	1.60 (0.68)
Peas	0.00 (0.00)	0.02 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.45 (0.01)	0.06 (0.01)	0.00 (0.00)	0.64 (0.00)	0.17 (0.12)	0.20 (0.06)	0.02 (0.00)	0.00 (0.00)	0.00 (0.00)	0.01 (0.00)
Potatoes	7.18 (0.00)	1.17 (0.08)	0.00 (0.00)	0.00 (0.00)	0.24 (0.13)	5.25 (0.20)	3.25 (0.24)	3.46 (0.20)	5.99 (0.00)	3.19 (0.36)	1.66 (0.28)	0.02 (0.03)	0.00 (0.00)	0.00 (0.00)	2.39 (0.73)
Pumpkins	2.03 (0.00)	0.16 (0.01)	0.00 (0.00)	0.00 (0.00)	1.86 (0.17)	2.90 (0.17)	3.33 (0.20)	0.00 (0.00)	0.14 (0.00)	0.01 (0.00)	0.01 (0.00)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.24 (0.11)
Tomatoes	0.83 (0.00)	1.81 (0.06)	0.01 (0.00)	0.00 (0.00)	17.76 (0.14)	11.68 (0.35)	3.34 (0.41)	1.10 (0.28)	0.34 (0.00)	0.54 (0.38)	0.98 (0.66)	0.00 (0.00)	0.00 (0.00)	0.00 (0.00)	0.09 (0.06)
Total	**16.07 (0.00)**	**44.35 (1.85)**	**1.14 (0.02)**	**0.00 (0.00)**	**47.75 (4.55)**	**87.54 (3.67)**	**79.05 (3.32)**	**29.55 (1.24)**	**20.36 (0.02)**	**38.61 (10.25)**	**53.24 (3.81)**	**0.76 (0.54)**	**0.00 (0.00)**	**0.02 (0.07)**	**28.71 (4.59)**

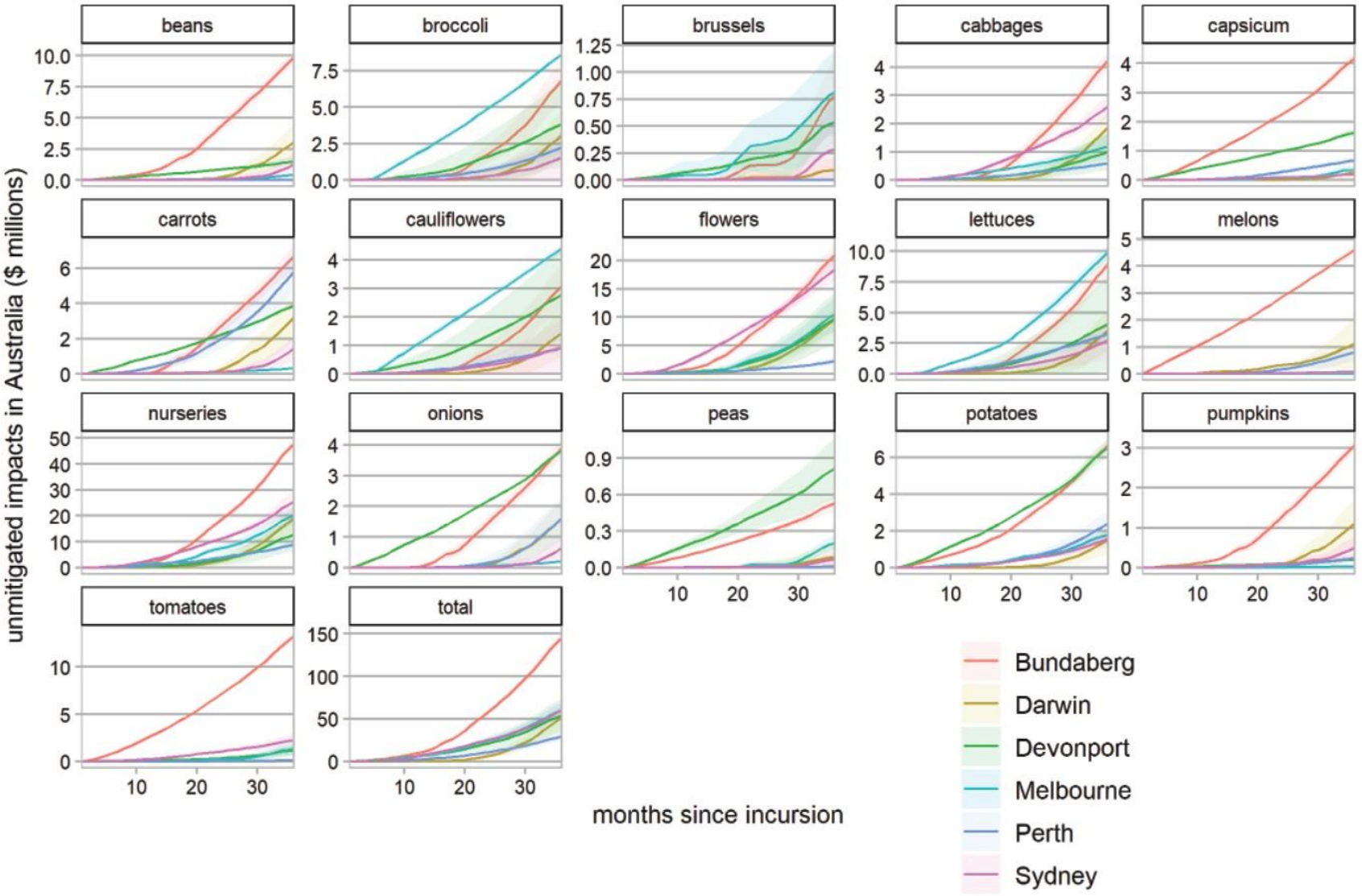


Figure 9 Australia wide accumulating unmitigated impacts in millions of dollars after 3 years resulting from a spring (September) incursion of L. trifolii *at key entry and establishment points across Australia.*

Appendix 13: Monitoring and evaluation data

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

Madeleine Quirk [1]

[1] AUSVEG

December 7, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

Industry monitoring data:

In a webinar aimed at Gippsland producers, knowledge change was measured following the webinar (Figure 1). Knowledge change was evident with eight attendees stating that their knowledge had increased moderately as a direct result of the information provided, while only four attendees had 'some' increase and two attendees had 'minimal' increase.

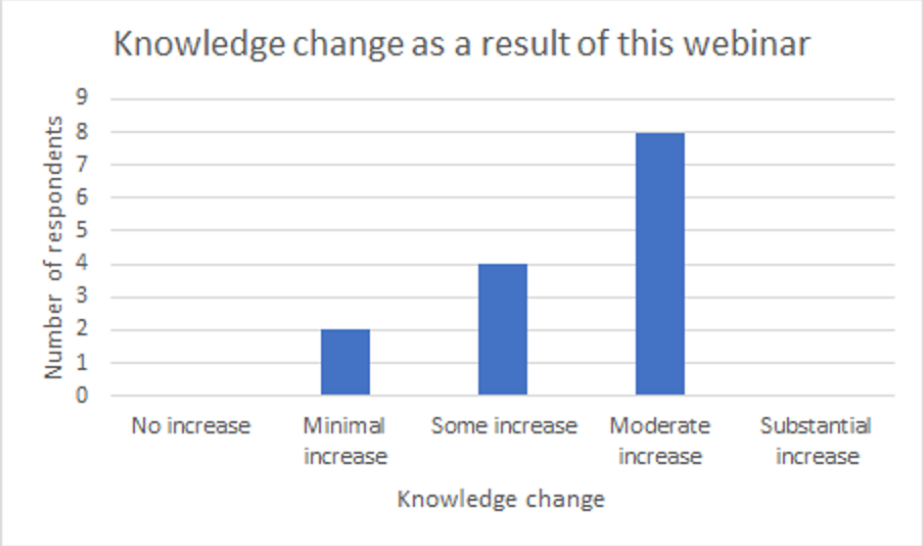


Figure 1. Knowledge change of exotic pests, including exotic leafminers (Liriomyza spp.), following the Gippsland-specific webinar

In a webinar aimed at Queensland producers, knowledge change was measured following the webinar (Figure 2). Knowledge change was evident with 16 attendees stating that their knowledge had increased substantially, 14 attendees stating that their knowledge had increased moderately, and four attendees stating their knowledge had increased to some extent.

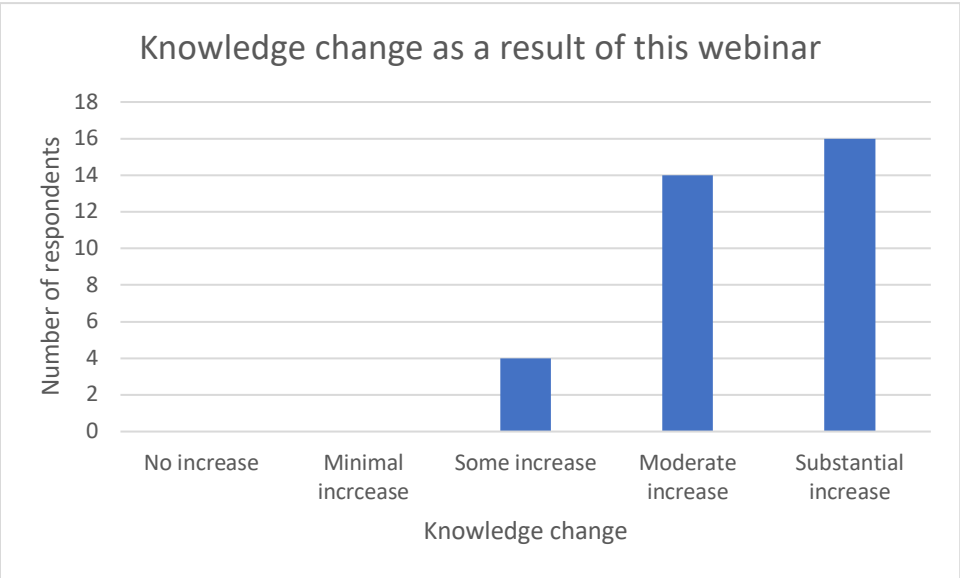


Figure 2. Knowledge change of plant pests, including exotic leafminers (Liriomyza spp.), following the Queensland-specific webinar

Table 1 below outlines monitoring and evaluation data collected during workshops in Ayr, Bowen, and Greater Sydney in 2019, covering all three *Liriomyza* spp. The data was collected prior to the presentation

and indicated that participants had differing levels of understanding of vegetable leafminer. In Ayr, six attendees were already well informed about VLM before the workshop, whereas in Bowen only five attendees were well informed about VLM, and in Greater Sydney only one attendee was well informed about VLM.

Table 1. Industry knowledge of exotic leafminers. Data collected in three Australian horticultural production regions, covering all three *Liriomyza* spp.

Level of understanding of exotic leafminers before presentation	Location: Ayr, QLD 2019	Location: Bowen, QLD 2019	Location: Greater Sydney, NSW 2019
I am well informed about exotic leafminers	6	5	1
I know a little bit about exotic leafminers	1	8	8
I have heard of exotic leafminers but I don't know much about them	10	0	20
I have never heard of exotic leafminers	1	1	0

Community monitoring data:

Community impact data was also collected by examining the suspected exotic leafminer reports made to the Victorian government by community members (see table 2). These reports, given their relatedness to the project, were forwarded to the project team. Seven reports were submitted based on information coming from the project webinar, the Department of Agriculture, Water and the Environment's website, or through MyPest Guide reporter. In most cases, reports were *Phytomyza syngenesiae* and in one case, the pest was not an agromyzid.

Table 2. Reports of suspected exotic leafminers (*Liriomyza* spp.) by Melbourne residents, the reason for reporting, and the outcome of the report.

Who	Location	Date	Reason for reporting	Crop	Outcome
Ag Vic staff	Attwood, VIC	August 2020	Viewed project webinar and followed up with Elia Pirtle.		Sent subsequent reports to Elia
Melbourne resident	St Kilda, VIC	August 2020	Reading DAWE website on exotic leafminers and reported to EPPH	Leafmining in agyranthums, everlastings, cosmos seedlings	*Phytomyza syngenesiae*
Melbourne resident	N/A	Sept 2020	Emailed plant.protection because it looked suspicious and he knew he had to report it. Took a	Broccolini	Samples sent to AgriBio to confirm it wasn't *P. horticola* in broccolini. It turns out that it was *P.*

			sample and put it in the fridge.		*syngenesiae* (not typical for broccolini)
Melbourne resident	N/A	Sept 2020	N/A	Leafmining in fig tree suspected to be leafminer fly	Not an agromyzid pest
Melbourne resident	Melbourne, VIC	Oct 2020	Emailed plant.protection because it looked suspicious and he knew he had to report it. Took a sample and put it in the fridge.	Mining on lettuce	*P. syngenesiae*
Melbourne resident	Brunswick, VIC	Oct 2020	My Pest Guide Reporter	Lettuce	*P. syngenesiae*
Melbourne resident	Camberwell, VIC	Oct 2020	Reading the DAWE website and decided to report	Leafmines in lettuce, marigold, sweet peas, tomatoes, cornflower, dahlias	Lettuce affected by *P. syngenesiae*, broccoli by *L. brassicae*, sweet peas *Ophiomya* spp. Requested images of tomatoes → outcome was that they were not leafmines.

TSI and CYP monitoring data:

A tourist survey was undertaken in 2018 to gauge the level of biosecurity awareness of short-term visitors to CYP, find out where they source biosecurity information before travelling, and determine the origins of these tourists. Table 3 below demonstrates the results of this survey.

Table 3. Tourist data collected in Seisia during the 2018 engagement trip

Question		
Are you a domestic or international visitor?	Domestic	International
	95%	5%
Have you ever heard of VLM?	Yes	No
	15%	85%
How did you find out about VLM? (If answered yes)	Landline, TAFE, TV, Online, employment at QDAF	
Has your new knowledge raised your awareness of the importance of biosecurity on CYP and TSI?	Yes	No
	97%	3%

Monitoring data was collected from rangers at the 2019 workshop. The training activities undertaken had demonstrably increased ranger awareness of VLM, SLM and ASLM. Table 4 below provides details of the key feedback received from rangers at this workshop.

Table 4. Summary of feedback received from rangers at the *Liriomyza* workshop held in 2019 on Thursday Island

Monitoring and evaluation question	Summary of feedback
How much time do you spend searching for or managing VLM?	In general, the NAQS rangers expressed that they were managing a large number of pests and diseases (both plant and animals, including aquatic threats). Therefore, the majority of rangers were relatively unaware of VLM. Some rangers noticed mining in their home gardens, but the amount of mining differed between islands.
Can the MT16004 project assist with your work?	In general, NAQS rangers stated that the project has assisted with their learning of exotic *Liriomyza*. The group were given resources including a PowerPoint presentation, TSI and CYP management guides and parasitoid wasp plans, and other resources to use to train new staff.

Images taken during TSI and CYP engagement trips



Figure 3. 2018 field trip meeting at the Coen Biosecurity Center. Image credit: Dr. Jessica Lye



Figure 4. In preparation for student visits to the community garden on Thursday Island in 2019. Image credit: Madeleine Quirk

Appendix 14: Communications and engagement update following Covid-19

A report prepared for Horticulture Innovation Australia, as part of Project MT16004 (*RD&E program for control, eradication and preparedness for vegetable leafminer*)

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[1] AUSVEG

December 8, 2020

The project *RD&E program for control, eradication and preparedness for vegetable leafminer* (MT16004) has been funded by Hort Innovation using the vegetable, nursery, potato, and melon research and development levies and contributions from the Australian Government. Hort Innovation is the grower-owned, not-for-profit research and development corporation for Australian horticulture.

During the final reporting period of the project, the project extension officer met and exceeded all output requirements in line with the project agreement with Cesar Australia. This is significant, given the impact that the covid-19 pandemic has had on business operations in 2020.

Extension

In early 2020, extension trips were undertaken in Darwin and Katherine (NT) and Bundaberg (QLD). During these trips, the project officer facilitated workshops and completed grower visits. The intention was to follow these extension trips with trips to Western Australia, Tasmania, Adelaide, Melbourne, and the Lockyer Valley, but the restrictions that were imposed in mid-March meant that these trips needed to be cancelled. Shortly afterwards, the project officer updated the communications and engagement plan to reflect the inability to travel. With approval of the C&E plan by Cesar Australia and Hort Innovation, AUSVEG moved towards online methods of engagement to communicate the relevant research and development outcomes. Webinars were delivered to target Melbourne, Gippsland, Western Australia, Tasmania, and Queensland (including Lockyer Valley). In addition to regional-specific webinars, an overall project-specific webinar was facilitated and was well received by industry. Direct calls and targeted engagement activities were also undertaken in the Adelaide and Lockyer Valley regions.

Evidence from the eight webinars suggests that vegetable, potato, melon and nursery growers value live webinars but they also value the video and audio resources that are developed using content from these webinars. The project has had high uptake in the way of views and link clicks following these engagement activities (see Monitoring and Evaluation section of final report).

Communications

During the final reporting period, communications requirements were followed in line with the C&E plan. An article was written for *Vegetables Australia* titled '*How parasitoid wasps are helping to fight the leafminer battle*', five articles were written as AUSVEG Front Line e-Bulletins, and 11 weekly update articles were developed. A project update was also developed for the Lockyer Valley Growers Association Bulletin, and an article called '*R&D Spotlight: keep an eye out for exotic leafminers*' was written for the AUSVEG SA Bulletin, an article titled '*Keep an eye out for the vegetable leafminer*' was written for WA Grower magazine, and a VLM update was included in the Bundaberg *Fresh Pickings* e-magazine. '*How parasitoid wasps are helping to fight the leafminer battle* was reproduced in Your Levy at Work (Greenlife Industry Australia bulletin), the Greenlife Industry Australia Nursery Network News, and Melon News. AUSVEG also updated the vegetable leafminer speed drawing to incorporate *Liriomyza huidobrensis* and *Liriomyza trifolii*. Social media was continually utilised throughout the course of the year to promote research, development and extension activities.