Department of Primary Industries

Biological control of weeds

A practitioner's guide for south-east Australia

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2021

Invasive Species Biosecurity
www.dpi.nsw.gov.au

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Disclaimer:
The information contained in this publication is based on knowledge and understanding at the time of writing (July 2021). However, because of advances in knowledge, users are reminded of the need to ensure that the information upon which they rely is up to date and to check the currency of the information with the appropriate officer of the Department of Regional NSW or the user's independent adviser.

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Foreword

I take great pleasure in presenting the manual 'Biological control of weeds – a practitioner's guide for south-east Australia'.

My sincere congratulations go to the NSW Department of Primary Industries (Weed Research Unit) which has produced an outstanding body of work that will contribute to the improved management of weeds throughout this region.

Weeds pose a significant threat to Australia's environment, biodiversity and primary production and negatively affect most aspects of human life. The cost of weeds to agriculture in Australia through lost production and management was estimated to be more than $6 billion in 2016, with a similar economic value being estimated for their impact on the environment and ecosystem services.

Many weeds can be successfully controlled using biological control. Biological control provides long-term solutions that are cost-efficient, effective, self-sustaining and environmentally friendly.

Australia has a long and distinguished record of successful weed biological control. Nearly 100 years ago prickly pear was successfully controlled using the Cactoblastis moth and a cochineal insect. This program generated a benefit: cost ratio of $313: $1. Australia has continued to develop many biocontrol programs, with over 70 programs being conducted from 1903 to 2019. Biological control agents were released for more than 50 of these programs, following rigorous scientific evaluation both overseas and in Australian quarantine facilities.

Weed professionals, land managers and the general community require the latest best practice information if they are to effectively implement successful weed biological control programs. This best practice is often generated and refined by both researchers and practitioners. Unfortunately, the best practice methodology required to implement successful weed biological control programs is often not available to the everyday land manager.

This best practice manual addresses common weed biological control questions and methodology issues and provides the key steps for undertaking weed biological control programs. The manual also provides a comprehensive overview of the biological control recommendations for more than 50 weed species. It provides detailed information about biological control agents, including their life cycle, impact and abundance; and how to collect, rear and monitor them.

I commend this book to weed professionals, land managers and the community as a valuable resource for many years to come. The information and recommendations therein should both inspire and enable those wishing to pursue biological control as a preferred option for dealing with invasive weed species.

Ultimately, an increase in successfully implemented weed biological control programs will lead to an improvement in Australia's environment, biodiversity, agricultural productivity and quality of life.

Royce Holtkamp
Chair, NSW Weed Biocontrol Taskforce
Horizon Ecological Consulting

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Introduction

Weeds and their impact

Some plants become weeds, and the reasons for this are varied. One reason suggests that plants that come from other geographic locations may become weeds because they lack control by their co-evolved suite of natural enemies. These natural enemies (e.g. insects, mites and pathogens) in their native range would ordinarily keep the plant in balance through actions such as herbivory or infection. If we can restore the balance between a weed and its natural enemies in the introduced range, it offers us a management solution, referred to as biological control (biocontrol), to reduce the impact of the weed.

Weeds adversely affect the Australian economy through agricultural, environmental, human health and amenity impacts. The financial impact of weeds to agriculture in 2016 was estimated to be in excess $6 billion per annum in lost productivity and associated control costs (Llewellyn *et al.*, 2016). The total cost of weeds to the natural environment is difficult to calculate, but is expected to be of similar or greater magnitude to agricultural costs.

P. Sullivan

Water flow impeded by invasion of alligator weed in the Hawkesbury-Nepean River, New South Wales.

P. Sullivan

Strong competition by Scotch broom infestation at Barrington Tops, New South Wales, displacing fragile native ecosystems.

P. Sullivan

Water quality and recreational activities negatively affected by water hyacinth.

Introduction

Managing weeds

Weeds can be managed using a variety of tools including: mechanical, chemical, cultural and or biological control approaches. Each of these has strengths and weaknesses. Therefore, we often use different tools in combination to enhance overall weed management as part of an integrated weed management program.

Biocontrol is playing an increasingly important role in managing weeds in Australia today. It is a technology that has proven to be an environmentally-friendly, self-perpetuating and cost-effective solution to many weed invasions.

This manual was written to assist weed practitioners interested in utilising and maximising the benefits of weed biocontrol as part of their weed management plans in south-east Australia.

What information is provided?

This manual provides key steps for undertaking weed biocontrol programs for more than 50 weed species in south-east Australia. It provides information on:

- weeds and their background
- how to identify biocontrol agents (the weed's natural enemies) and their potential impact on the weed
- how to source biocontrol agents
- how to redistribute these agents
- how to monitor establishment and dispersal of such agents.

Where does the information come from?

The information contained in this manual has been sourced from published literature and the experience of national biocontrol practitioners. It is acknowledged that our current understanding of best practice management is not the final word and that best practice will continue to evolve over time.

P. Sullivan

Negative economic returns from gorse invasion of a commercial pine plantation.

P. Sullivan

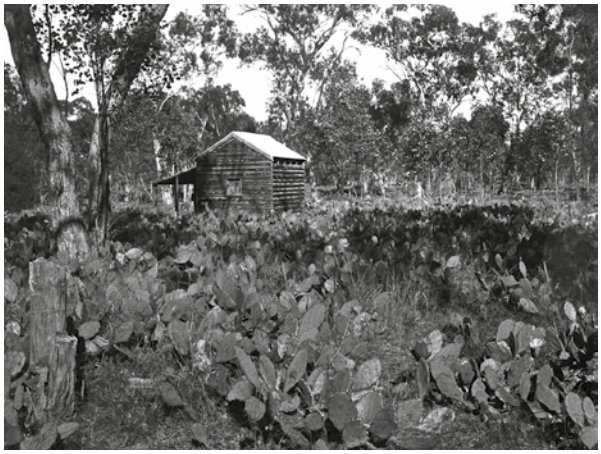
Gorse invasion within a national park.

The Australian biocontrol story

For over 100 years, Australians have been involved in biocontrol programs with more than 70 introduced plant species and have released more than 200 agents (McFadyen, 2008; McFadyen, 2011; Julien *et al*., 2012a; McFadyen, 2012a; Winston *et al*., 2014). The first and most famous biocontrol program in Australia began in 1925 with the successful control of prickly pears (*Opuntia* spp.). In the early 1900s, prickly pears were spreading at an annual rate of 400,000 ha per year. In total, prickly pears invaded an area of 25 million ha at their peak (an estimated combined area of England, Scotland and Wales), having significant impacts on agriculture and the environment. Despite tremendous efforts to mechanically and chemically manage prickly pears, the control costs were calculated to be of greater value than that of the land. As a result, many farmers abandoned their land.

In 1920, the Commonwealth Government, in conjunction with the New South Wales and Queensland state governments, set up a joint prickly pear board to investigate all options of control, including biocontrol. After many years of research, 20 biocontrol agents were approved for release and success was eventually found in the form of two insect agents: the Cactoblastis moth (*Cactoblastis cactorum*) and a cochineal (*Dactylopius opuntiae*). The results were outstanding, with good levels of control being achieved in less than six years.

The return on investment for this program was estimated at $312 for every dollar invested (Page and Lacey, 2006). This equates to a net value (in 2005 terms) in productivity gains of $3.1 billion (Page and Lacey, 2006). This program is still regarded as one of the most spectacular successes in weed biocontrol globally. Australia has since gone on to be one of the global leaders in weed biocontrol, providing solutions for many agricultural and environmental weeds. On average, financial returns on investment for all Australian weed biocontrol programs up until 2005 was 23:1 (Page and Lacey, 2006).



John Oxley Library, Queensland

Abandoned property overtaken by prickly pear, Chinchilla, Queensland, May 1928 (top). Reclaimed property, 17 months post-control using the Cactoblastis moth, Chinchilla, Queensland, October 1929 (bottom).

P. Sullivan

Effective control of wheel cactus with a cochineal biocontrol agent.

Introduction

How does biocontrol work?

Biocontrol aims to reduce a weed's population to a level where it no longer creates a problem. It is a long-term commitment. Control usually takes several years, sometimes decades, to be achieved and will not result in the eradication of the target weed. Biocontrol agents are selected based on their host specificity to the target weed (i.e. will only impact the intended target weed) and as a result, are limited by the presence of the weed (Figure 1).

There are very rare cases where weed biocontrol agents have changed their host plant affinities after their release to include plants other than those recognised to be acceptable hosts. Agents have evolved with their host plants over millions of years to feed and develop only on a particular plant species or a group of closely related species. If the weed is greatly reduced by the biocontrol agent, then the population of the biocontrol agent will fall to lower levels.

Is biocontrol safe?

Biocontrol agents undergo the strictest testing through the safeguards of Australia's *Biosecurity Act 2015* to ensure that they only attack the target weed before being released into the environment. To meet the requirements of this Act and Australia's *Environment Protection and Biodiversity Conservation Act 1999*, biocontrol agents must be subject to risk analyses to determine their approval for release in Australia. As part of the risk analysis, potential agents undergo strict host-specificity testing in quarantine conditions before consideration of their suitability for release.

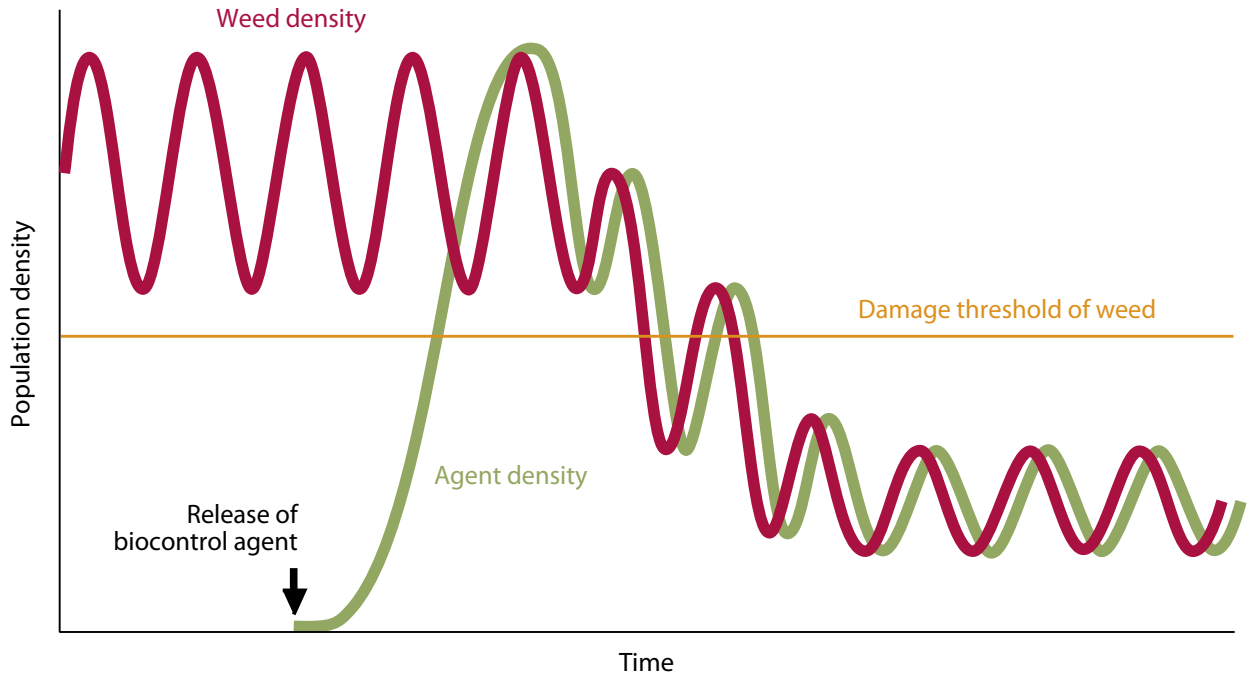


Figure 1. *The relationship between a weed and its biocontrol agent illustrating the critical point at which successful control is achieved (adapted from Briese 2000).*

Several steps are required before biocontrol agents are released into the environment. First, the weed species proposed for biocontrol research must be endorsed through a national committee called the Environment and Invasives Committee. Later, for any agent to be approved for release, each must demonstrate a very low or negligible risk to the environment via risk analysis undertaken by the Department of Agriculture, Water and the Environment in accordance with both Commonwealth Acts. The analysis is extensive, and includes information pertaining to host-specificity testing results, ensuring that the proposed agent is specific to the target, as well as information gleaned form wide consultation with technical, scientific experts and interested stakeholders.

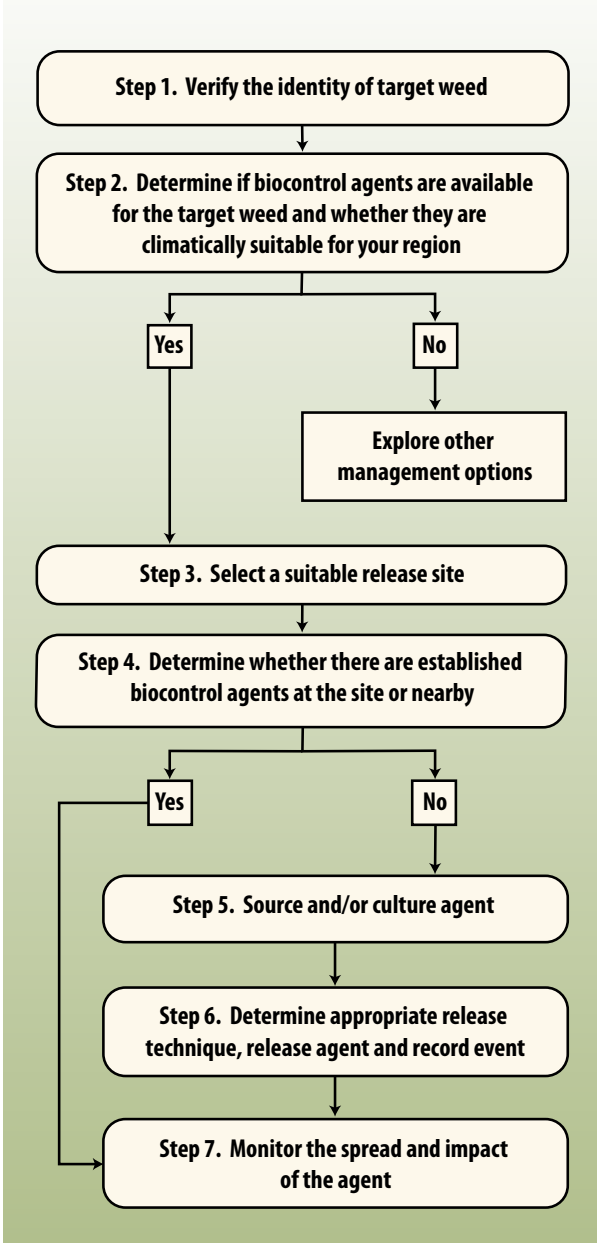
The processes and information requirements for importation, testing and approval of biocontrol agents of plants and invertebrates are set out in the Biosecurity Guidelines for the Introduction of Exotic Biological Control Agents for the Control of Weeds and Plant Pests and revised periodically on the Department of Agriculture website (DAWE 2020). Before approval is given, the risk analysis needs to show that the agent will not damage native flora or agricultural stock or crops.

Weeds and the law

The use of weed biocontrol agents on your property may not be sufficient for landholders/managers to meet their general biosecurity duty and/or legal obligations to prevent, eliminate, minimise or manage certain weeds. Refer to your relevant state and territory's biosecurity/weed legislation and talk to your local weed or biosecurity officer for information.

Undertaking a weed biocontrol program

Once a weed biocontrol agent is approved for release, your weed biocontrol program needs to be undertaken in a systematic sequence to ensure maximum probability of success. The key steps outlined below recommend an approach for best practice management.



Recommendation

Weed biocontrol programs often form part of an integrated best practice weed management plan. The details of this plan will depend on the target weed, location and timeframe for control. However, when integrating chemical use with biocontrol management, there are a couple of rules to follow:

- Do not use herbicides in close proximity to your release site, but outlying infestations and perimeters can be managed using chemical and/or mechanical means.
- Densely invaded areas can be left solely for control by agents. This allows a good opportunity for the agent to establish and spread beyond the release site.

P. Sullivan

A mechanical harvester is used to control an outlying infestation of water hyacinth away from the core biocontrol release site, allowing the agent to multiply at the release site.

Step 1

Verify the identity of target weed

It is important to correctly identify the target weed to ensure the correct agents are released against it. For example, common prickly pear (*Optuntia stricta*) is only controlled by the cochineal *Dactylopius opuntiae* while tiger pear (*Opuntia aurantiaca*) is controlled mainly by *Dactylopius austrinus*. These agents are not effective on the alternate species. Invasive *Cylindropuntia* species are controlled by different lineages[1] of the cochineal *Dactylopius tomentosus*. With this in mind, it is clear to see that effective control will not be achieved if the *Cylindropuntia* species are not correctly identified and matched to the appropriate lineage of agent[1].

Refer to online weed identification guides. See page 201 in further information for resources. If in doubt contact your local weed or biosecurity officer.

Step 2

Determine if biocontrol agents are available for the target weed and whether they are climatically suitable for your region

Refer to the summary table of target weeds and available biocontrol agents in Appendix 4 and, if necessary, use additional literature (see further information section, or refer to ibiocontrol (https://www.ibiocontrol.org/catalog/); Winston *et al.*, 2014). Select biocontrol agents that are climatically suited to your area and have been demonstrated to negatively impact the target weed. Your local weed or biosecurity officer should be able to supply you with this information.

[1] Lineages described here are populations of the same insect species (e.g. *Dactylopius tomentosus*) that can only be separated by their different abilities to feed, lay eggs and develop on a target species. Only molecular tools can distinguish between different lineages.

Step 3

Select a suitable release site

Selection of a good release site (often called a nursery site) is important because it gives the biocontrol agent the best chance to establish and control the weed. Good sites will have an abundance of a vigorously growing weed species, be located in a region where the weed is widespread, and generally be free from disturbance that may interfere with your biocontrol program. It is important to record release site details (see Appendix 2 for form and guidelines). Release sites should be revisited to monitor agent establishment and impacts. Release sites also act as nursery sites for agent collection and redistribution to other sites.

Recommendation

Minimal site disturbance is required over an extended post-release period to ensure that the agent establishes and disperses well. In farming situations, where stock or farming practices may impact the weed, consider fencing the area where the biocontrol agent will be released to minimise nursery site disturbance.

W. Mitchell

A fenced release site is used to exclude livestock for the biocontrol of stemless thistle using the rosette weevil, Burra, South Australia. Minimum disturbance will optimise agent establishment.

Ensure that:

- the land owner/manager is in agreement with the establishment of the release site and its conditions
- the land owner/manager has some understanding of biocontrol theory, including realistic expectations of how long it may take and what results may be expected
- the land owner/manager understands how to manage the release site
- no pesticides are utilised in or near the release site
- the release site is accessible for regular monitoring.

It is good practice to have the land owner/manager understand their responsibilities that include:

- continual management of the weed across their property (remind them of their biosecurity duty)
- creating a buffer zone around the release site, where no other management techniques are implemented
- communication with local weed/biosecurity officer for ongoing access to the site in case of property sale or transfer.

It is desirable not to release biocontrol agents at sites where:

- there is a low density of target weed
- the target weed is environmentally (usually water) stressed
- there is planned site disturbance in the near future.

Step 4

Determine whether there are established biocontrol agents at the site or nearby

It is important to establish whether any biocontrol agents have been released in the near vicinity of your site. Information about agent releases and establishment in the area can be sourced from local weed/biosecurity officers or weed biocontrol researchers. Also check the **Australian Biocontrol Hub** on the **Atlas of Living Australia** website (https://biocollect.ala.org.au/biocontrolhub). In addition, systematically monitor your release sites for any presence of other biocontrol agents on your target weed.

Step 5

Source and/or culture agent

Contact your local or state government weeds professional for biocontrol advice, including where the biocontrol agents may be sourced. Keep in mind your state or territory agency may be rearing agents suitable for your target weed. The Australian Biocontrol Hub is a useful source of information on where various biocontrol agents are established and can be collected.

Sometimes it is advantageous to collect, redistribute and monitor your own biocontrol agents. If your agent lends itself to being easily collected and redistributed by your local community, consider the guidelines specified for each agent and its target within this manual, and refer to the biocontrol agent collection and redistribution techniques in Appendix 1.

Step 6

Determine appropriate release technique, release agent and record the event

Each biocontrol agent has an optimum release technique, designed to provide optimal control of the weed in the shortest possible time by enhancing biocontrol establishment, population growth and dispersal. All techniques focus on the responses and needs of the agent to different climatic conditions, habitat, its mating requirements and dispersal ability. While this manual tailors optimal techniques for each agent and its target, there are a few universally helpful tips.

If possible, releases should be made on **quality (good condition) host plants** as this is likely to improve the establishment and impact of the agents. Drought stressed or plants sprayed with herbicide do not make good host plants.

Don't be tempted to break up the population intended for one release into several smaller batches

G. Lindschau

Releasing Paterson's curse pollen beetles on good quality host plants.

of biocontrol agents that are widely scattered. Doing this increases the risk of the agent not establishing. Releasing one large group of a particular agent enhances their ability to find mates, increases their population size, and improves dispersal.

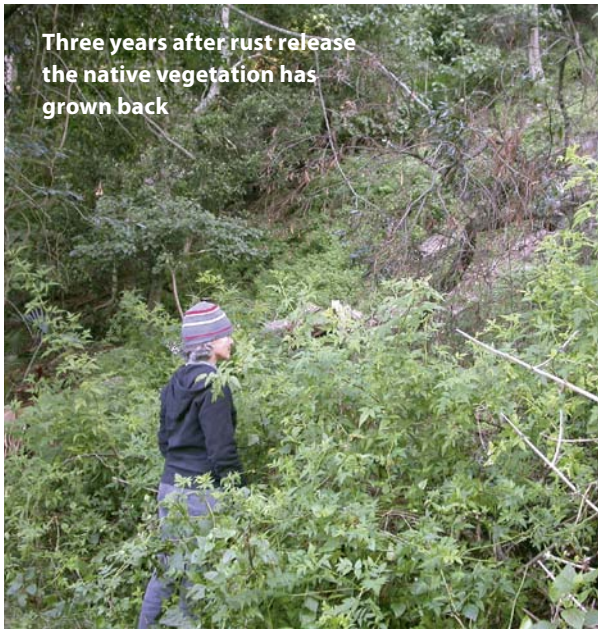
Make a record of your releases using the form in Appendix 2. Also upload your release data and field release observations to the Australian Biocontrol Hub. Recording release data is important for assisting others who are also releasing agents at other sites. It can provide others with useful information such as determining environmental factors outlining successful or unsuccessful establishment.

Step 7

Monitor the spread and impact of the agent

Monitoring and evaluation is an essential component of any weed management program. It is very important to regularly and systematically monitor and analyse a release site for agent establishment, population increase, dispersal and impact of the biocontrol agent on the weed. Take a look at the monitoring form in Appendix 3 to assist you with correctly capturing your data.

Fixed-point photography is a great tool for assessing before and after impacts of biocontrol agents. Consider using a marker such as a star picket to mark the spot. Always take photographs from the same point and preferably at the same time of day.



L. Morin

L. Morin

Before and three years after pictures of Crofton weed suppressed by the Crofton weed rust and resulting native vegetation recovery.

Introduction

To monitor the response of invasive and native plants before, during and after weed control programs, refer to the Monitoring Manual for Invasive and Native Flora (Watson *et al*., 2021). The manual should be used to design your own monitoring plan, relative to your expertise, resources and a timeframe over which you will be able to see the results of your control program. The manual outlines a two-tiered approach to monitoring, with techniques ranging from simple qualitative assessments, to more rigorous advanced methods, allowing landholders/managers to adopt the most suitable options for their objective.

Watson, G., French, K., Burley, A. and Hamilton, M. (2021). Monitoring Manual for Invasive and Native Flora. Department of Planning, Industry and Environment.

Hygiene

It is important to remain vigilant when undertaking weed management activities. Seed and plant fragments are easily moved on clothing, backpacks, machinery and shoes and often without detection.

When choosing any control method, and prior to starting any program, it is important to plan how to prevent the target weed (and other weeds) being spread further. Refer to any relevant biosecurity or weed management plans that relate to your management site.

In biocontrol programs, agents by themselves, or when transported with other plant material, may contain contaminants such as weed seeds, plant fragments, soil pathogens and arthropod pests, and predators and parasitoids that can weaken or kill biocontrol agents.

A. McConnachie

Thoroughly inspect your vehicle before leaving a contaminated site.

Tips for reducing spread of weeds and pests

- Collect agents from 'clean' release/nursery sites, i.e. sites that are free from other known problematic weeds and pests.
- Scrutinise all collected material for unwanted contaminants before transport.
- Minimise the transport of plant material with biocontrol agents to help reduce the risk of spreading the weed and or pests associated with the weed. Where possible, separate agents from plant material using a sieve or aspirator before transport (see Appendix 1 for collection methods).
- Dispose of plant material by on-site or off-site deep burial (>1 m depth) or by solarisation. For solarisation, material is transferred into black plastic bags and left in the sun to cook for a period of two to three months to destroy reproductive material, after which the bags can be disposed of off-site or composted on-site. Do not include reproductive material in green waste.
- When leaving a site, thoroughly inspect and clean vehicles, collecting equipment and machinery suspected of carrying soil or weed material.
- Ensure clothing and footwear is free from soil and weed material before leaving a site.

Weeds and an overview of their biocontrol options

Biocontrol research is ongoing and continues on a variety of other weed targets. At the time of writing new agents have been approved for release and are not covered within this manual. Please contact an appropriate officer in your state or territory for up to date information on target weeds and agent availability.

Alligator weed
Alternanthera philoxeroides

Alligator weed is a perennial herb native to South America where its range extends from Argentina to Brazil (Sosa *et al.*, 2008). It grows in aquatic, semi-aquatic and terrestrial habitats in tropical, subtropical and temperate regions (Julien *et al.*, 2012b). Plants have dark green, opposite leaves (5 to 40 mm wide), hollow stems (aquatic form) and papery white flowers (8 to 10 mm in diameter). The roots are short and filamentous in water but are larger and more extensive in soil. Thought to be a sterile hybrid, alligator weed in Australia does not produce viable seed (Sosa *et al.*, 2008).

First reported in Newcastle Harbour and Botany Bay, alligator weed most likely came from shipping ballast and or cargo during the Second World War (Julien *et al.*, 2012b; Parsons and Cuthbertson, 2001). Recognised in the 1970s as a serious aquatic weed, alligator weed was listed as a Weed of National Significance in 1999 due to its environmental and

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Alligator weed foliage and flowers.

NSW DPI

P. Sullivan

NSW DPI

Invasions of alligator weed (above and right).

economic impacts, invasiveness and potential to spread extensively by vegetative fragments in both aquatic and terrestrial systems.

Australia introduced three species of insects from South America to test their potential as biocontrol agents for alligator weed. Two of the three species released, a flea beetle and a moth, established in the field.

Recommendation

The alligator weed flea beetle and moth can control alligator weed in aquatic environments only. Biocontrol efforts should focus primarily on the flea beetle, as it is more damaging and more abundant than the moth.

Be careful when working with alligator weed. New infestations easily occur from transported broken off plant fragments.

Alligator weed flea beetle
Agasicles hygrophila

First released in New South Wales in 1977, the alligator weed flea beetle from Argentina provides excellent control of the aquatic form of alligator weed but not the terrestrial form (Winston *et al.*, 2014). Feeding damage and emergence holes created by adult flea beetles, allow the entry of water and organisms that assist with the desiccation of floating mats causing them to break up and sink. With faster insect development in warmer climates, control of aquatic forms of alligator weed occurs in less than nine months.

Identification

The adult flea beetle is approximately 5 mm long and readily identified by black and yellow stripes on its back. It has enlarged hind legs, which enable it to jump between plants. Small shot holes in leaves are damage typically inflicted by adult flea beetles.

Larvae are small (around 5 mm long) and grey. Clear windowpanes on top of leaves occur from larvae feeding on their underside. Larvae also feed externally on stems (Center *et al.*, 2018).

NSW DPI

Alligator weed flea beetles.

P. Sullivan

Feeding damage by alligator weed flea beetle.

G. Buckingham, USDA, Bugwood.org

Alligator weed flea beetle adult and larvae.

Life cycle

In Argentina, the flea beetle has five generations per year. In Australia, the generation time in summer can be as short as one month. The adult female lays around 1000 eggs on the underside of leaves during her six-week lifespan. After hatching, larvae feed on the underside of leaves and externally on stems. They pupate within the hollow stems above the waterline prior to emerging as adults. Adults and larvae do not feed on the roots of alligator weed (Center *et al.*, 2018).

Note: Flea beetles are unable to complete their life cycle on the terrestrial form of alligator weed as the stems are solid which prevents pupation. Adults and larvae can still damage the terrestrial form of alligator weed where it grows adjacent to aquatic plants.

Field collecting and rearing

Rearing is unnecessary. Flea beetles are widely distributed across the east coast of Australia. Adults can be collected from early summer to early autumn. Although adults and larvae can overwinter, releases to new areas are best in summer when populations are at their peak.

Using a sweep net collect at least 100 adults for each release site (see Appendix 1 for technique). Collect on warm sunny days when insects are most active. Prior to redistribution, flea beetles can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

Recommendation

Practise hygiene

To prevent the spread of plant material and contaminants, it is preferable to collect and release adult flea beetles rather than larvae attached to plant material (see page 10 on hygiene).

How and when to release

Release collected flea beetles directly onto healthy plants as soon as possible. To assist with nursery site establishment, release beetles in small bays away from the main waterway channel. This gives the flea beetles the best opportunity to establish decent-sized populations with a reduced chance of washing downstream. Record release information on your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for flea beetle presence (adults and larvae) and feeding damage (holes in leaves and defoliated plants) at the nursery site one year post release and record its presence or absence as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Alligator weed moth
Arcola malloi

First released in New South Wales in 1977, the alligator weed moth from Argentina provides limited control of the aquatic form of alligator weed (Winston *et al.*, 2014). Although impact is occasionally good in localised infestations of alligator weed, overall its impact is minimal in comparison to the flea beetle and does not contribute to the significant control of alligator weed.

The adult moth is brown and 13 mm long. Females lay between 200 and 300 eggs during their week-long adult life. Larvae feed inside the stem and consume up to eight stems before pupating within.

Due to its limited impact and dispersal, redistribution is unnecessary.

Monitoring establishment and dispersal

Record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. If seen, monitor for its presence annually as per your guidelines (Appendix 3).

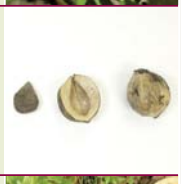
NSW DPI

Alligator weed moth.

Bitou bush and Boneseed

Bitou bush and boneseed are two closely related perennial shrubs introduced to Australia from South Africa. They look similar and are subspecies of *Chrysanthemoides monilifera* that have invaded a wide range of habitats in southern Australia. Bitou bush (subsp. *rotundata*) is predominately restricted to coastal areas on sandy or medium-textured soils of south-eastern Australia whereas boneseed (subsp. *monilifera*) can invade a range of soil types and habitats across southern Australia. In 2000, they were collectively listed as Weeds of National Significance due to their combined impact as widely established, serious environmental weeds of sand dunes, grasslands, heathlands, woodlands and forests.

See the Bitou bush Management Manual: current management and control options for bitou bush (*Chrysanthemoides monilifera* ssp. *rotundata*) in Australia (Winkler *et al.*, 2008 – see full citation below[1]). Available via https://profiles.ala.org.au/opus/weeds-australia.

bitou bush (ssp. *rotundata*)			boneseed (ssp. *monilifera*)	
	sprawling shrub, 1–2 m high, sometimes erect	**habit**	erect shrub, up to 3 m high	
	3–7 cm long, broader oval shape, smooth or only slightly toothed edges	**leaves**	3–9 cm long, elongated oval shape, irregularly toothed edges	
	11–13 'petals' flowers year round with a peak from April to July	**flowers**	4–8 'petals' flowers from late winter to spring (mainland), to early summer (Tas.)	
	egg-shaped fruit, black when ripe	**fruit**	round fruit, black when ripe	
	seed coat is egg-shaped, rough, dark brown to black	**seeds**	seed coat is round, smooth, bone-coloured (seed also shown)	
	leaves with smooth edges	**seedlings**	leaves with toothed edges	

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[1] Winkler, M.A., Cherry, H. and Downey, P.O. (eds) (2008). Bitou bush Management Manual: current management and control options for bitou bush (*Chrysanthemoides monilifera* ssp. *rotundata*) in Australia. Department of Environment and Climate Change (NSW), Sydney.

Bitou bush
Chrysanthemoides monilifera subsp. *rotundata*

Bitou bush is a thicket-forming, semi-succulent, woody shrub (1 to 2 m) with bright yellow flowers (2 to 3 cm in diameter with 11 to 13 petals). Its shallow and extensive root system can give it the appearance of a creeper rather than a shrub. Plants have bright green, glossy, broadly oval-shaped leaves (3 to 8 cm long) with smooth edges (though sometimes slightly toothed). Flowering occurs year-round with a peak from April to July. The egg-shaped fruits when mature are black and contain a single, hard coated seed. Individual mature plants can produce up to 48,000 seeds annually (Weiss *et al.*,1998).

I. Burnett

Bitou bush tolerates saline conditions well and readily invades coastal sand dunes.

Bitou bush was introduced to Australia, most likely in the ballast of ships, and from 1946 to 1968 was deliberately planted for dune erosion control along the east coast of Australia. Its vigorous growth, prolific seed production and ability to tolerate saline conditions enhance its ability to readily out-compete and displace native flora. Its invasion is further facilitated by chemicals that are exuded from the roots which build up in the soil and prevent native seedling germination (French *et al.*, 2008).

Twelve insect species were introduced to Australia to test their potential as biocontrol agents for bitou bush. From 1989 to 2018 seven of these were released and four established, including bitou tip moth (*Comostolopsis germana*), bitou leaf-roller moth (*Tortrix* sp.), bitou seed fly (*Mesoclanis polana*), and bitou tortoise beetle (*Cassida* sp. 3).

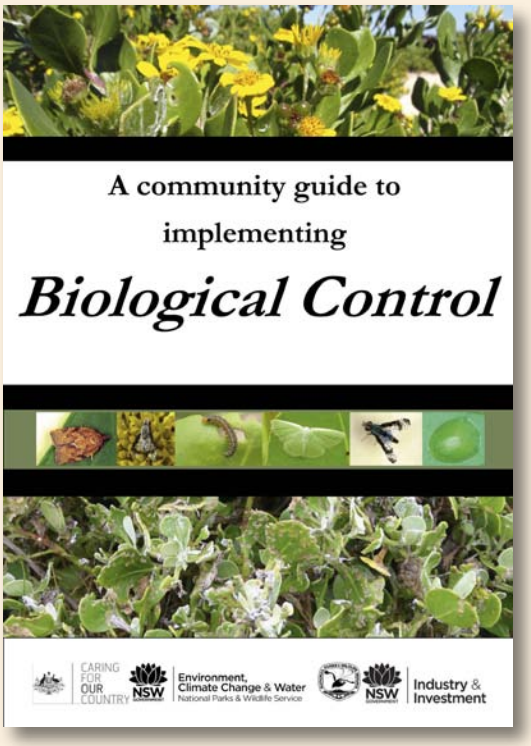
Recommendation

Effective control of bitou bush is best achieved using conventional control methods in combination with biocontrol, particularly with the presence of several complementary agents in the field. For example, the bitou leaf-roller moth, bitou tip moth and bitou seed fly coexist well in the field and their damage is complementary. In combination their impact minimises seed production in bitou bush.

R. Holtkamp

Bitou bush showing signs of heavy attack from the combined impact of biocontrol agents.

A useful community guide for redistributing bitou bush biocontrol agents is available via the Literature & Links tab at https://profiles.ala.org.au/opus/weeds-australia/profile/Chrysanthemoides monilifera subsp. rotundata.



Bitou tip moth
Comostolopsis germana

First released in New South Wales in 1989, bitou tip moth from South Africa is now widespread throughout the invaded range of bitou bush (Adair *et al.*, 2012; French *et al.*, 2019). As its name suggests, bitou tip moth larvae feed on the soft apical ends of leaves, stems and buds. Damage by the larvae is highly variable but has been found to reduce flowering by 30% (French *et al.*, 2019) and seed production by over 50% (Holtkamp, 2002).

Identification

Adult moths are pale green with narrow, white-wavy-lines across the fore and hind wings. Adults rest with their wings flat to the surface and their wingspan is approximately 15 mm in this position. Larvae are pale green to white (10 mm long) with a dark brown head capsule (Adair *et al.*, 2012).

T. Morley

Bitou tip moth.

R. Holtkamp

Larva of bitou tip moth showing pale green to white body within its silk canopy.

Life cycle

The bitou tip moth has multiple generations per year with an egg to adult generation time of six to nine weeks. Adult moths lay an average of 110 eggs on stems and foliage. Within a few days of hatching, larvae construct protective canopies using leaves and white silk where they feed on young foliage, soft stems and developing buds before pupating between folded leaves. Adults are non-feeding.

Field collecting and rearing

Rearing bitou tip moth is time consuming and not recommended as larvae are widely distributed and may be easily collected in the field.

The bitou tip moth is widely distributed across the range of bitou bush and generally does not require redistribution. However, should bitou bush populations be located where no signs of bitou tip moth damage are evident, then larvae and pupae (not adult moths as they are nocturnal and unlikely to be seen) can be collected from established sites. Look for the presence of silk canopies (which have a white webbing appearance on the tips of young foliage and developing buds) to find larvae and pupae in the warmer months (September to February). Larvae of the bitou tip moth can be easily confused with the bitou leaf-roller moth. To differentiate, open leaves and look for pale green to white coloured larvae with a brown head as opposed to the dark green larvae and orange head of the bitou leaf-roller moth. Select small cuttings (10 to 20 cm long) with multiple larval canopies that are prevalent during spring and summer. Prior to redistribution, cuttings containing larvae and pupae can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

P. Sullivan

White webbing and plant tip damage by bitou tip moth.

How and when to release

Secure each larva/pupa infested cutting near the growing tips of at least 20 healthy bitou bush plants. You will need around 10 cuttings per large bitou bush plant to ensure establishment. Larvae will move onto healthy plants to feed as cuttings dry out and die. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Monitor the establishment and combined impact of both the bitou tip moth and bitou leaf-roller moth together as they have similar impact, their identities are easily confused, and they live within similar structures.

Look for the white webbing created by the bitou tip moth and bitou leaf-roller moth on the tips of plants one year post release. Separate by identifying the pale green larvae of bitou tip moth and dark green larvae with orange heads of bitou leaf-roller moth within the silk canopies and pupae between folded leaves. Pupae of these two species are virtually indistinguishable, other than size, with bitou leaf-roller moth being much larger. Record the presence or absence of larvae and pupae as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Bitou leaf-roller moth
Tortrix sp.

The bitou leaf-roller moth from southern Africa was released across south-eastern Australia between 2000 and 2004 (Adair *et al.*, 2012). The long-term impacts of the bitou leaf-roller moth have not been determined. In its native range it can reach high densities, significantly reduce seed production and kill entire bushes. Since its introduction, minimal establishment has occurred in south-eastern Australia and it is still the subject of ongoing research. Redistribution programs and follow up observations will assist in enhancing our understanding of this agents' impact.

Identification

You are unlikely to see adult bitou leaf-roller moths as they are nocturnal. However, they are beige-coloured (up to 15 mm long) with a light brown mottled zig-zag band across their wing. When at rest their wings form a triangular shape. The larvae have an orange head and darken with age from dark green to black (up to 20 mm long). To protect themselves against predators, larvae construct white silk-like canopies by webbing together leaves at the shoot tips of plants. Here they consume the leaves and stems from within, which results in plant tip death.

Life cycle

Bitou leaf-roller moths have up to three generations per year. Adults lay eggs in clusters on the upper and lower surface of leaves. After around 8 days, eggs hatch and develop into dark green caterpillars. Like the bitou tip moth, newly hatched larvae move to the shoot tips to construct protective feeding shelters by joining two or more leaves together with silk. Older larvae can feed lower down on stems. Pupation occurs within the feeding shelters after which adults emerge approximately 10 days later. Adult moths live for about 14 days.

A. Swirepik

Bitou leaf-roller larva within its silk canopy shelter.

P. Sullivan

Adult bitou leaf-roller moth in its resting position.

P. Sullivan

Egg cluster of bitou leaf-roller moth.

Field collecting and rearing

For rearing and redistributing the bitou leaf-roller moth see A Community Guide to Implementing Biological Control (Jenner *et al.*, 2010 – currently available via the Literature & Links tab at https://profiles.ala.org.au/opus/weeds-australia/profile/Chrysanthemoides monilifera subsp. rotundata).

Rearing the bitou leaf-roller moth is time consuming and some groups or organisations may find it easier to collect larvae from the field for redistribution.

Similar to the methods described for redistributing the bitou tip moth, collect bitou leaf-roller moth larvae and pupae from November through to late January. Look for the presence of silk canopies that have a white webbing appearance on the tips of young foliage and on developing buds. As canopies of bitou tip moth are similar to those of the bitou leaf-roller moth, open the canopies and look for the dark green larvae with orange heads of the bitou leaf-roller moth and not the pale green larvae of the bitou tip moth. Bitou leaf-roller moth larvae are very active when exposed and will vigorously wriggle backwards and fall to the ground, so place a container beneath the leaves for collection. You can also select small cuttings (10 to 20 cm long) with larvae that have massed together. This will potentially lead to the collection of both agents, so check samples prior to releasing. Prior to redistribution, cuttings containing larvae and pupae can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

How and when to release

Secure each larva- and pupa-infested cutting near the growing tips of at least 20 healthy bitou bush plants. You will need around 10 cuttings per large bitou bush plant to ensure establishment. Larvae will move onto healthy plants to feed as cuttings dry out and die. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for the white webbing canopies created by the bitou tip moth and bitou leaf-roller moth on the tips of plants within one year of release. Differentiate between the moth species within the silk canopies by identifying the pale green larvae of the bitou tip moth to the dark green larvae with orange heads of the bitou leaf-roller moth. The pupae within folded leaves of these two species are virtually indistinguishable other than size with the bitou leaf-roller moth being much larger. Record the presence or absence of larvae and pupae as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Bitou seed fly
Mesoclanis polana

First released in New South Wales in 1996 the bitou seed fly from southern Africa is now widespread throughout the invaded range of bitou bush in Australia (Adair *et al.*, 2012). Larvae of the seed fly play a vital role in reducing the overall seed production of bitou bush by feeding on and destroying developing seeds. The combined activity of bitou seed fly with bitou tip moth and bitou leaf-roller moth, while complementary, may cause a decline in the seed fly's activity due to the reduction in available flowers and seed heads caused by the damage from the two moth species.

CSIRO

Bitou seed fly.

Identification

The bitou seed fly is easy to identify by its small, black body (up to 5 mm long) with two white stripes down its length and distinctive black pigmented wing pattern. Larvae are barrel-shaped (up to 5 mm long) and white with a dark head capsule. Damage caused by the bitou seed fly is easily identified by shrivelled ray floret seeds and small adult emergence holes beneath the flower head.

Life cycle

Bitou seed flies have multiple generations each year with a very short lifespan of up to one week. Adult flies lay white cigar-shaped eggs between flower buds. Larvae tunnel through flowers, and feed on flower stalks and developing seeds destroying their viability before pupating within the seed heads.

P. Sullivan

Seed of bitou bush destroyed by a bitou seed fly larva.

T. Morley

Flower heads of bitou bush demonstrating typical shrivelled seed damage caused by bitou seed fly.

Redistribution

Redistribution of bitou seed fly is unnecessary as they are widely established throughout the invaded range of bitou bush.

Monitoring establishment and dispersal

Look for bitou seed fly presence (adults and larvae) on and within flower heads within one year of release. Damage caused by the bitou seed fly is easily identified by shrivelled ray floret seeds. If present, record your sighting on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Bitou tortoise beetle
Cassida sp. 3

The bitou tortoise beetle has established and is present at most of the initial release sites after its release in 1995. However, its numbers remain low, dispersal rate is extremely slow and its impact on bitou bush appears to be negligible (Adair *et al.*, 2012). It is thus not recommended for redistribution.

Boneseed
Chrysanthemoides monilifera subsp. *monilifera*

Boneseed is a thicket-forming, woody shrub (up to 3 m tall) with bright yellow flowers (2 to 3 cm in diameter with four to eight petals). In contrast to the sprawling habit of bitou bush, boneseed is an erect shrub with a shallow root system. Leaves are oval- to spoon-shaped (3 to 9 cm long) with irregular serrations along margins. Flowering occurs from late winter to spring and to early summer in colder regions. The round fruits turn black when mature and contain a single, hard bone-coloured seed. Individual mature plants can produce up to 50,000 seeds annually (Weiss, 1984).

Boneseed was introduced into Australia in the 1850s as an ornamental plant (Brougham *et al.*, 2006) and as a form of erosion control in parts of coastal New South Wales and Victoria. Its prolific seed production, dispersal over long distances aided by a range of animals, and ability to establish and grow rapidly in undisturbed and disturbed vegetation in a range of habitats (from dunes, mallee, open woodlands and sclerophyll forests) and varying soil conditions enhances its ability to outcompete and displace native species. Unlike its impact on native bushland, it does not impact agricultural land as it will not persist when regularly grazed or cultivated.

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Boneseed flowers and foliage.

NSW DPI

Boneseed flowers and foliage.

Of the eight agents released since 1989 for biocontrol of boneseed, only the boneseed leaf-buckle mite

Recommendation

Due to uncertainty of biocontrol impact for boneseed, conventional control methods (including herbicide treatment, hand-pulling and fire) should first be prioritised. Refer to the Boneseed management manual: current management and control options for boneseed (*Chrysanthemoides monilifera* ssp. *monilifera*) in Australia available via the Literature & Links tab at https://profiles.ala.org.au/opus/weeds-australia/profile/Chrysanthemoides monilifera subsp. monilifera.

(*Aceria* sp.) has established. The reasons for the failure of other agents are unknown, although they may be related to predation and parasitoids.

Boneseed leaf-buckle mite
Aceria sp.

First released in Tasmania in 2008, boneseed leaf-buckle mite, from South Africa, has since been released at more than 90 sites in Victoria, Tasmania and South Australia but has only established at a handful of sites in Victoria and Tasmania (Adair *et al.*, 2012). The long-term impact of the mite has not been determined but redistribution programs are encouraged. In its native range, boneseed leaf-buckle mite is known to heavily infest boneseed plants with a resultant lower growth rate and reproductive output being commonplace.

Identification

Adult boneseed leaf-buckle mites are invisible to the naked eye (approximately 0.15 mm long) with a worm-like body and four legs. Boneseed leaf-buckle mites feed by using piercing and sucking mouthparts to extract plant cell contents. This feeding induces the formation of erinea (a type of gall). Erinea are dense patches of white/brown coloured hair-like structures which lead to the disruption of normal leaf development causing leaf twisting and buckling.

Life cycle

Generation times of boneseed leaf-buckle mite are unknown but like other closely related eriophyid mites, boneseed leaf-buckle mites are assumed to have multiple generations per year (temperature dependent) with a short adult lifespan of a few weeks. Females can lay one or more eggs daily and those that are unfertilised give rise to male offspring. Adult boneseed leaf-buckle mites lay eggs within shoot tips or inside erinea, both of which provide feeding sites and shelter for juvenile mite populations before they emerge as larvae to continue their development through to nymphs and adulthood.

S. Ivory, SARDI

A colony of boneseed leaf-buckle mites on boneseed causing the formation of specialised galls (abnormal outgrowths) called erinea.

Field collecting and rearing

The boneseed leaf-buckle mite is difficult to rear, but reasonably easy to transfer via infected leaves. To do this look for signs of erinea on leaves as this indicates mite activity. Collect at least 15 (but more is better) cuttings (around 30 cm long) with leaves containing erinea in spring or autumn when conditions are mild. Prior to redistribution, cuttings containing mites can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

How and when to release

Secure infested cuttings onto at least ten healthy boneseed plants with tie wire. Mites will move across to the new host plant as infected leaves dry out. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for the formation of erinea the following spring and autumn. Record the presence or absence of larvae and pupae as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Blackberry

Rubus fruticosus aggregate

Blackberry represents a collective of at least 16 closely related species, subspecies, varieties and hybrids belonging to the *Rubus fruticosus* aggregate (Morin and Evans, 2012). Due to difficulties in distinguishing between species, this aggregate is grouped together for convenience.

Native to Europe, blackberry is a semi-deciduous, woody shrub with scrambling and arching prickly biennial stems (canes) (up to 7 m long) that form from a perennial woody crown (up to 20 cm in diameter). These stems can grow vegetatively by taking root to form dense impenetrable thickets (up to 4 m high). Plants have compound leaves, with clusters of three to five leaflets that are usually dark green on the upper surface and lighter on their underside. The leaflet veins and stalks are covered with short, curved prickles and leaves arise alternately along the canes. Flowers are white or pink (2 to 3 cm in diameter) and form clusters at the end of branches. The edible 'berries' transition from green, through red to black as the fruit ripens. A single plant can produce up to 400,000 seeds per year with thickets producing up to 13,000 seeds per m^2 (Amor *et al.*, 1998).

NSW DPI

Blackberry plants produce clusters of white or pink flowers.

NSW DPI

Fruit colours change from green to red to black as blackberry ripens.

NSW DPI

Blackberry invasion.

Blackberry was introduced as a garden ornamental plant in the 1830s and later was promoted as a hedge plant, source of edible fruit, and as way of controlling erosion. From the late 1800s blackberry was declared a noxious species in parts of Victoria, and by the early 1900s was declared a state-wide noxious species due to its negative impacts on agriculture, forestry and within natural systems (Parsons and Cuthbertson, 2001). Recognised as one of Australia's worst weeds, blackberry at one time was estimated to occupy close to 9 million ha of temperate Australia; an area greater than the size of Tasmania. In 1999, blackberry was listed as a Weed of National Significance due to its invasiveness, environmental and economic impacts and potential to spread.

Currently the only biocontrol agent approved and released on blackberry in Australia is the leaf rust fungus (*Phragmidium violaceum*) (Morin and Evans, 2012). This rust is highly efficient at spreading by natural means and will colonise blackberry when environmental conditions are suitable. Land managers do not need to redistribute the rust as it is widely established.

Different species, subspecies, varieties and hybrids belonging to the *Rubus fruticosus* aggregate react differently to the biocontrol agent. For example, species originating from North America or Asia are not susceptible to the rust in Australia. Consequently, where infestations are made up of mixed species, a species that has been biologically controlled can be replaced by a species with a higher tolerance. If the rust is not present then it is highly likely the conditions are not suitable, so it will be necessary to use another control method such as herbicides or mechanical control.

Blackberry leaf-rust fungus
Phragmidium violaceum

The blackberry leaf-rust fungus was first recorded in Victoria in 1984 and has since spread naturally throughout southern Australia (Morin and Evans, 2012). Laboratory testing confirmed the pathogenicity and host specificity of this strain, and since 1991, nine additional strains of the rust were sourced, tested and released against blackberry with variable impact (Morin *et al.*, 2006). As its name indicates, the fungus primarily attacks the leaves causing defoliation. The rust can also be found on flower buds and unripe fruit. Tips of the heavily rust-infested stems can die back and prevent the production of new daughter plants that develop after taking root in the ground. Successful biocontrol of blackberry is dependent on matching virulent

NSW DPI

Blackberry leaves with rust.

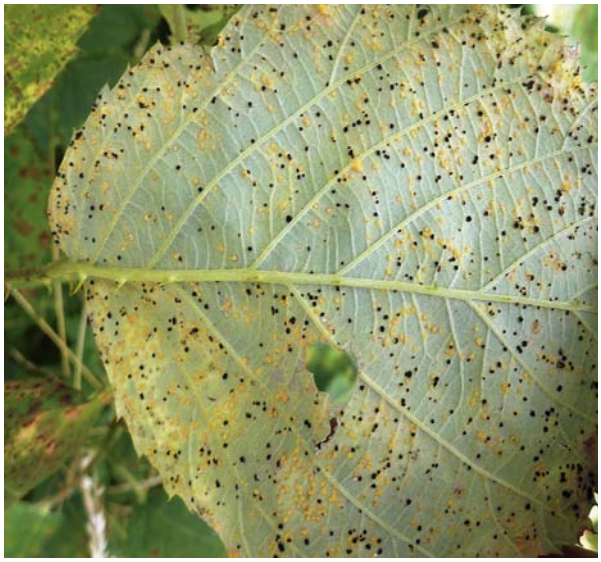
rust strains to susceptible blackberry entities. Just as important are suitable weather and blackberry growing conditions for development of the rust.

More generally, the rust has had the greatest impact in areas of moderate temperatures (>20°C) with high rainfall (>750 mm) and or high humidity. Blackberry is generally unaffected by rust in low summer rainfall regions, shaded habitats and high-altitude areas (Adair and Bruzzese, 2006).

Identification

Symptoms of the blackberry leaf-rust fungus can be seen at any time of the year on flower buds, unripe fruit, and green parts of the growing canes, but is most obvious on leaves. The rust fruiting bodies (spores) damage the leaves. On the leaf upper surface, the rust appears as characteristic purple-brown blotches (2 to 3 cm in diameter). On the leaf lower surface, however, it appears as corresponding yellow or black powdery pustules. Bright yellow, wind-dispersed spores are responsible for epidemics during the growing season, while black spores, produced at the end of the growing season, allow the rust to overwinter on infected plants. Severely infected leaves begin to dehydrate, turn brown, shrivel and will fall from the canes. Flowers and infected fruit can fail to ripen and similarly stems will die back.

Note: Heavily infected plants may sometimes look as though they have been sprayed with herbicide, so check the leaves for signs of infection.



S. Ivory, SARDI

Blackberry rust appearing as yellow summer spores and black overwintering spores.

Life cycle

The blackberry leaf-rust fungus has several different spore stages representing the asexual and sexual components of its life cycle. The two most commonly seen stages include the yellow 'summer' spores (urediniospores) and the overwintering, sticky black spores (teliospores). Around spring, which corresponds with the emergence of new canes, the yellow summer spores will germinate in the presence of moisture. During their generation time of around 8 to 10 days, these microscopic spores can be easily spread by wind to infect surrounding blackberry plants. To infect leaves, spores enter through stomata (breathing pores) on the lower surface of leaves. In late summer and autumn, they develop pustules and produce new black sticky spores that remain dormant and attached to the leaf throughout the winter, after which they begin the infection cycle again in the following spring.

S. Ivory, SARDI

S. Ivory, SARDI

Blackberry leaf-rust showing purple to brown blemishes on the upper surface (top) and orange pustules on the leaf underside (bottom).

Three other fungal diseases are found on blackberry and may be confused with the blackberry leaf-rust fungus. These include *Kuehneola uredines*, *Sphaerulina westendorpii* (formerly *Septoria rubi*) and *Cercospora rubi*. Keep in mind that the spores of blackberry leaf-rust fungus appear as characteristic purple-brown blotches (2 to 3 cm in diameter) on the leaf upper surface, with corresponding yellow or black powdery pustules on the under surface. To differentiate, check both sides of leaf surface as none of the other fungal diseases have corresponding upper and lower leaf surface pustules.

Field collecting and redistribution

This rust is widespread, and little is to be gained by redistributing it. Where plants are not infected, conditions are likely to be suboptimal and too dry for the rust to thrive.

Monitoring establishment and dispersal

Look for the blackberry leaf-rust fungus at any time of the year on any part of the plant, but particularly the leaves. If present, report your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Blue heliotrope

Heliotropium amplexicaule

Blue heliotrope is a hairy, drought hardy, perennial herb native to South America (Parsons and Cuthbertson, 2001). It is a low growing creeping (prostrate) plant (up to 30 cm in height) with numerous branched stems (up to 1 m long) that radiate from a central taproot. It grows along roadsides, old cultivation areas and pastures in warm temperate, subtropical and semi-arid regions of eastern Australia.

Blue heliotrope contains alkaloids that are toxic and largely unpalatable to stock. Continued ingestion causes chronic liver damage that can result in reduced productivity and even death.

Plants have dull green, long and tapered alternate hairy leaves (up to 80 mm long) that are soft to touch. The highly aromatic blue or purplish tubular flowers with yellow centres are distinctly arranged in two rows along the upper side of the narrow-coiled stems which straighten as the small, dark brown, warty seeds begin to mature.

P. Sullivan

Blue heliotrope in flower.

Largely dependent upon rainfall, abundant flowering can occur year-round in frost free areas but predominately coincides with several flushes of new growth between spring and autumn. While the seeds are dispersed by animals, water and contaminated soil and agricultural produce, the plant can also spread by root fragments.

P. Sullivan

Blue heliotrope invasion.

blue heliotrope

Blue heliotrope was likely introduced into Australia as an ornamental plant in the latter part of the 19th century (Parsons and Cuthbertson, 2001). First reported in New South Wales in the Hunter Valley, it soon spread throughout eastern New South Wales to occupy more than 110,000 ha. It has since rapidly spread into south-eastern Queensland with scattered infestations also found in Victoria and South Australia. Its adaptability to a wide range of soil and climate types indicates that its potential distribution has not been reached.

Australia introduced two species of insects from South America to test their potential as biocontrol agents for blue heliotrope. One of these, the blue heliotrope leaf-beetle (*Deuterocampta quadrijuga*) was approved for release and has subsequently established in the field (Briese, 2012a).

NSW DPI

Blue heliotrope leaf-beetle.

P. Sullivan

Blue heliotrope leaf-beetle larva.

P. Sullivan

Blue heliotrope leaf-beetle egg cluster on underside of leaf.

Blue heliotrope leaf-beetle
Deuterocampta quadrijuga

First released in 2001 and later redistributed from 2003 to 2010, the blue heliotrope leaf-beetle is now widespread throughout the cooler regions of New South Wales (e.g. tablelands and North West Slopes) where rainfall is high. Establishment remains poor in arid areas and in places with high summer temperatures, as beetles do not breed well on water-stressed plants. Where high densities of the beetle can establish, complete defoliation of above-ground plant material by adults and larvae occurs and localised control is achieved. Repeated defoliation assists to deplete the plant's underground reserves leading to plant death.

Identification

This large shiny black beetle (up to 10 mm long) is readily identified by its orange to red stripes along the length of elytra (hardened forewing). Larvae are cream to light pink with seven orange bands across their back. Depending on stage of development, larvae can range from approximately 2 to 15 mm in length. Both larvae and adults feed on the leaves and stems and can completely defoliate the above-ground biomass, particularly early in the growing season. Egg clusters can be found under the leaves and transition from orange to brown as they develop.

Recommendation

Blue heliotrope leaf-beetle has been released at a variety of sites in New South Wales, covering a range of climatic and ecological situations, but with variable results. Beetles prefer healthy, actively growing plants but this has still not been a guarantee of establishment. When rearing and releasing blue heliotrope leaf-beetle, it is important that you release the beetle in spring and on healthy plants where your site has sustained soil moisture (e.g. riparian or higher rainfall regions). It is also important to recognise that blue heliotrope is best managed when utilising biocontrol as one option of a wider integrated management strategy.

P. Sullivan

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Impact of the blue heliotrope leaf-beetle.

Life cycle

Blue heliotrope leaf-beetle can undergo several overlapping generations per year. Females are extremely fecund and are able to lay up to 1400 eggs over a four-month period. Eggs hatch in a little over a week and the emerged larvae feed on leaves before leaving the plant and pupating in the adjacent soil. Pupation takes place over the winter period from late May to September, before adults emerge to feed and reproduce over the warmer months (Briese, 2012a).

Field collecting and rearing

Adult beetles and larvae can be collected for redistribution from spring to summer. Either collect by hand or beat the plants' foliage (see Appendix 1 for techniques). Collect at least 500 beetles for each planned release site. Prior to redistribution, beetles can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

Alternatively, these beetles are easy to rear in insect proof cages containing potted blue heliotrope. On average, ten large plants can fit in a 1.2 × 0.6 m cage which will support the progeny of five to seven egg laying females. Assuming 50% of beetles are female; add 10 to 14 adult beetles to each cage. Because larvae are voracious feeders, they can defoliate plants within a few days, so it is important to regularly monitor and replace their food source. Keep plants well-watered as water-stressed plants potentially can stop the beetles breeding.

Collect pea-sized and larger larvae for redistribution, leaving adults to continue to reproduce to their full potential. Larvae are best collected by hand using soft touch forceps, which are available from an entomological equipment supplier. As per field collected material, larvae may be stored temporarily prior to release.

R. Holtkamp

Handling blue heliotrope leaf-beetle larvae with soft touch forceps.

R. Holtkamp

Rearing setup of the blue heliotrope leaf-beetle.

How and when to release

Release collected beetles (adults and larvae) directly onto healthy plants as soon as possible. To assist with establishment, consider making multiple releases at a single site over a season. As populations of beetles are less likely to build up on water-stressed plants, be sure to regularly water the plants at your release/ nursery site.

Field cages can also assist establishment by eliminating predators and containing beetles to restrict dispersal prior to mating and egg production. Ensure you regularly check that there is sufficient food supply. Simply move the cages around locally on a regular basis to achieve this.

Adults can be released whenever they are present over the warmer months. Cages are best used if trying to build populations in the late summer or early autumn so that the population is contained as the pupae overwinter. Cages should be removed the following spring once there is evidence of egg and larval production.

Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for leaf-beetle presence (adults and larvae) and feeding damage (chewed leaf edges) at the nursery site within one year of release. Be sure to examine the underside of leaves for egg clusters and or larvae. Record their presence or absence as per your monitoring guidelines (Appendix 3) and if present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Bridal creeper (common form)

Asparagus asparagoides

Bridal creeper is a scrambling, perennial vine native to South Africa (Parsons and Cuthbertson, 2001). It thrives in the warm temperate climates of Australia in habitats ranging from coastal heath to sandy dunes (including woodland, forest and natural areas) and moist gullies (Parsons and Cuthbertson, 2001; Morin and Scott, 2012). Bridal creeper has soft and shiny, broadly ovate, grass green cladodes (flattened, leaf-like stems 10 to 70 mm long), sweet smelling white flowers and pea-sized berries that are readily eaten and dispersed long distances by birds, other animals and water. It's twisting climbing stems (up to 4 m long) branch extensively while below-ground rhizomes (stems) and tubers form thick root mats that can constitute up to 90% of the plant's biomass (Raymond, 1999). With the onset of hot weather, above-ground biomass often dies back but new shoots will emerge from the below-ground root system in autumn, allowing bridal creeper to persist year-round, withstand disturbance and outcompete native species. Flowers appear in early spring. Green berries turn pink then red/burgundy in late spring-early summer. Over 1000 berries can be produced per square metre with seeds ready to germinate from depths of up to 10 cm. Seeds can remain viable for at least three years.

Recorded as a common garden plant in the 1870s, and likely introduced to Australia in the mid-1800s as a garden ornamental plant, bridal creeper was popular in floral arrangements, particularly wedding bouquets. First recorded as naturalised in 1886 in

P. Turner

Bridal creeper infestation.

Victoria, bridal creeper rapidly spread to become a significant and problematic environmental weed across southern Australia (Parsons and Cuthbertson, 2001; Morin *et al.*, 2002). Declared as a Weed of National Significance in the late 1990s, bridal creeper's dense foliage can dominate and smother understory vegetation in both disturbed and undisturbed systems.

Australia introduced a pathogen and two insect species as biocontrol agents for bridal creeper (Morin and Scott, 2012). From 1999 to 2002 a rust fungus (*Puccinia myrsiphylli*), leafhopper (undescribed from the tribe Erythroneurini) and leaf beetle (*Crioceris* sp.) were released and established. However, the leaf beetle established poorly and there is currently no scope to redistribute it. Further collection and release details are therefore not provided within this manual.

Recommendation

Widely released and redistributed across southern Australia, the rust fungus and leafhopper are having an effective impact, and bridal creeper is in decline. Redistribution is unnecessary, and only recommended at specific sites (e.g. isolated heavy infestations of bridal creeper). Speak to your local weed or biosecurity officer to make this assessment.

Be on the lookout for incursions of Western Cape bridal creeper in south-west Victoria and south-east South Australia. While under containment and in some parts under eradication, this form of bridal creeper (potentially a different species) is resistant to the bridal creeper rust fungus and if present could potentially reinfest vegetation where bridal creeper is suppressed. Report any suspected infestation to your local weed or biosecurity officer.

To differentiate, do not just rely on above-ground features because in ideal conditions bridal creeper seedlings and leaves (cladodes) can appear similar to the Western Cape form. Dig up the tubers to confirm its identity. Refer to page 9 of the NSW Office and Environment and Heritage (2013) Asparagus weeds management manual (available from https://www.environment.nsw.gov.au/-/media/OEH/Corporate-Site/Documents/Animals-and-plants/Pests-and-weeds/asparagus-weeds-management-manual-130486.pdf) which compares the two forms of bridal creeper.

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Bridal creeper fruits (top) and flowers (bottom).

Bridal creeper rust fungus
Puccinia myrsiphylli

First released in 2000, and widely redistributed in partnership with the community across southern Australia, the bridal creeper rust fungus is now widespread throughout bridal creeper's invasive range (Morin and Scott, 2012). The rust has provided a substantial reduction of biomass and shoot production in areas of high humidity, but impacts are minimal in dry inland infestations. Infecting the leaves and stems, the rust is able to obtain nutrients from the plant, thereby limiting its resources to produce stems and fruits. It also depletes tuber reserves. Destroying leaf tissue, the rust causes severely diseased plants to also shed their infected cladodes (leaves). Rust impacts improve over several years as the pathogen exhausts bridal creeper's below-ground biomass. Combining the rust with the leafhopper provides further plant stress, enhancing control.

Identification

Symptoms of the rust are predominately seen between July and September when the plant is actively growing, flowering, and fruiting. The rust appears as yellow, circular areas on the upper sides of cladodes with corresponding orange pustules surrounded by yellowing tissue on the undersides. As the rust destroys plant tissue, severely diseased plants will shed their infected cladodes prematurely and fruiting can be prevented. Over consecutive years of reinfestation, the rust exhausts bridal creeper's below-ground biomass.

Life cycle

The rust completes its entire life cycle on bridal creeper and is comprised of five different spore states representing the asexual and sexual components of the life cycle. The two most commonly seen spore stages include the wind dispersed, orange coloured, asexual urediniospores and the brown black, 'over-summering', sexual teliospores found on infected dead foliage. These teliospores remain dormant but viable until rain and cooler temperatures trigger the rust to produce new infective spores. This occurs around autumn and corresponds with new shoot growth. During this time, the rust infects the plant and produces urediniospores that spread easily by wind to infect neighbouring bridal creeper plants. These spores are the most infectious stage of the rust's life cycle. Peak spore production is usually between July and September. To infect the cladodes, spores enter through stomata (breathing pores) on the lower surface of the cladodes.

S. Ivory, SARDI

Bridal creeper cladode with orange urediniospores.

Field collecting and redistribution

This rust is widespread, and redistribution is largely unnecessary except for specific sites of isolated and dense bridal creeper infestations with high humidity. Speak to your local weed or biosecurity officer for advice prior to redistributing the rust, keeping in mind you may also wish to redistribute the leafhopper at your site concurrently.

If redistribution of the rust is suitable for your site, two methods are available including:

- Method 1: the rust fungus redistribution method described below and developed by CSIRO to establish nursery sites, or
- Method 2: the spore water method detailed on page 174 Appendix 1. Noting infection is dependent upon seasonal conditions and moisture or humidity is required (with foliage remaining moist for at least 8 hours).

Rust spores are not toxic but could cause irritation to people hypersensitive to pollens. As a precaution when handling rust-infected foliage, wear safety equipment including goggles, a respiratory mask, and gloves.

Method 1. Rust fungus redistribution method

The first signs of rust appear in autumn but collection for redistribution is more effective when spore production is at its peak. The NSW Office and Environment and Heritage (2013) Asparagus weeds management manual (available from https://www.environment.nsw.gov.au/-/media/OEH/Corporate-Site/Documents/Animals-and-plants/Pests-and-weeds/asparagus-weeds-management-manual-130486.pdf) details the following:

1. Cut approximately a dozen, 30 cm long, infected stems of bridal creeper that have well-developed sporulating pustules and place in a paper bag to move to the new release site.
2. A dozen infected stems are required to inoculate a 1 to 2 m^2 bridal creeper infestation on either the ground, or the equivalent amount of foliage climbing up a bush or tree.
3. Releases should be made at the end of the day to avoid hot temperatures.
4. Rub infected foliage onto healthy foliage in the field by sliding infected foliage back and forth to dislodge spores from pustules to be deposited on the under surface of healthy cladodes.
5. After inoculation, mist inoculated field plants with water.
6. Cover the area with a sheet of clear plastic held in place with sticky tape, rocks or pegs to provide a humid environment for 16 to 24 hours, or overnight.
7. If the site is in full sun, the plastic sheet should be removed the next morning to prevent plants heating up or burning.
8. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Follow-up monitoring of the release sites should take place a month after inoculation to check for signs of infection. Look for signs of rust, indicated by leaf yellowing or chlorosis and orange pustules on the underside of leaves. If monitoring coincides with summer, look for brown to black spores on dead stems and leaves. If rust is present, begin monitoring for dispersal at incremental distances away from each nursery site as per your monitoring guidelines (Appendix 3). If there is no sign of the rust within two months of inoculation, consider inoculating the plants again if time permits before the onset of summer.

P. Turner

P. Turner

Impact of the rust on bridal creeper before (top) and after the release of the rust (bottom).

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The bridal creeper rust fungus attacks leaves and stems, reducing the amount of green plant material.

P. Sullivan

Early rust infection.

Bridal creeper leafhopper
Erythroneurini tribe – undescribed species

First released in 1999, and widely redistributed in partnership with the community across southern Australia, the bridal creeper leafhopper is now widespread (Morin and Scott, 2012). Feeding damage from adults and nymphs sucking on cell contents causes cladodes to whiten and fall off. Continual damage over several seasons exhausts tuber reserves and production. This reduces the competitiveness of bridal creeper. Leaf hopper populations are seasonal and can be further influenced by parasitism and bridal creeper availability. As a result, impact can vary from limited to good. By releasing both the leaf hopper and the rust fungus at any one site, the combined stress and impact on the plants is enhanced.

Identification

The adult leafhopper is approximately 2.5 mm long and yellowish-white. The nymphs get progressively larger and whiter over their five growth stages

CSIRO

Bridal creeper leafhopper.

S. Ivory, SARDI

Leafhopper damage.

(instars). Fragile first instar nymphs are 0.8 mm long with soft yellow bodies. Wing buds appear at the third instar stage and identification between the sexes is possible at the fifth instar (females develop a dark brown ovipositor, and males are shorter with genital claspers). Eggs are initially transparent and oblong and transition to a deep yellow. When nymphs are ready to hatch, red eyespots are visible. Feeding damage typically inflicted by both adults and nymphs appears as small white to yellowish flecks (chlorosis) on the top of leaf cladodes, that develop into merged zig-zag patterns causing cladode discolouration. Complete discolouration can occur, leaving cladodes completely white.

Life cycle

The leafhopper can have several generations per year and they tend to breed more quickly at higher temperatures, continuing to breed year-round with bridal creeper present. The adult female lays around 180 eggs on the underside of leaves in her six to eight-week lifespan. Within two weeks, eggs hatch and nymphs begin to feed on the underside of the same cladode (from egg to adulthood) unless they are disturbed or they run out of food. During this time, nymphs undergo five growth stages that take approximately two weeks before they become winged adults. Feeding on cell content, with their piercing and sucking mouthparts, increases with each nymphal moult, with the greatest damage caused by adults.

Field collecting and rearing

Redistribution is unnecessary, and only recommended at specific sites (e.g. heavy infestations of bridal creeper and potentially with year-round, above-ground foliage) to boost management in some years. Keep in mind that if your site is suitable, the leafhopper in combination with the rust can achieve greater impact. Speak to your local weed or biosecurity officer to assist you with your decision.

Collection is easy because the leafhopper has multiple generations per year and they are widely distributed. They can be collected by harvesting infested foliage, or for adult leafhoppers, by using a sweep net (see Appendix 1 for technique), during autumn and winter. Collect at least 100 adults for each release site. Alternatively, to collect both adults and nymphs, cut a large bunch of heavily infested foliage (as nymphs are mostly stationary) in the morning when cool, and place contents into a large plastic bag. Seal the bag and keep it out of the sun. Prior to redistribution, leafhoppers can only be stored temporarily with air holes for ventilation (i.e. a day or so at cool temperatures of around 15°C).

How and when to release

Release collected adult leafhoppers directly onto healthy plants as soon as possible. For infected plant material, tease apart foliage and spread the collected material thinly over the bridal creeper infestation while pushing infected foliage into the infestation. Invert the plastic bag and shake out any insects onto the bridal creeper infestation. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for leafhopper presence (adults and nymphs on underside of cladodes) and damage (zig zag pattern of white spots or white cladodes) at the nursery site within one year of release and record its presence or absence as per your monitoring guidelines (Appendix 3). In the year or two immediately following release, the leafhopper may not always be easy to find as they may have moved several metres from this release point. You may need to have a good look around on nearby plants. If the leafhopper is present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Bridal creeper leaf beetle
Crioceris sp.

The leaf beetle was introduced from South Africa in 2002. Both adults and larvae feed on the developing shoots and cladodes. It established at only three of 82 release sites, possibly due to predation or parasitism (Morin and Scott, 2012). It is unknown if it has survived at the sites where it initially established.

CSIRO

Bridal creeper leaf beetle.

Field collecting and rearing

The leaf beetle is currently not available for redistribution due to limited establishment.

How and when to release

The agent is not available for release.

Monitoring for natural dispersal

Record your leaf beetle sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually (Appendix 3).

S. Ivory, SARDI

Bridal creeper leaf beetle larva.

Cacti

Invasive cacti are a group of over 30 introduced perennial succulent species from the cactus tribes Cacteae, Opuntieae and Pereskieae. The most problematic of these are species within genera *Opuntia* (e.g. the famous prickly pears), *Cylindropuntia* and *Harrisia*. Biocontrol has been a highly successful integrated management tool used in managing invasive cacti in Australia and globally.

HANDLE WITH CARE!

Invasive cacti must be handled with care as their spines can cause significant injuries. Wear PPE to prevent spine and bristle injury to skin and eyes. Use long-handled barbecue tongs when handling cacti. Refer to the manual Managing Opuntioid Cacti in Australia – Chapter 3 'Safety and Welfare' (available from https://www.agric.wa.gov.au/invasive-species/opuntioid-cacti-best-practice-control-manual).

Prickly pears *Opuntia* spp.

The term 'prickly pear' broadly describes several species in the *Opuntia* genus. Native to the Americas, earliest records indicate that prickly pears came to Australia with the first fleet (1788). While many were introduced as ornamental plants (*Opuntia stricta*), others including *Opuntia monacantha* were introduced to establish a cochineal dye industry[1].

Many of these species quickly became widespread and out of control causing significant impacts to agriculture and the environment.

Prickly pears typically grow as shrubs, or occasionally as trees. Their flattened stems (called cladodes – which replace leaf function) distinguish them. Each cladode is capable of vegetative reproduction and along with seed, assists the plants with successful dispersal. Prickly pears use modified tissue to store water. This helps them survive periods of drought, allowing for successful invasion into the semi-arid regions of Australia. In 2012, all *Opuntia* and *Cylindropuntia* species (except the Indian fig, *Opuntia ficus-indica*) were listed as Weeds of National Significance due to their invasiveness and impacts across all mainland states and territories in Australia. Long before this time, however, biocontrol of prickly pears began.

From 1911 to 1939, over twenty species of biocontrol agents were released on prickly pears with 14 establishing. Control of prickly pears was ultimately achieved with the Cactoblastis moth (*Cactoblastis cactorum*) and cochineal insects (*Dactylopius* spp.). For the rest of this section, the different cochineal species (and their respective lineages) will largely be discussed as a single entity due to similarities in biology and impact. Any differences will be specified where required.

Recommendation

Use cochineal insects for the control of many prickly pear species.

For *Opuntia* spp., it is recommended to use cochineal insects and the Cactoblastis moth in combination, as they coexist well in the field and their damage is complementary. Using them in combination also minimises the chance of vegetative re-growth.

[1] Prickly pears are hosts to cochineal insects which produce a reddish-purple (carmine) dye, which is used for protection against predators. The dye is used in the production of cosmetics, drugs, food and textiles.

Cochineal insects
Dactylopius spp.

Cochineal are soft-bodied scale insects that solely feed on cactus plants, especially species of *Opuntia* and *Cylindropuntia*. *Dactylopius ceylonicus* was the first cochineal introduced into Australia, by Captain Arthur Phillip with the First Fleet, with the sole purpose of establishing a cochineal dye industry (particularly to colour the British army's red coats). In 1914, this same species was released as a biocontrol agent for drooping tree pear (*O. monacantha*) (Winston *et al.*, 2014). Since then, an additional six cochineal species from the Americas have been released in Australia for the control of a variety of cactus species. Five of the six cochineal species established. Some cochineal species (especially *Dactylopius tomentosus*) have different lineages[2] that target the different *Opuntia* and *Cylindropuntia* species. The different lineages show great variation in their impact, so it is important to use the correct virulent lineage for each target species. Cochineal-infested prickly pear cladodes usually wither and die within three years. More rapid control is usually achieved in drier years. Predation of cochineal insects by ladybirds and lacewings can reduce field populations.

The correct cochineal species (or in some cases lineage) must be matched to its target prickly pear species, otherwise successful control will not be achieved.

[2] Lineages are populations of the same insect species (e.g. *Dactylopius tomentosus*) that can only be separated by their different abilities to feed, lay eggs and develop on a target species. Only molecular tools can distinguish between different lineages.

P. Sullivan

Dactylopius ceylonicus cochineal colonies on an Opuntia monacantha pad.

P. Sullivan

Adult winged males of Dactylopius ceylonicus.

P. Jones

First instars (nymphs) of Dactylopius tomentosus (crawlers).

Identification

Cochineal insects are soft-bodied, oval-shaped and deep red. Newly emerged nymphs called crawlers are approximately 0.5 mm long. The wingless adult females (up to 5 mm long) are hidden beneath a white, waxy, wool-like covering. The seldom-observed adult males have two wings, do not feed, and live only long enough to mate with females.

A. McConnachie

Egg mass of a female Dactylopius tomentosus.

A. McConnachie

Egg mass dewaxed.

Life cycle

The cochineal insect has an unusual life cycle. The male has four main life stages (egg, nymph, pupa and winged adult) while the female only has three life stages (egg, nymph and adult). The egg stage is usually brief with eggs from most species hatching within a couple of days, although eggs from *Dactylopius tomentosus* hatch in about 17 days. Nymphs produce long wax filaments. On warm days with a gentle breeze, nymphs 'crawl' to the edge of cladodes and wait for the wind to catch the wax filaments and carry them to new host plants. Nymphs then establish protected feeding sites on new hosts, such as adjacent to the base of spines or where cladodes join one another. Female cochineal insects only disperse as first instar nymphs for a few days and then settle in the one spot for the rest of their lives feeding actively on cell contents. Male cochineal insects can disperse as nymphs and winged adults but only feed until they reach sexual maturity. Cochineal insects have a generation time of 30 to 45 days at 25 to 30°C (Sullivan, 1990).

Field collecting and rearing

Cochineal insects can be easily collected from nursery sites (see release site selection on page 7) by picking infested cladodes with long-handled tongs (or a spade) and placing them into a sturdy cage or box for transportation. Only collect cochineal from the same prickly pear species as the target species as it is important to use the correct virulent lineage of the cochineal. Ensure that predators such as ladybirds and lacewing larvae are not collected with the cladodes.

Cochineal insects are easy to rear. Place several cochineal-infested cladodes into a box or cage together with uninfested cladodes of the same species. Rear the cochineal in a dry area that has warm temperatures of 25 to 27°C. At these temperatures, the newborn nymphs will infest all cladodes quickly and the cochineal-infested cladodes should be ready for redistribution in about six weeks.

How and when to release

For *Opuntia* species, the cochineal-infested cladodes should be placed among cladodes of the target population where they are protected, out of direct sunlight and covered with a few broken-off cladodes. This covering will protect the cochineal adults and nymphs from extreme weather events, such as heavy rainfall, where they could easily be damaged by raindrops.

For *Cylindropuntia* species, the cochineal infested cladode should be placed high up in the plant, and if necessary, barriers or baits can be used to minimise ant predation.

Infested cladodes for all species should be placed on plants growing along the upwind side of a release site to assist with the wind dispersal of the nymphs. Place cladodes near one another due to the relatively slow dispersal rates of some cochineal species (dispersal varies from 15 to greater than 100 m per year). For tiger pear (*Opuntia aurantiaca*), place the cochineal infested cladodes at intervals of every 10 m. For other *Opuntia* species, place the cochineal infested cladodes at intervals of 10 to 50 m, and for *Cylindropuntia* species place the cochineal infested cladodes at intervals of 10 to 30 m. Releases should be made in spring or summer as populations build up quickly in the warmer months before decreasing in winter. It is best to avoid making releases during times that are likely to experience heavy rainfall.

Monitoring establishment and dispersal

Look for the white, waxy-wool covered female cochineal on both sides of cladodes, especially adjacent to spines and between cladodes, within one year of release. In higher rainfall areas, the cochineal will often be more abundant on the lower (protected) side of cladodes. Record the presence and absence of the cochineal as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the provided guidelines. Monitor annually.

A. McConnachie

Cochineal crawlers (Dactylopius tomentosus) moving up the spines of white-spined Hudson pear ready for wind dispersal.

Cactoblastis moth
Cactoblastis cactorum

The Cactoblastis moth from Argentina was first released in Australia in 1926 (Winston *et al.*, 2014). The Cactoblastis moth, along with *Dactylopius opuntiae*, was instrumental in controlling common prickly pear (*Opuntia stricta*) on 25,000,000 ha of land in Queensland and New South Wales between 1925 and 1932, resulting in previously abandoned farms being reclaimed and brought back into production. This is considered an example of one of the world's most successful weed biocontrol programs.

P. Sullivan

Velvet tree pear heavily infested with the cochineal (Dactylopius opuntiae).

P. Sullivan

Impact of the Cactoblastis moth on common prickly pear.

Cactoblastis moths feed on many *Opuntioid* species, giving excellent control of common prickly pear (*Opuntia stricta*) and creeping pear (*Opuntia humifusa*) throughout Australia. Entire plants are often destroyed but any uneaten cladodes can grow vegetatively into new plants. To prevent this from occurring it is best to combine the Cactoblastis moth with a cactus species appropriate cochineal agent; because cochineal species are likely to kill uneaten cladodes left by the moth. Keep in mind that heavy rain or cold weather can inhibit the effect of both agents (e.g. the coastal belt or cold microclimates created from shaded hillsides, thickly timbered areas or south-facing slopes).

Identification

The seldom-seen Cactoblastis moth adult is brownish-grey with darker bands on their wings. They have an average wingspan of 23 to 40 mm. The newly-hatched larvae are yellow-pink and transition to bright orange with distinctive black spots or bands.

Life cycle

Female moths lay their eggs in the form of a chain or egg stick (each contain up to 100 eggs and reach around 25 mm in length), the first of which is attached onto the end of spines or cladodes. The larvae then burrow quickly into the cladode to feed within, reducing it to a rotting mass. Pupation often takes place inside the cladode. The moth has approximately two overlapping generations per year in colder areas but can have more than three generations per year in warmer areas.

Field collecting and rearing

The Cactoblastis moth is widespread and usually does not need redistribution. However, egg sticks or cladodes containing larvae can be collected for redistribution, especially if new infestations are some distance away from existing populations of the moth. Moving the insect around will speed up the rate of establishment. You can transfer the egg sticks to new plants, but egg sticks can be hard to find. Because the moths are so mobile, an easier and more reliable method to assist movement is achieved by creating a series of cladodes stacks to attract Cactoblastis moths during peak activity in the warmer months (spring to

P. Greb, USDA

Cactoblastis moth (Cactoblastis cactorum).

P. Sullivan

Cactoblastis moth larvae (Cactoblastis cactorum) on common prickly pear.

J. Hosking

Egg stick of the Cactoblastis moth (Cactoblastis cactorum) on common prickly pear.

summer). Common prickly pear cladodes are piled on top of one another (around 2 × 2 m and 1 m high) and act as a beacon to the moths thereby enabling faster reproduction, population increase, and plenty of food for hatching larvae. Don't make the stacks too big, because the lower plant material can begin to rot under the weight.

Alternatively, physically attaching infested cladodes to healthy plants, in a similar fashion to that used to release cochineal species, can also achieve good results.

How and when to release

Due to its high mobility, the Cactoblastis moth will usually find common prickly pear stacks by themselves. To hasten population increase, add Cactoblastis larva-infested cladodes to the top of these stacks. While physically transferring cladodes containing larvae to uninfected plants can work well, ants can be a problem by attacking larvae through broken areas of cladodes. If necessary, barriers or baits can be used to minimise ant predation but always place infested cladodes as high up as possible in the plant.

Developing stacks or transferring infested cladodes should be done in spring or summer as populations build up quickly in the warmer months before decreasing in winter. It is best to avoid making releases during times that are likely to experience heavy rainfall.

Monitoring establishment and dispersal

Look for bone-coloured, hollowed-out cladodes with a leathery, translucent epidermis that hardens and becomes opaque several months later – this will alert you to the presence of the Cactoblastis moth. Record the presence or absence of the moth as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the provided guidelines. Monitor annually.

Boxing glove cactus *Cylindropuntia fulgida* var. *mamillata*

Boxing glove cactus (also known as coral cactus) from North America is an erect shrub (up to 1 m) and, as its name suggests, it has cylindrical, club-shaped, wavy or boxing glove-shaped cladodes. Introduced in the 1980s, it is now naturalised in drier inland areas of Australia. First released in 2016, the cochineal insect *Dactylopius tomentosus* ('cholla' lineage) is already achieving an excellent kill rate (>95%) within two years in Queensland and New South Wales. Cochineal redistribution is required for new infestations, particularly large ones. This cochineal lineage can disperse up to several hundred metres per year through dense, continuous boxing glove cactus infestations. Control is not immediate and can take several years.

Recommendation

Use *Dactylopius tomentosus* 'cholla' lineage to achieve good control of boxing glove cactus.

H. Rutherford

Boxing glove cactus.

Hudson pear
Cylindropuntia pallida, Cylindropuntia tunicata

Hudson pear describes two species from North America; the white-spined Hudson pear *Cylindropuntia pallida* (syn. *C. rosea*) which grows to 1.5 m with pink flowers and the smaller (to 0.6 m) brown-spined Hudson pear (*Cylindropuntia tunicata*) with yellowish-brown flowers. Both species were first detected in Australia in the 1960s. A different lineage of *Dactylopius tomentosus* has been released for each Hudson pear species. *D. tomentosus* 'californica var. parkeri' lineage should only be used to control the white-spined Hudson pear, while *D. tomentosus* 'acanthocarpa var. echinocarpa' lineage should only be used to control the brown-spined Hudson pear. Both lineages of *D. tomentosus* are providing good control for white- and brown-spined Hudson pear (A. McConnachie pers. comm., 2020).

Recommendation

It is not possible to physically differentiate between the two *Dactylopius tomentosus* lineages for the white- and brown-spined Hudson pears. Source the cochineal from your weed or biosecurity officer rather than field collecting.

S. Potter

R.J. Chinnock

A. McConnachie

Brown-spined Hudson pear (top), its fruit (middle) and impacted by Dactylopius tomentosus 'acanthocarpa var. echinocarpa' lineage (bottom).

P. Sullivan

Brown-spined Hudson pear on the left compared to white-spined Hudson pear on the right.

A. McConnachie

White-spined Hudson pear impacted by Dactylopius tomentosus 'californica var. parkeri' lineage.

Jumping cholla
Cylindropuntia prolifera

Jumping cholla from North America is an erect shrub (up to 1.5 m) with brown to dark-brown spines and green fruit (Sheehan and Potter, 2017). Released in 2018, the same cochineal lineage used for the white-spined Hudson pear (*Cylindropuntia pallida*) (*Dactylopius tomentosus* 'californica var. parkeri') is providing good control (A. McConnachie pers. comm., 2020).

Recommendation

Source the cochineal *Dactylopius tomentosus* 'californica var. parkeri' lineage from your local weed or biosecurity officer to control jumping cholla (*Cylindropuntia prolifera*).

S. Potter (left), R.J Chinnock (right)

Jumping cholla (left) and its flowers (right).

A. McConnachie

Jumping cholla impacted by Dactylopius tomentosus ('californica var. parkeri' lineage).

Klein's cholla
Cylindropuntia kleiniae

Klein's cholla from North America is a straggly shrub (up to 2.5 m) (Sheehan and Potter, 2017). The cochineal *Dactylopius tomentosus* 'imbricata' lineage is approved for release against Klein's cholla, however, it has not yet been field released in Australia. The original *D. tomentosus* lineage released against rope pear (*Cylindropuntia imbricata*) is currently providing good control.

Recommendation

Use the original *Dactylopius tomentosus* lineage released on devils rope (*Cylindropuntia imbricata*) to control Klein's cholla (*C. kleiniae*).

P. Sullivan (left), S. Potter (right)

Klein's cholla (left) and its flowers (right).

A. McConnachie

Klein's cholla impacted by Dactylopius tomentosus.

Pencil cactus
Cylindropuntia leptocaulis

Pencil cactus from North America (also known as pencil pear) is a spreading shrub (up to 1.8 m) (Sheehan and Potter, 2017). The cochineal, *Dactylopius tomentosus* 'cylindropuntia' lineage, is approved for release on pencil cactus. However, the original *D. tomentosus* lineage released against rope pear (*Cylindropuntia imbricata*) is providing effective control of *C. leptocaulis*.

Recommendation

Use the original *Dactylopius tomentosus* lineage released on rope pear (*Cylindropuntia imbricata*) to achieve good control of pencil cactus.

S. Potter (left), R.J Chinnock (right)

Pencil cactus (left) and its flowers (right).

A. McConnachie

Pencil cactus impacted by Dactylopius tomentosus.

Rope pear
Cylindropuntia imbricata

Rope pear from North America (also known as devil's rope) is an erect shrub or small tree (up to 3 m) (Sheehan and Potter, 2017). This species was the first *Cylindropuntia* species targeted by a biocontrol agent. First released in 1925, *Dactylopius tomentosus* is still effective today. However, a new 'cylindropuntia' lineage has also been released to complement and improve current biocontrol of rope pear. While current control can take several years, felling or cutting back larger plants and stacking the cut segments after the cochineal has established will accelerate control. This is because the stacked material provides a protective environment for the cochineal to flourish.

Recommendation

Complement the original lineage of *Dactylopius tomentosus* released in 1925 with the cochineal *D. tomentosus* 'cylindropuntia' lineage to achieve good control.

P. Sullivan

P. Sullivan

Rope pear (top) and its flowers (bottom).

Snake cactus
Cylindropuntia spinosior

Snake cactus from North America is an erect shrub (up to 3 m) which often forms patches several metres wide. It is similar in appearance to jumping cholla (*C. prolifera*) except that it has yellow fruit and white to grey spines (Sheehan and Potter, 2017). Released in 2018, the cochineal *Dactylopius tomentosus* 'bigelovii' lineage is established and its impact is currently being monitored.

Recommendation

Source the cochineal *Dactylopius tomentosus* 'bigelovii' lineage from your local weed or biosecurity officer.

P. Sullivan (left), R.J. Chinnock (right)

Snake cactus (left) and its flowers (right).

A. McConnachie

Snake cactus impacted by Dactylopius tomentosus ('bigelovii' lineage).

Harrisia cactus
Harrisia martini, Harrisia pomanensis, Harrisia tortuosa

Harrisia cactus, native to South America, is a sprawling perennial shrub (up to 1 m) with large white funnel-shaped flowers growing singly at the end of stems. Introduced as a garden ornamental plant between 1885 and 1900, it readily established throughout central Queensland, New South Wales and Western Australia (Parsons and Cuthbertson, 2001). The large bright red fruit when split open reveals sugary sweet-coated seeds that are attractive to animals. It also spreads readily via its stems, which can set root if touching the ground.

Recommendation

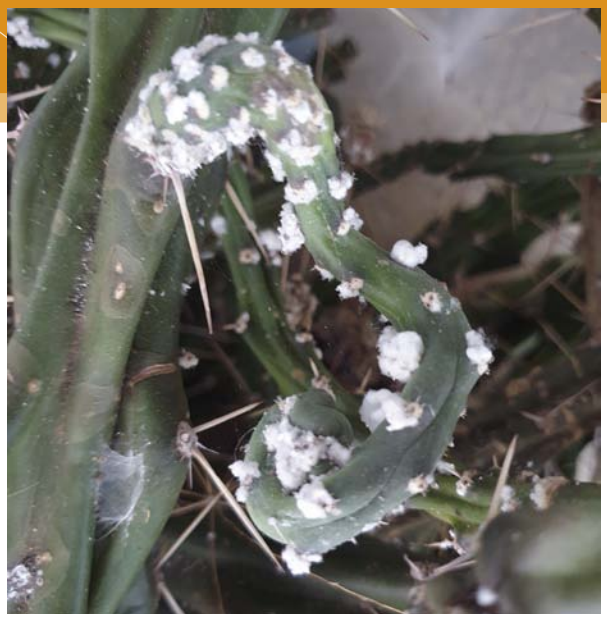
Use the mealybug *Hypogeococcus festerianus* for control of dense harrisia populations.

R. McFadyen

Hypogeococcus festerianus infesting harrisia cactus.

P. Sullivan

Harrisia cactus.



R. Holtkamp

Harrisia cactus infested with Hypogeococcus festerianus (mealy bug).

Four species of insects were tested and released as biocontrol agents for Harrisia cactus; two established and one, the cactus mealybug (*Hypogeococcus festerianus*), rapidly achieved good control in the in the northern areas but was less effective in the southern range (McFadyen, 2012c). Due to their limited establishment and impact, the other agents will not be discussed further in this section.

Cactus mealybug
Hypogeococcus festerianus

The cactus mealybug from Argentina was first released in Australia in 1975 (Winston *et al.*, 2014). The cactus mealybug usually lives in colonies and feeds on stem tips and buds where it causes deformities that limit plant growth. These deformities result in the knotting of the plant stem and are where mealybugs live and feed on stem tips protected from predators. Dense infestations of the mealybug result in woolly masses on the tips of stems. Flowering and fruiting are immediately impacted, causing affected plants to draw on tuber reserves until depleted, resulting in plant death. The impact of the cactus mealybug is greater in wetter conditions, than dry. This is because under dry conditions, plant tubers become dormant. Consequently, the plant does not draw on tuber reserves in dry conditions resulting in energy reserve depletion and plant death. With no new plant growth the mealy bug is at risk of dying out.

Identification

Cactus mealybugs are soft-bodied, tiny oval-shaped insects. The wingless females (about 3 mm long) are similar in appearance to cochineal insects being reddish-brown under a white, waxy, wool-like covering. Adult males do not resemble females, are seldom seen and are winged.

Life cycle

The mealybug has multiple generations each year, with an egg to adult generation time of around two months in northern Australia and a little longer in southern Australia (McFadyen, 1979). Adult females lay up to 100 eggs in their lifetime with each hatching in less than 20 minutes of laying (McFadyen, 2012c). The newly emerged nymphs, called crawlers, actively 'crawl' over the plant for around 24 hours before settling in protected sites or being dispersed by wind. Males disperse to mate with virgin females, whereas females reach maturity within one month of hatching and remain fixed to the one spot for their lifespan. They continue to breed throughout the winter, but at a slower rate than in summer.

Field collecting and rearing

Although rearing is relatively easy, it is easier to redistribute the cactus mealybug by field-collected infested cuttings. To collect the bug, cut infected stems around 15 cm below distorted stems and store in boxes temporarily (up to five days) prior to redistribution. Collections for redistribution are best during spring and early summer when mealybug populations are at their peak. The mealybug disperses slowly, so redistribution of the cactus mealybug-infected cuttings should be done at intervals of no greater than 50 m. To ensure rapid reproduction and dispersal collect as many stems as there are plants for your site.

How and when to release

Place infested stem cuttings onto actively growing plants. Avoid placing them on stressed or dried out plants. For effective dispersal, small plants require one distorted stem, whereas larger plants require at least three. Record your release as per the release guidelines (Appendix 2). Make releases between September and December.

Recommendation

Release the mealybug *Hypogeococcus festerianus* on harrisia plants in full sun (in preference to those in shade) to achieve more rapid establishment.

Monitoring establishment and dispersal

By the end of the first summer look for the knotted stem tips and then examine the plant and knobs for the presence of mealybugs. Good control is expected within four years. Monitor establishment and dispersal as per supplied guidelines (Appendix 3) and supply your weed or biosecurity officer with a copy.

Common prickly pear
Opuntia stricta

Common prickly pear (also known as common pest pear) from the Americas is a sprawling, erect shrub (up to 1.5 m) with yellow flowers (60 mm in diameter). Introduced to Australia in the early days of settlement, it exists today as many different forms (Hosking, 2012).

Recommendation

For effective control, use the cochineal *Dactylopius opuntiae* 'stricta' lineage together with the Cactoblastis moth.

P. Sullivan (left), S. Potter (right)

Common prickly pear (left) and its flowers (right).

The Cactoblastis moth has achieved excellent control of common pear. Redistribution of the Cactoblastis moth is unnecessary, as it is widespread. However, as control can take several years, you can speed it up by integrating your management approach with the cochineal *Dactylopius opuntiae* 'stricta' lineage.

Riverina pear
Opuntia elata

Riverina pear from South America is an erect shrub (up to 2 m) with orange flowers. Spines are generally absent, although some areoles (small bumps on cactus segments) may have one to three short spines present. It is thought to have been introduced into Australia in the 1960s (Sheehan and Potter, 2017). Naturalised populations are recorded throughout most of Australia.

Recommendation

Two species of cochineal, *Dactylopius opuntiae* ('stricta' and 'ficus' lineages) and *Dactylopius ceylonicus*, when combined with the Cactoblastis moth, provide control of Riverina pear.

P. Sullivan

R.J. Chinnock

Riverina pear (top) and its flowers (bottom).

Smooth tree pear
Opuntia monacantha

Smooth tree pear from South America (also known as drooping tree pear) is a small tree (up to 5 m). By the 1840s, smooth tree pear appeared regularly in nursery catalogues throughout Australia after its introduction with the first fleet (1788) (Parsons and Cuthbertson, 2001). The cochineal *Dactylopius ceylonicus* provides good control, which ordinarily takes several years and is slower in high rainfall areas. Felling or cutting back larger plants (>2 m) and stacking the cut segments after the cochineal has established will accelerate control. This is because the stacked material provides a protective environment for the cochineal to flourish. While the Cactoblastis moth and soft rot pathogens (e.g. *Phyllosticta concava*) attack smooth tree pear, overall control is not achieved.

Recommendation

Use the cochineal *Dactylopius ceylonicus* to achieve good smooth tree pear control. Control is likely to take several years.

P. Sullivan

Smooth tree pear (left) and holes from the black spot fungus Phyllosticta concava (right).

Tiger pear
Opuntia aurantiaca

Tiger pear from South America is a low-spreading shrub (up to 0.5 m). It was first recorded in herbarium records in New South Wales in 1883 and was flagged as a potential problem by 1910 due to its widespread and increasing distribution (Sheehan and Potter, 2017). Good control of tiger pear can be achieved using the cochineal *Dactylopius austrinus* and to a lesser extent the two moths *Cactoblastis cactorum* and *Tucumania tapiacola*. The cochineal insect achieves better control than the moths during hot dry summers. During wetter years, biocontrol is less effective because tiger pear produces more cladodes than those destroyed.

Recommendation

Use the cochineal *Dactylopius austrinus* to achieve effective control of tiger pear.

R.J. Chinnock

J. Hosking

Tiger pear (top), tiger pear cochineal and damage (bottom).

Velvety tree pear
Opuntia tomentosa

Velvety tree pear from North America (also known as velvet tree pear) is a small tree (up to 5 m) that can develop a large trunk (up to 0.5 m in diameter). Its introduction history to Australia is unknown, but by 1912, it was a common weed across southern Queensland (Sheehan and Potter, 2017). Today the cochineal *Dactylopius opuntiae* 'stricta' lineage is providing good control. Control usually takes several years but felling or cutting back larger plants (>2 m) and stacking the cut segments after the cochineal has established will accelerate control. This is because the stacked material provides a protective environment for the cochineal to flourish.

Recommendation

Use *Dactylopius opuntiae* 'stricta' lineage to achieve good control of velvety tree pear.

P. Sullivan

S. Potter

Velvety tree pear (top) and its flowers (bottom).

Wheel cactus
Opuntia robusta

In its native country, Mexico, wheel cactus is a rare and endangered shrub or small tree (up to 4 m) (Sheehan and Potter, 2017). Likely introduced to Australia for ornamental purposes in the early 1900s, details relating to its introduction history remain largely unknown. Proclaimed as a serious weed in the 1960s, wheel cactus became widespread across drought tolerant regions of southern Australia. Today the cochineal *Dactylopius opuntiae* 'ficus' lineage provides good control, particularly in South Australia where land managers achieved control in less than four years. While the Cactoblastis moth attacks wheel cactus, it provides limited control.

Recommendation

Use *Dactylopius opuntiae* 'ficus' lineage for good control of wheel cactus. Control is likely to take several years.

P. Sullivan

S. Potter

Wheel cactus (top) and its flowers.

P. Sullivan

Wheel cactus under heavy attack by cochineal.

P. Sullivan

Wheel cactus heavily infested with cochineal.

Other less common prickly pear species and their known cochineal lineage pairings

- ***Opuntia elatior*** – use the cochineal *Dactylopius opuntiae* 'ficus' lineage.

S. Potter

- ***Opuntia engelmannii*** – use the cochineal *Dactylopius opuntiae* 'ficus' lineage.

P. Sullivan

- ***Opuntia ficus-indica*** – use the cochineal *Dactylopius opuntiae* 'ficus' lineage.

S. Potter

- ***Opuntia humifusa*** – unknown but well controlled by the Cactoblastis moth.

D. Campbell

- ***Opuntia leucotricha*** – no agents are available.

S. Potter

- ***Opuntia microdasys*** – no agents are available.

P. Sullivan

- ***Opuntia polyacantha*** – no agents are available.

S. Potter

- ***Opuntia puberula*** – the cochineal *Dactylopius opuntiae* 'ficus' lineage has minimal impact.

P. Sullivan

- ***Opuntia schickendantzii*** – use the cochineal *Dactylopius ceylonicus*.

P. Sullivan

- ***Opuntia streptacantha*** – use the cochineal *Dactylopius opuntiae* 'ficus' lineage (photo shows cochineal infestation).

P. Sullivan

- ***Opuntia sulphurea*** – no agents are available.

P. Sullivan

Cape broom

Genista monspessulana

NSW DPI

Cape broom flowers and foliage.

Cape broom (also known as Montpellier broom) is an erect, evergreen shrub native to the Mediterranean region of Europe (Parsons and Cuthbertson, 2001). Primarily found in temperate regions of southern Australia, Cape broom readily colonises disturbed areas forming high-density infestations (up to 3 m) in pastures, open woodlands, forest margins, grasslands, commercial plantations, and neglected amenity areas. Plants have one main stem with numerous hairy lateral branches, which gives them a spreading-like appearance. Hairy, alternatively arranged, short-stalked leaves have a soft feel to them and consist of three leaflets, with the central one being slightly longer (up to 30 mm long) than the other two. Flowering mostly occurs during late winter, spring and summer with bright yellow, pea-shaped flowers (up to 1.2 cm long) occurring singly or in small clusters (up to nine flowers) in the leaf axils or at the end of the branches. The pea-like silky flattened seed pods (up to 2.5 cm long) transition from green to brown or black as they mature. Each pod contains four to eight small black seeds (around 2 mm in diameter) that are naturally dispersed from the plant by explosive pods flicking seeds short distances (up to 3 m). Once on the ground, seeds are readily moved longer distances by vehicles, machinery, water, birds and other animals. Mature plants produce between 3272 and 12,098 seeds each, with seed banks up to 100,000 per m^2 being recorded under mature plants (Lloyd, 2000).

Introduced as an ornamental plant, particularly for hedging before the 1850s, Cape broom soon became a problematic weed invading a wide range of native

NSW DPI

Cape broom invading eucalypt woodlands.

habitats (sclerophyll forests, open woodlands and grasslands) in addition to forestry, pastures, amenity areas, roadsides and railways throughout the temperate regions of southern Australia. Due to its high seed productivity, long-lived seedbank, and ability to germinate in disturbed and undisturbed soils, Cape broom readily forms monocultures. Its nitrogen fixing ability increases soil fertility, encourages further infestation of broom and other weeds making native regeneration of communities particularly challenging.

With its invasion estimated at over 600,000 ha in southern Australia, Cape broom is recognised as a serious environmental and economic weed occurring in all states of Australia and the Capital Territory (Sheppard and Henry, 2012). In 2012 it was collectively listed with other broom species as a Weed of National Significance due to its dense, impenetrable thickets arising from a long-lived soil seed bank with the potential to become even more widespread.

Australia introduced two species of insects from the Mediterranean region to test their potential as biocontrol agents for Cape broom. However, in 2004, presence of the Cape broom psyllid (*Arytinnis hakani*), spanning an area 40 km to the south and 80 km to the north of Adelaide, was recognised prior to its release. A risk assessment subsequently approved the redistribution of this agent throughout southern Australia. No other agents tested during this time have been released (Sheppard and Henry, 2012).

Recommendation

Areas that experience hot summer temperatures or northern facing slopes are likely to experience diminished impact by the psyllid as it is sensitive to high temperatures. Biocontrol, particularly over summer periods, should be integrated with other control options.

Cape broom psyllid *Arytinnis hakani*

The Cape broom psyllid was found to have established in the vicinity of Adelaide, South Australia, in 2004 and from 2009 to 2014 was redistributed throughout New South Wales, Victoria and Tasmania. It is now widely established throughout southern Australia. The Cape broom psyllid feeds on the sap of the host plant, reducing plant health vigour and seed set. When populations build up, their impact is effective with large sections of plants dying back, occasionally leading to shrub death (Sullivan, 2013).

Identification

Adult Cape broom psyllids are highly mobile, sap sucking insects that are approximately 3 mm long, green with clear wings and two large red eyes. The nymphs get progressively larger and greener over their five growth stages (instars). First instars are 1 mm long, orange and while wingless are still highly mobile on plants. Eggs are tiny (<1 mm long) and cream to orange. Adult and nymph feeding on young shoots causes them to blacken, wither and defoliate. Psyllid presence is further evidenced by their excretion of honeydew, which appears as white, sugary crystals in the growing tips of plants.

S. Ivory, SARDI

Cape broom psyllid.

P. Sullivan

White sugary honeydew psyllid excretion.

P. Sullivan

P. Sullivan

Defoliation by Cape broom psyllid.

Life cycle

Cape broom psyllids can have multiple generations per year and breed more quickly at higher temperatures. The adult female lays up to 200 eggs among young leaves and flower buds during her one to two month lifespan. Overwintering eggs hatch in early spring and complete their development by late spring. After hatching, nymphs feed on young leaves and buds as they move through their five instars before becoming adults. Psyllid populations decline during the hot dry summer months and over winter. During this time, they are usually found as nymphs or adults sheltering in young shoots.

Field collecting and rearing

Rearing is unnecessary. Cape broom psyllids are widespread across southern Australia. Adults and nymphs are best collected from late spring or early summer when Cape broom is actively growing. Hot dry weather should be avoided as adults and nymphs are sensitive to temperatures over 26°C.

Several methods can be used to collect psyllids. Adults and nymphs can be easily collected by lightly beating or shaking the foliage over a tray and looking for green winged adults, or through the use of a sweep net, to collect ideally more than 100 active adults for each release site (see Appendix 1 for techniques). Alternatively, harvest nymphs by pruning off infested plant material (around

20 cm long) and place contents into a large plastic bag. Collect around 10 infested cuttings. Prior to redistribution, leaf hoppers can only be stored temporarily with air holes for ventilation (i.e. a day or so at cool temperatures of around 15°C).

How and when to release

Release collected psyllids directly onto healthy plants as soon as possible (ideally within 24 hours of collection to enhance establishment).

For collected plant material, attach infested cuttings onto healthy Cape broom shrubs with tie wire. To achieve successful establishment, attach one infested cutting for every tenth shrub at your nursery site. Each cutting should contain several hundred individuals, which should be ample to ensure establishment. Nymphs will simply move onto healthy plants to feed, as cuttings dry out and die.

Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Within one year of release look for the white sugary honeydew deposits (they look like big grains of sugar) left on the upper surface of leaves by psyllids as they feed. Closer examination of leaf undersides will likely reveal nymphs and adults. The beating method or sweep netting can assist you with a more detailed monitoring program. Record psyllid presence or absence as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Cat's claw creeper
Dolichandra unguis-cati

Cat's claw creeper is a high climbing, perennial woody vine native to Central and South America and the West Indies (Dhileepan, 2012). It typically thrives in the riparian and rainforest vegetation of the warm-temperate, tropical and subtropical regions of Australia. Plants have a basal pair of lance-shaped leaflets (up to 7 cm long) with a third leaflet that is modified into a three-pronged claw-like tendril that aids in climbing. Stems are thick and woody (up to 15 cm thick) that can climb vertically, while those along the ground can form roots. Large bright yellow bell- or trumpet-shaped flowers are formed in groups of two or three throughout the spring. The long, narrow and flat pods contain many papery, winged seeds suitable for dispersal by water and wind. While seed is plentiful, its viability is low, so cat's claw creeper's main mechanism of persistence is via its vigorous root system which forms tubers (up to 40 cm long) that develop multiple stems (climbing runners) for rapid growth.

QDAF

Aerial and ground invasion of cat's claw creeper in Gympie, Queensland.

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Cat's claw creeper trumpet flowers (top), leaves and hook-like tendrils (middle) and papery seed (bottom).

cat's claw creeper

Two morphologically distinct varieties of cat's claw are recognised in Australia, including the 'short-pod' variety found predominately north of Sydney New South Wales to northern Queensland and the 'long-pod' variety which is restricted to several sites in south-eastern Queensland (Shortus and Dhileepan, 2011). As its name suggests, the 'long-pod' variety has pods around twice the length of the 'short-pod' variety. It also has distinctly larger and hairier leaves than the 'short-pod' variety.

QDAF

Morphological differences between the long- and short-pod varieties of cat's claw creeper.

Cat's claw creeper was introduced as a garden ornamental plant to Australia and was first reported as naturalised in the 1950s (Shortus and Dhileepan, 2011). It has since become a major weed of native forests and riparian areas in eastern Australia due to its ability to climb, smother and kill mature native trees leading to extensive canopy collapse. Understories are also impacted by cat's claw creeper's ability to smother, outcompete and hamper native seedling recruitment (Shortus and Dhileepan, 2011; Dhileepan, 2012). In 2012 cat's claw creeper was listed as a Weed of National Significance due to its environmental and economic impacts, invasiveness and potential to spread.

Australia introduced five species of insects from existing biocontrol programs against cat's claw creeper in South Africa, to test their potential as biocontrol agents. Of these, three agents including, a leaf-feeding tingid (*Carvalhotingis visenda*), jewel beetle (*Hylaeogena jureceki* now known as *Hedwigiella jureceki*) and a leaf-tying moth (*Hypocosmia pyrochroma*) were released and have established in the field (Dhileepan, 2012). All agents released readily feed on both the 'long-pod' and 'short-pod' varieties of cat's claw creeper.

Recommendation

Owing to the abundant tuber reserves of cat's claw creeper, effective control is best achieved by using multiple agents, targeting various parts of the plant.

Cat's claw creeper leaf-feeding tingid *Carvalhotingis visenda*

First released in 2007 in south-eastern Queensland, the leaf feeding tingid has been widely redistributed along the east coast and hinterland of eastern Australia north of Sydney, New South Wales (Dhileepan, 2012). Both adults and nymphs feed on the cell content of leaves with their piercing and sucking mouthparts. Ultimately this feeding reduces the rate of photosynthesis, which leads to a reduction in stem and tuber growth. Although widely established, the field incidence of this agent is patchy and shows variable impact.

Identification

Adult leaf-feeding tingids are approximately 3 mm long, are creamy white when they emerge and turn grey as they age. Adults are identified by their delicate lace-like wings, with two raised dark marks on their elytra (forewings). The nymphs (juveniles) are smaller but also grey. The nymphs, unlike adults

(who feed singly on leaves), move and feed in groups on the underside of leaves. Damage by both adults and nymphs causes a speckling on the leaves, reducing the rate of plant photosynthesis (Dhileepan, 2012).

QDAF

Juvenile tingids.

QDAF

Adult tingids.

QDAF

QDAF

Skeletonising or speckling associated with tingid feeding, showing whole plant damage (top), close up of leaf damage (bottom).

Life cycle

The leaf-feeding tingid can develop and reproduce throughout the year, with each generation lasting around 38 days (Dhileepan, 2012). Adult females can lay an average of 187 eggs in their lifetime, which may be greater than two months under favourable conditions (e.g. temperatures between 20 and 30°C). Eggs are laid in groups of around 19 on the underside of leaves, along the central vein and partially embedded in the leaf tissue. Clutches of eggs are covered in a black coating, which is assumed to be for protection. Within two weeks, eggs hatch and nymphs begin to feed in groups on the underside of leaves while passing through five instars (growth stages) before becoming mature adults in three to four weeks.

Field collecting and rearing

Leaf-feeding tingids are widely distributed and both adults and nymphs can be collected on leaf undersides throughout the year. However, collection and redistribution is best undertaken in the cooler months, avoiding the hot summer period. Look for signs of chlorosis on leaves (speckling) to find the tingid. Ideally, at least 200 adults are required for your release site however less is acceptable, it may just take longer for damage to occur and repeated redistributions may be required for better establishment. Collect adult tingids by placing a vial or small container directly behind and below the tingid, and gently touch it so that it will fall backwards into the container. Nymphs can be collected throughout the day by carefully removing plant runners showing signs of chlorosis. Carefully remove tingid infested leaves or runners and place these in a cool dark container for transport to your release site. As only late instar nymphs are likely to transfer and develop, be sure to collect enough plant material that would be of similar magnitude to your adult release. While immediate release is recommended, adults and nymphs can be stored temporarily (at cool temperatures using an ice brick) in sealed containers containing some cat's claw creeper. Cover the container with a lid containing small air holes or insect mesh for ventilation (i.e. for a few days at around 20°C).

Alternatively, rearing is easy as the tingid has multiple generations per year. Humid conditions with temperatures in the mid-20°C range are best for rearing tingids. They can be reared in insect proof cages over an approximate four-week period, which varies depending upon temperature and humidity. As an example, a cage containing 12 potted plants (each plant with 10 to 12 cm long shoots) would require approximately 40 adults to produce 600 new individuals over a two month period. These can be used for field release, or for setting up new cages. To maintain a good supply of food for your tingids, host plants need to be prepared prior to setting up your rearing enclosure. Allow the host plants to grow enough foliage by providing plenty of water and apply treatments of liquid fertiliser if required. Once cat's claw creeper has developed at least 10 to 12 long shoots, the plants are ready to receive the adults and/or larvae. Regularly monitor your enclosures and keep them free from predators.

How and when to release

Release adult tingids (ideally approximately 200 per release site) directly onto healthy plants as soon as possible. For infected leaves and plant runners, intertwine cuttings among healthy cat's claw creeper vegetation. Repeated releases are recommended over a seven-month period, between September and March, to establish a balanced age cohort. As tingid dispersal is slow, averaging around six metres per year (Dhileepan *et al.*, 2010), varying release intervals in the landscape for redistribution should also be considered. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for tingid presence (adults and nymphs on underside of leaves) by identifying chlorosis (speckling) on mature leaves at the nursery site within one year of release and record presence or absence as per your monitoring guidelines (Appendix 3). As the tingid readily feeds in the lower canopy be sure to check the ground cover. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Cat's claw creeper jewel beetle
Hedwigiella jureceki

First released in 2012 in south-eastern Queensland (Snow and Dhileepan, 2013), the jewel beetle is still undergoing redistribution along the east coast and hinterland of eastern Australia north of Sydney. Both adults and larvae are very damaging from the ground level to low in the canopy. Larvae mine within the leaves, whereas adults feed predominantly on young leaves. Feeding damage, by larvae mining within the leaves and adults feeding on young leaves, can slow plant growth through limiting flowering and seed production. The beetle is widely established and continues to spread from release sites, but populations are low, seasonally variable and impact is still largely undetermined.

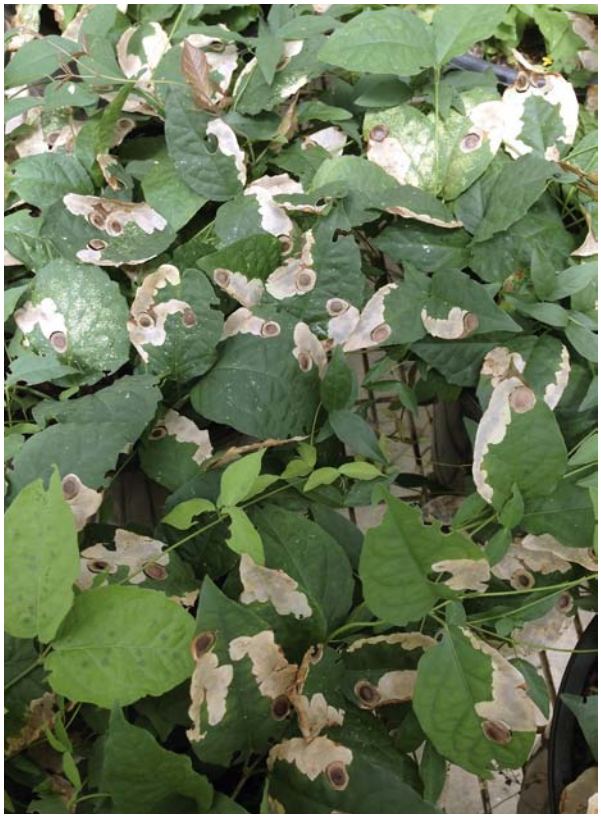
QDAF

Adult jewel beetles.

Identification

The adult jewel beetle is approximately 3 mm long and readily identified by its metallic black body with three lighter coloured wavy lines across its back. Larvae are yellow and appear to be almost triangular, with a wider head and a very narrow posterior end. Adults cause chewing foliar damage initially on leaf edges, preferring new leaflets, before working their way towards the interior of the leaf, whereas larvae tunnel within the leaf, forming characteristic blotch-like mines.

QDAF



NSW DPI

Blotch mines caused by larvae tunnelling and pupal discs.

Life cycle

Jewel beetles have several generations per year, with each lasting approximately two months. They are hardy and can survive sub-zero winter temperatures, but will not breed during this time. The adult female lays around 80 eggs, which are laid singly on the underside of leaf margins during her relatively long-lived lifespan of around five months (Dhileepan, 2012). Within 12 days, larvae emerge and tunnel directly from the egg into the leaf to feed. As they feed, they create a bladder or blotch mine effect visible on the leaf's surface. Larvae pupate after three growth stages which takes between 10 and 13 days. Larvae pupate by chewing out a distinct disc-like pupal case between the upper and lower leaf surfaces, which can remain in the leaf or can drop to the ground before adults emerge (from 11 to 24 days) (Snow and Dhileepan, 2014).

Field collecting and rearing

Jewel beetles are widely distributed and can be readily collected from September through to May. Look for adults basking in the sun on the upper side of leaves. As larval instars feed internally, transferring them for redistribution is not feasible. Ideally, approximately 400 adults are required for your release site. Collect adults by placing a vial or small container near the insect, gently touching the underside of the leaf and covering over the vial so that the beetle is trapped in the container. Jewel beetles can also be collected with aspirators (see Appendix 1 for relevant technique). Prior to redistribution, adults can be stored temporarily (at cool temperatures using an ice brick) in sealed containers containing cat's claw creeper. Cover the container either with a lid with small air holes or insect mesh for ventilation.

Recommendation

If you are rearing two or more biocontrol agents for your weed target, rear your agents in separate enclosures to try and prevent competition for food.

Rearing is easy as the jewel beetle has multiple generations per year. Humid conditions, with temperatures between 27 and 30°C, are optimal, but the insects will develop well in higher temperatures. Jewel beetles can be reared in insect-proof cages over an approximate eight to ten-week period. As a rule, about 10 beetles are required per potted plant (each plant with 10 to 12 cm long shoots), or to produce a decent-sized population for release about 80 adults across 30 potted plants in enclosures will suffice. Maintain a good supply of food and water. Remember to regularly monitor your enclosures and keep them free from predators.

How and when to release

Release adult jewel beetles (ideally 400 per release site) directly onto healthy plants as soon as possible. A single release should result in establishment, however, up to two additional releases may be required within the same year. To ensure establishment, releases are best made at sites with a northerly aspect in full sun between September and March. Avoid sites that are prone to flooding or frost. Dispersal is good, with research demonstrating an average dispersal rate of 100 m over a 15-month period, and site establishment from initial releases of greater than 70% (Snow and Dhileepan, 2014). Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for jewel beetle presence (adults, larvae and pupae) and feeding damage (blotch mines, disc-like pupal cases and leaf margin chewing) in the autumn and spring within one year of release. Record its presence or absence as per your monitoring guidelines (Appendix 3). Adults can generally be

found on the lower trunks of vines in sunlit areas. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Cat's claw creeper leaf tying moth
Hypocosmia pyrochroma

First released in 2008 in south-eastern Queensland, (Dhileepan, 2012) the leaf tying moth has established; albeit at a restricted number of sites (L. Snow pers. comm., 2019). Larvae feed destructively on the leaves which leads to premature leaf drop and a reduction in the tuber reserves and subsequent plant growth. Larvae protect themselves by tying leaves together with silk to create tunnels in which they move about and feed. Though establishment is still restricted, increasing populations and damage levels are now observed but overall impact is still being determined.

Identification

Adult leaf-tying moths are pinkish-orange to brownish-orange, up to 15 mm long with distinctive banding across their wings. They have a prominent white band across the centre of their wings and a dark, v-shaped band across the posterior end of their wings. The young, light-grey coloured larvae transition to dark brownish-grey as they develop measuring up to 2 cm long at maturity. Adults are non-feeding and nocturnal. Damage typically inflicted is by larvae, causing skeletonisation and leaf abscission in their silken tunnels.

QDAF

QDAF

Leaf tying moth (top) and larvae (bottom).

Life cycle

The leaf tying moths can have several generations per year, each lasting around 72 days. Adult females lay up to 120 eggs over their short lifespan of 10 days, which are laid singly on the underside of leaves and stems. Within two to three weeks, larvae emerge and conceal themselves by tying leaves together to create a network of silken tunnels to feed destructively and develop through six growth stages. Larvae pupate within these tunnels and in the soil. Pupae undergo diapause from April/May to September/October, with most adults emerging after four weeks from early summer (Dhileepan *et al*., 2013).

QDAF

Extensive damage caused by leaf tying moth.

QDAF

Skeletonisation by the leaf tying moth.

Field collecting and rearing

The leaf-tying moth is currently not available for redistribution due to limited establishment at release sites in south-eastern Queensland. Additionally, the leaf-tying moth requires specialised rearing techniques because it undergoes winter diapause, so rearing is not recommended. Establishment has occurred in Queensland at a few sites and, with time, may spread naturally.

How and when to release

The agent is not available for release.

Monitoring for natural dispersal

Look for presence of the leaf-tying moth by initially looking for strings of leaves tied together by silken threads, then look for the hidden larvae inside their protective covering. As larvae wriggle energetically and may fall when disturbed, place a tray under the string of tied leaves before looking. If present, report your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually.

Crofton weed

Ageratina adenophora

Crofton weed is a multi-stemmed, perennial shrub native to Mexico (Julien and Griffiths, 1998). It typically thrives in coastal and subcoastal high rainfall regions of New South Wales through to south-east Queensland (McFadyen, 2012b). Unpalatable to cattle and poisonous to horses, Crofton weed causes acute respiratory disease in animals that can be fatal (O'Sullivan, 1979). Plants have upright, purplish-branched stems (1–2 m tall) that form from a shallow rootstock (McFadyen, 2012b). The stems are sticky and hairy when young and bear pairs of oppositely arranged, trowel-shaped leaves (up to 15 cm long) with toothed margins. In spring, plants produce masses of small white flowers (5 to 6 mm in diameter) in dense clusters at branch tips. Each plant can produce between 10,000 and 100,000 seeds per year, and can also spread vegetatively from its rootstock (Morin, 2013). Thousands of tiny windborne seeds have the ability to colonise disturbed patches over long distances. It is possible that Crofton weed releases chemicals into surrounding soils which may prevent native seedling germination and contribute to its spread (Zheng and Feng, 2005).

F. & K. Starr

Crofton weed flowers and foliage.

Introduced as an ornamental plant in the late 1800s and first recorded as naturalised near Sydney in 1904, Crofton weed quickly spread north into Queensland (McFadyen, 2012b; Parsons and Cuthbertson, 2001). Recognised in the 1930s as a serious weed of pastures and crops along Australia's east coast, Crofton weed, once germinated, can readily tolerate shade and quickly form dense infestations. Widespread in eastern Australia, it is most prevalent along waterways and on cleared, non-grazed land in the Sydney to Wollongong region, the New South Wales North Coast and up into south-east Queensland.

Australia introduced an insect and a pathogen (rust fungus) to test their potential as biocontrol agents for Crofton weed. In 1952, a gall fly (*Procecidochares utilis*) (McFadyen, 2012b) and in 2014 a rust fungus (*Baeodromus eupatorii*) (Morin, 2013) were released, with both establishing in the field. In 1954 a previously misidentified leaf-spot fungus (*Passalora ageratinae*), likely introduced on the bodies of adult gall flies, was recorded in Queensland on Crofton weed (Dodd, 1961). It is now widespread across the invaded range of Crofton weed in Australia (McFadyen, 2012b).

The combined effect of the Crofton weed gall fly and leaf-spot fungus has had a significant impact on Crofton weed populations in Australia (McFadyen, 2012b). Although Crofton weed still remains a problem in some areas, the subsequent introduction of the Crofton weed rust fungus has resulted in successful establishment on Crofton weed and greater defoliation. In combination, the fly and the pathogens have a complementary impact on Crofton weed; the gall fly attacks the stems, while young leaves are first infected by the rust fungus and later colonised by the leaf spot fungus which overall results in greater plant defoliation.

Crofton weed rust fungus
Baeodromus eupatorii

First released in 2014, the Crofton weed rust has been widely redistributed in partnership with land managers and the community in eastern New South Wales and more recently in south-east Queensland and on Lord Howe Island (Morin, 2015). Infecting young leaves and stems, the rust absorbs nutrients and water from the plant, thereby limiting its resources available for reproduction. The fungus also destroys leaf tissue by producing fruiting bodies (spores), which reduce the photosynthetic capacity of the plant leading to cell death. Now widespread, the rust has great potential through regular defoliation over time to reduce the competitiveness, reproduction, and spread of the weed.

Identification

Symptoms of the rust are best seen during autumn and winter, coinciding with higher rainfall and active plant growth. The rust first appears on young leaves and stems but can be seen on older plants as they grow. Small orange pustules (up to 0.3 mm in diameter) occur predominately on the upper surface of leaves in circular groups but can also be seen on leaf stalks (petioles) and stems, which can lead to swelling, contortion and sometimes cell death.

Life cycle

The rust completes its entire life cycle on Crofton weed in three to four weeks and is comprised of two commonly seen spore stages, the pycnia (also called spermogonia) and telia which produce wind-dispersed basidiospores. Pycnia are flask-shaped, orange-yellow and mostly produced on the upper surface of leaves. Alternatively, production of telia occurs on the underside of leaves but sometimes both can appear on leaf stalks and stems causing swelling and distortion.

L. Morin

L. Morin

Crofton weed rust fungus on the upper (top) and lower surface of leaves (bottom).

Around autumn and winter, and corresponding with active plant growth and rainfall, microscopic basidiospores will readily germinate on the young stems and leaves of Crofton weed. During this time, basidiospores infect the plant by penetrating the plant's epidermal cells and can also be wind dispersed to infect neighbouring Crofton weed plants. Dispersal of up to 15 km in one year has been recorded (Morin, 2015).

Within two to three weeks, signs of infection appear in the form of pycnia (golden orange pustules that have visible mucus, on the upper surface of leaves and sometimes on leaf stalks and stems). Cross fertilisation between pycnia is then required for telia to develop. This occurs through sweet mucus attraction, enabling transport of pycniospores by insects. Within a few days, and after cross fertilisation, telia develop on underside of leaves and on petioles and stems, after which, they are immediately capable of germination to produce basidiospores to start the infection cycle again. The rust thrives in shady sites with mild temperatures (18 to 25°C) and high rainfall and humidity (Morin, 2015).

Field collecting and redistribution

CSIRO has developed a step-by-step pictorial guide for three methods of redistributing Crofton weed rust fungus including: 1. the potted plant method, 2. layering method and 3. transplanting method (https://research.csiro.au/crofton-weed/wp-content/uploads/sites/68/2020/08/Guidelines-redistribution-Crofton-weed-rust-fungus.pdf). It is not recommended to transfer rust-infected leaves from one site to another because, when infected material is removed from a plant, the rust dies very quickly and will seldom produce the necessary spores for a new infection to occur.

The potted plant method (Method 1) or the layering method (Method 2) are the preferred redistribution techniques because they do not require the movement of soil between sites, thus reducing the risk of spreading soil pathogens and weed seeds. It is best to redistribute the rust fungus in autumn and winter at shady sites when environmental conditions are cool (18 to 25°C) and moist. Hot summers should be avoided.

Practise hygiene

Do not contribute to spreading other weeds and pathogens by transferring soil between field sites.

Method 1. Potted plant method (CSIRO Australia)

1. Propagate three to four Crofton weed plants by cutting new woody stems (around 30 cm long) and place individually in pots containing potting mix (not soil from the field). Retain three to four young leaves and remove the rest. Place pots in partial shade and water regularly until healthy new shoots develop.
2. Place several potted healthy plants in a saucer underneath infected plants at a Crofton weed rust-infested site. Pour water into the saucer and water regularly, particularly in the absence of rain. Mark your site (e.g. with a stake).
3. Within four weeks, rust infections will develop on the leaves of potted plants.
4. Move newly infected potted plants to your new Crofton weed infested nursery site and place these under weeds. Add water to the saucer, mark your site and water plants regularly particularly if there is no rain.

Method 2. Layering method (CSIRO Australia)

1. Make two holes opposite each other and approximately two inches below the rim of three to four pots. Slide a piece of rigid wire through the holes and fill the pots three quarters full of potting mix (not soil from the field).
2. At a Crofton weed rust-infested site, select two to three long stems of Crofton weed plants growing in the shade, that have rust on the leaves.
3. Gently bend stems to fit under the wire within the pot and fill the pot with the rest of the potting mix to cover the stems.
4. Place a deep saucer under the pot, water and mark your area (e.g. with a stake). Water regularly particularly if there is no rain.
5. Within four weeks, rust infections will develop on the leaves of these potted, layered stems. By this stage, the stems buried in the pots would have developed roots and the potted infected plants will be able to survive on their own. Cut the stems linked to the pot from the site.
6. Remove newly infected potted plants to your new Crofton weed infested nursery site and place these under infestations. Add water to the saucer, mark your site and water regularly particularly if there is no rain.

Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Follow-up monitoring of the release sites should take place a month after inoculation to check for signs of infection. Look for signs of rust, indicated by orange pustules on the leaves. If rust is present, begin monitoring for dispersal at incremental distances away from each nursery site as per your monitoring guidelines (Appendix 3). Monitor annually. If there is no sign of the rust within two months of inoculation, consider inoculating the plants again if time permits before the onset of summer.

Crofton weed gall fly
Procecidochares utilis

First released in Queensland in 1952 and later redistributed in New South Wales the Crofton weed gall fly, which is native to Mexico, was introduced from an existing biocontrol program in Hawaii (McFadyen, 2012b). Early indications were positive with immediate establishment and rapid dispersal of the fly, resulting in good impact on the weed (stem death and greatly reduced plant growth). Within a few years however, the gall fly's impact on Crofton weed reduced, likely due to native parasitism (Dodd, 1961). Due to the fact that the gall fly is widely distributed and its impact is relatively limited, redistribution is unnecessary.

Identification

Adults are 3 to 4 mm long, with striking black and grey patterns across their wings. Characteristic of the tephritid family, adults move in an agitated manner with jerky leg movements and wing flexure. Larvae are a semi-transparent white colour and live within galls. They are elongate and become larger and flatter as they move through their growth stages (instars). Newly laid eggs are translucent white and are elongate and elliptical in shape (up to 0.6 mm).

T. Murray

Crofton weed gall fly.

Life cycle

The Crofton weed gall fly has multiple generations per year with each taking a little over two months. Adult females lay up to 70 eggs in stem tips between the paired bud leaves during their two-week lifespan over spring and summer. Within a week, eggs hatch and larvae immediately tunnel into the stem and begin feeding on plant tissue. Larval feeding within the stem induces gall formation by the plant. Each gall may contain two to four larvae (McFadyen, 2012b). Over a period of around 40 days, the larvae develop through several growth stages (instars) before pupating within the galls. Pupation lasts around 20 days before adults emerge through clear epidermal 'window panes' on the side of galls.

R. Holtkamp

Gall formation.

Redistribution

While the gall fly remains widespread throughout the distribution of Crofton weed, parasitism levels remain high and sufficient gall numbers are rarely achieved to have a significant impact. Stem die back does not occur and flowering is not prevented by this agent. Redistribution is not recommended.

Monitoring

The easiest way to monitor for the gall fly is to look for galls on plant stems. Adult flies are also active during the day and may be observed on the plants. If present, record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Crofton weed leaf spot fungus
Passalora ageratinae

First recorded in Queensland in 1954, the Crofton weed leaf spot fungus was likely introduced into Australia through spores carried on the bodies of the original gall flies imported from Hawaii (Dodd, 1961). It is now widespread on Crofton weed in southern Australia. The leaf spot fungus forms small, brown spot lesions on the older leaves. This is followed by leaf yellowing and premature leaf-fall, resulting in stems becoming leafless, except for the first three to four youngest leaves. Although infections are common and widespread, there appears to be minimal impact on mature plants other than early leaf drop and an inhibition of side-shoot development. In favourable wet conditions however, the fungus can rapidly kill seedlings.

Identification

Symptoms of the leaf spot fungus are predominately on the lowest leaves of stems when the plant is actively growing. The fungus appears on older leaves as small, angular brown spot lesions with raised

dark edges (2 to 8 mm in diameter). As the fungus progresses, lesions coalesce, and with sufficient infection leaves will senesce and drop from the plant. Under moist conditions, you may be able to see the tiny felt-like, grey to brown, spore producing structures emerging from the lesions.

Life cycle

The leaf spot fungus completes its entire life cycle on Crofton weed. Around autumn and winter, and corresponding with active plant growth and rainfall, spores readily germinate on the surface of leaves from groups of spore producing structures that emerge from dead tissue. Under ideal temperatures (20 to 25°C) and suitable humidity, spores germinate. Spores grow on the leaves for up to five days and during this time the fungus infects the plant by entering the leaves through stomata (breathing pores). Within two to three weeks, signs of infection appear in the form of light coloured spots that turn brown by five weeks.

Redistribution

This leaf spot fungus is widespread, and little is to be gained by redistributing it. Impacts are driven by environmental conditions. Where plants are not infected, conditions are likely suboptimal and too dry for the fungus to thrive.

L. Morin

Lesions of the leaf spot fungus.

Monitoring

Look for leaf pustules on the lowest leaves of stems in autumn and winter. If present at your site, record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Docks

Rumex species

Many of the docks, genus *Rumex*, are serious weeds of pasture globally. In Australia, of concern are *Rumex crispus*, *Rumex conglomeratus*, *Rumex obtusifolius* and *Rumex pulcher*, which are four species of erect, perennial herbs native to Eurasia that thrive in high rainfall agricultural areas of Australia. Docks are distinguished by their long large taproots (up to 3 m in depth) and fleshy to leathery leaves that form a basal rosette with erect branching flowering stalks (up to 150 cm). Leaves are hairless, often with short stalks and may be narrow or broad, depending on the species. Flowers are generally green at first but may become reddish-brown as the fruits mature. Seeds are long-lived and prolific (around 60,000 per plant) (Parsons and Cuthbertson, 2001).

Docks were recorded growing in the Melbourne Botanical Gardens in the 1850s and by the 1950s were recognised as serious pasture weeds (Whittet, 1958). By the 1970s primary producers indicated that docks were seriously reducing pastoral productivity, particularly in south-western Western Australia where dock rapidly expanded its range to about 100,000 ha (Allen, 1975). A long-lived seed bank and

R.G. Richardson

Dock rosette.

S. Ivory, SARDI

Flowerin dock plant.

R.G. Richardson

Dock fruits.

P. Sullivan

Infestation of dock in New South Wales pasture.

large tap roots enable docks to survive long, dry summers before rapidly regenerating to aggressively outcompete beneficial pastures following autumn rain. Largely unpalatable to stock (especially to cattle) accumulated oxalates in docks may lead to poisoning and death.

Australia introduced nine species of insects to test their potential as biocontrol agents for docks (Strickland *et al.*, 2012), with only the dock moth (*Pyropteron doryliformis*) establishing in the field. Three other insect species have been found in Australia on docks that were the result of unauthorised introductions (Strickland *et al.*, 2012).

Dock moth
Pyropteron doryliformis

First released in 1989 in Western Australia, the dock moth, which is native to Mexico, was imported from an existing biocontrol program against dock in France (Strickland *et al.*, 2012). Impressively, releases at more than 700 sites were made over the following 20 years in Western Australia, New South Wales, Victoria, South Australia and Tasmania, resulting in establishment at approximately 70% of sites (Strickland *et al.*, 2012). Larvae feed extensively inside the taproot during the summer months when the host plant is effectively dormant, preventing normal plant regeneration in autumn. With only one generation per year, it can take ten or more years for populations to build up and disperse. Despite this, impact is substantial with some sites being reduced by up to 100%, some five to six years after the dock moth was released (Strickland *et al.*, 2012).

Identification

Adult female moths have a body length of approximately 15 mm and a wingspan of 12 to 14 mm. Male moths have a slightly smaller body length of approximately 12 mm and a fan-like tuft of scales at the end of their abdomens. Moths have a brownish-black body and distinctive bands across their abdomen that are white and either brownish-orange for females or yellowish-orange for males. Wings of adult dock moths have clear sections ('windows') and dark-brown edges. Larvae can reach 25 mm long and are creamy-white with an orange-coloured head. Larval feeding occurs internally in the rootstock and lower stem. Attacked plants have holes bored into the lower stem or upper tap root or stem, which indicate where adult moths have exited the plant. The growth of these plants will be stunted, and the aerial plants parts will die off prematurely.

Recommendation

Widely distributed, the dock moth is providing substantial control of docks. It can be redistributed to enhance dispersal, if there are no signs of dock moth populations at your site. Be warned, the current best practice for its redistribution is laborious and time consuming.

Agriculture WA

Male dock moth.

Tasmania Institute of Agriculture

Female dock moth.

J. Heap, PIRSA

Dock moth larva.

Life cycle

Dock moth has one generation per year, most of which is spent as larvae in the roots of dock plants. Adult moths are day-active and feed on the nectar of flowering plants (Strickland *et al.*, 2012). Females lay eggs on mature, flowering stems in spring and summer. Larvae tunnel into the rootstock to feed over summer and autumn. Several larvae may occupy a single, large taproot. Mature larvae undergo diapause during winter. As spring/summer approaches larvae become active and, in preparation for pupation, mature larvae create holes from within the lower stem or upper tap root. Once pupation is complete, the adult moth emerges.

Field collecting, rearing and redistribution

Rearing and redistribution of the dock moth is extremely difficult and requires considerable entomological expertise, equipment and a lot of time. The dock moth is widely established through the invasive range of docks in Australia, however, should dock populations be located where no sign of the moth can be detected, it may be possible to field collect rootstock containing pupae in winter from well-established sites for redistribution. Current best practice also involves regular site visits to provide food for the moths in addition to releasing females from your enclosure. See Appendix 1 ('field cage technique') for details on rearing and redistributing your moths. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

If rearing and redistributing the dock moth, you will need to differentiate between female and male moths. Males' abdomens end with a fan-shaped tuft. See photos above to differentiate between male and females.

Monitoring establishment and dispersal

Wait two to three years after an initial release before monitoring, as moth populations will be low and destructive sampling may impair the viability of the release site. It is best to destructively sample, i.e. dig up and examine the stem and rootstock, during winter for the presence of larvae (see Appendix 1). Examine stunted plants that have prematurely died off with holes bored into their lower stems. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

Other insects and pathogens that have established on docks in Australia

- **Dock aphid**, ***Brachycaudus rumexicolens***, was first recorded in Western Australia in 1985 and is thought to have been an unauthorised introduction from either Eurasia, Africa or North America (Yeoh *et al.*, 2012). It is widespread on docks and many other species from the Polygonaceae family in dryland agricultural areas of Western Australia. Its impact on dock species is unknown. Risk assessments to see if this agent is safe to use as a biocontrol agent have not been carried out so it should NOT be redistributed.

- The **rust fungus**, ***Uromyces rumicis***, was first recorded in south-western Western Australia during surveys conducted from 1990 to 1992. It is thought to have been an unauthorised introduction from Europe (Scott and Shivas, 1993). The rust fungus, which had previously been considered as a potential biocontrol agent as part of the overall program, was found attacking *R. pulcher* at sites where other *Rumex* and *Emex* species were present. Risk assessments to see if this agent is safe to use as a biocontrol agent have not been carried out so it should NOT be redistributed.

- **Dock sawfly**, ***Ametastegia glabratea***, was first found in Victoria in 1993 and is thought to have been an unauthorised introduction from Europe (Malipatil *et al.*, 1995). Sawfly larvae feed on dock leaves, however, its pupae have been recorded from a range of plants. It is considered a pest and should NOT be redistributed.

Gorse

Ulex europaeus

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Gorse flowers and foliage.

Gorse is an erect, branched, long-lived (up to 30 years) spiny shrub native to Europe (Parsons and Cuthbertson, 2001). These thicket-forming, woody shrubs can reach 7 m in height but are more commonly between 1 and 2.5 m. Plants have dark green, spine-like leaves (up to 30 mm long) and green stems armed with numerous spines (up to 50 mm long) that turn pale brown as they age. The pea-like, bright yellow flowers are present throughout the year and seed pods are densely hairy (up to 20 mm long). While the roots are deep and extensive, any stems running level with the plant base can produce roots. A mature plant can produce up to 18,000 seeds annually (Broadfield and McHenry, 2019).

Introduced as an ornamental plant particularly for hedging during the early 1800s (Parsons and Cuthbertson, 2001), gorse soon became problematic in agricultural and urban environments of temperate regions. From the 1930s gorse became recognised as a serious weed due to its negative impacts on agriculture, forestry, riparian and native environments and in 1999, gorse was listed as a Weed of National Significance due to its environmental and economic impacts, invasiveness and potential to spread.

Australia introduced three insects and one mite, from existing biocontrol programs against gorse in New Zealand, for further testing. All agents were released, with the seed weevil (*Exapion ulicis*), thrips (*Sericothrips staphylinus*), soft shoot moth (*Agonopterix umbellana*) and mite (*Tetranychus lintearius*) establishing (Ireson and Davies, 2012).

Recommendation

Treat isolated plants using an appropriate management technique for the site (e.g. herbicides, mechanical clearing, cultivation and or grazing) and then identify areas in the core invasion for good establishment of biocontrol agents. Speak to your local weed or biosecurity officer for advice.

P. Sullivan

Gorse infestation in forestry.

Gorse seed weevil
Exapion ulicis

First released in 1939 the gorse seed weevil is now widespread in Tasmania, Victoria, South Australia and New South Wales (Ireson and Davies, 2012). While larvae are capable of slowing gorse reproduction (by destroying between 12 and 55% of seed) (Davies *et al.*, 2008) this damage is not high enough to reduce overall plant density (Ireson and Davies, 2012). As a result, other agents are required to complement the seed weevil's activity.

Identification

Adult weevils are 2 to 3 mm long, light grey, with a long snout (rostrum) about half as long as its body. The adults feed by digging into the stems and spines of gorse, creating characteristic round holes. The white larvae grow to about 2.5 mm long within the seed pods. Larvae are more effective at reducing seed density than adults.

W. Chatterton

Gorse seed weevil.

Life cycle

Seed weevils are present throughout the year and can live for up to 12 months. Adults only breed once per year, with females laying an average of nine eggs in developing seed pods during spring and summer (Davies *et al.*, 2008). After hatching, larvae feed on developing seeds for up to eight weeks. Pupation takes approximately two months before emerging as adults when dry pods burst open. Adults will die if pods fail to open. Newly emerged adults suspend their development over winter and breed the following spring.

Field collecting and rearing

Rearing is unnecessary. Adult weevils are best collected and redistributed in spring to allow for population increase over summer prior to winter hibernation. Collect at least 100 weevils for each planned release site by beating the plant's foliage (see Appendix 1 for technique). Prior to redistribution, weevils can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

How and when to release

Release collected weevils (at least 100 adults per site) directly onto healthy plants near one another (to aid establishment) as soon as possible. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Use the beating method (see Appendix 1 for technique) to monitor for the presence of adult weevils within one year of release. Monitoring can occur at any time throughout the year but should be repeated annually. Begin monitoring for dispersal at incremental distances away from each nursery site as per the monitoring guidelines (Appendix 3).

Gorse spider mite
Tetranychus lintearius

First released in 1998, the gorse spider mite is now widespread throughout the invaded range of gorse in Australia (Ireson and Davies, 2012). Extensive feeding from piercing and sucking mouthparts can kill shoots, reduce plant growth and overall plant biomass. Although the mite is having a good impact, predators (e.g. the introduced Chilean predatory mite *Phytoseiulus persimilis* and the native ladybird *Stethorus* sp.) can decimate populations, reducing the overall impact of the mite (Ireson *et al.*, 2003; Davies *et al.*, 2009). As a result, other agents help to complement the mite's activity.

Identification

Adult mites are smaller than a pin head (about 0.5 mm long) and reddish-orange with dark-grey patches. Juveniles look similar to adults but are smaller and a brighter orange. Males are smaller than females and triangular. Despite their small size, mites live in colonies and are identifiable all year round by their white webs. During colder weather, mites are more inclined to cluster at the centre of the web for protection from the elements. Their feeding damage further identifies their presence, as their sucking and piercing mouthparts cause the foliage to appear bleached (or even brown).

P. Sullivan

Tell-tale webbing of gorse spider mite and bleaching.

W. Chatterton

Gorse spider mites.

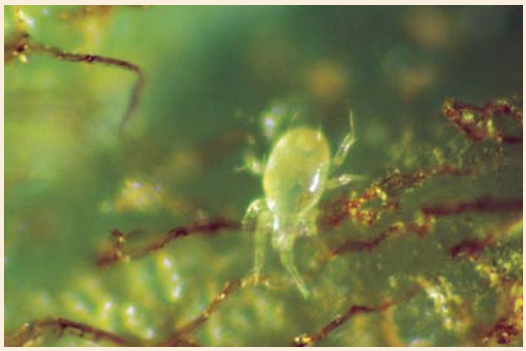
Life cycle

Mites have several generations per year. In less than six weeks, depending upon temperature, their life cycle is complete. Each female lays a few eggs each day for around 30 to 40 days. Eggs hatch within two weeks and there are several juvenile stages before they develop into adults (Gerson *et al*. 2003).

Field collecting and rearing

Rearing is unnecessary. Look for webbing and bleached foliage to locate mites at any time of the year. Mites are easier to find in the warmer months when colonies are at their largest. Also, look for mite activity as predators can decimate colonies. Select small cuttings (10 to 20 cm long) with mites that have massed together. Preferably collect small cuttings from several mite infested gorse plants rather than large cuttings to help minimise the transfer of predatory mites that are prevalent during spring and summer. Prior to redistribution, mites can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

Watch out for two predators of the gorse spider mite. The introduced Chilean mite (*Phytoseiulus perisimilis*) and a species of native lady beetle (*Stethorus* sp.) are widespread among spider mite populations and have the potential to reduce the spider mite's potential as a biocontrol agent.



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Phytoseiulus mite.

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Stethorus lady beetle eating a mite.

How and when to release

Attach several mite-infested cuttings to each healthy gorse shrub with tie wire. Each mite infested cutting should contain several hundred individuals, which should be ample to ensure establishment. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for mite activity within one year of release and preferably in the warmer months when mites are more active. Look for mite activity in the webbing and/or signs of their past presence (indicated by bleached foliage). Record their presence or absence as per the monitoring guidelines (Appendix 3). Monitor annually and record their dispersal at incremental distances away from each nursery release site.

Gorse thrips
Sericothrips staphylinus

Population densities of gorse thrips, since their release in 2001 throughout temperate Australia, have established but remain low. Dispersal is slow and there is limited evidence of their impact (Ireson and Davies, 2012). However, a glasshouse study indicated the potential for the gorse thrips to have a significant impact on the growth and survival of gorse if populations in the field are able to increase to high enough densities over time (Davies *et al.*, 2005).

Identification

Adult gorse thrips are tiny (approximately 1 mm long) and black, except for the white undeveloped wing buds. The juveniles are yellow and look similar to the adults. White spotting along gorse stems and spines, and an overall mottled, blotchy-like appearance of the plant, are signs of damage typically inflicted by juvenile and adult thrips piercing and sucking on plant cell contents.

Life cycle

Gorse thrips have two generations per year, one in late winter to early spring and a second in late spring to early summer. At 20°C their life cycle takes approximately 32 days (Ireson *et al.*, 2008). Eggs are undetectable, about 0.3 mm long and hidden within

W. Chatterton

Gorse thrips.

gorse stems. Female thrips lay their eggs within slits cut into young stems. Eggs hatch in the warmer months to coincide with succulent new plant growth for developing juvenile thrips. When stems harden towards the end of summer, the adult population suspends its development until late winter when adults resume egg laying for the following season.

Field collection and redistribution

Gorse thrips are difficult to collect due to their small size and they are easily confused with other thrips species. To improve the chances of collecting good numbers of gorse thrips, make collections in the warmer months of the year, especially focussing on the new plant growth. Avoid collecting thrips when the gorse is flowering as other native flower thrips may be present. Most species of gorse thrips do not have wings and are therefore poor dispersers. Collecting good numbers of gorse thrips from established sites, and then releasing them at uninfected sites can help overcome the slow dispersal of this species. Redistribute the gorse thrips between October and March, to enable recovery of populations before winter.

Gorse soft shoot moth
Agonopterix umbellana

The long-term impacts of the gorse soft shoot moth, since its first release in Tasmania and Victoria in 2007 (Ireson and Davies, 2012) and later releases in South Australia, Victoria and New South Wales between 2016 and 2017, have not been determined. Larval feeding on new spring growth may cause severe foliar damage, restrict growth and possibly limit flower and seed production.

Initially the gorse plants appear to compensate for the severe foliar damage by initiating new shoots, but the long-term effects of this agent on gorse has not been determined. Redistribution programs are encouraged.

Identification

You are unlikely to see the adult gorse soft shoot moths as they are nocturnal and hide deep within gorse during the day. However, they are light tan with striking dark tan lines over the forewings. They are approximately 10 mm long with a wingspan of up to 21 mm. The olive-green coloured larvae (up to 35 mm long) live inside a silken tube that they spin near the growing shoot tips. Webbed or deformed shoot tips during the warmer months are signs of caterpillar activity.

P. Sullivan

Gorse soft shoot moth.

Life cycle

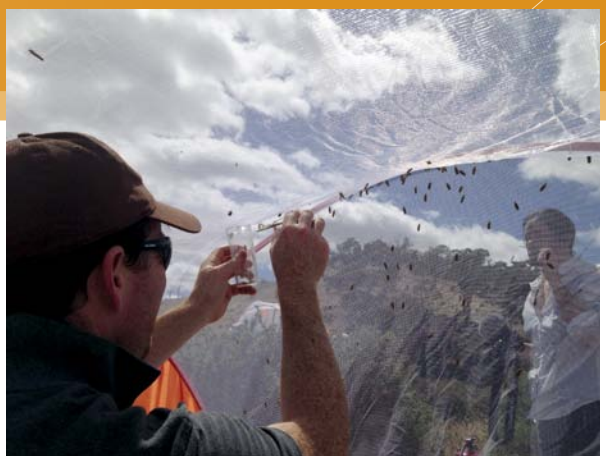
The gorse soft shoot moth has only one generation per year and its life cycle takes around eight weeks, or up to 32 weeks in cooler conditions. Adults lay eggs near buds at the base of spines, or on stems, from late winter until late spring. Larvae hatch in mid-spring and feed on succulent new growth from developing spines near the shoot tips. As larvae develop, they spin large silken tubes before pupating within these structures from mid-summer. Adults emerge during summer and suspend their development over winter (Ireson *et al.*, 2013).

W. Chatterton

Gorse soft shoot moth final instar larva.

A. McConnachie

Damage from the gorse soft shoot moth.



A. McConnachie

Collection of gorse soft shoot moths in a tent.

Field collecting and rearing

Rearing is unnecessary. To collect large numbers of adult moths (at least 250 adults are required per release site) use a smoker tent (see Appendix 1 for technique) in early February to flush them out. Alternatively, in mid-December collect larvae (greater than 500) approaching maturity from their webbed shelters at branch tips. To achieve this, cut branches using a pair of secateurs and place material in an esky (with an ice brick with a barrier from the insects, e.g. newspaper) for transport to your new release site.

How and when to release

Release collected moths (at least 250 adults per site) or larvae (500 to 1000 per site) directly onto healthy plants near one another as soon as possible. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

As adult moths are not easily seen, look for the olive-green larvae hiding inside the cream silken tubes near the end of growing shoot tips within one year of release and record their presence or absence as per your monitoring guidelines (Appendix 3). Monitoring for larval presence from late spring to early summer is best, when larvae are actively feeding on new growth. Begin monitoring their dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Horehound

Marrubium vulgare

Horehound is an erect, bushy perennial herb native to Europe, Asia and North Africa (Parsons and Cuthbertson, 2001). It thrives on poor soil and readily invades overgrazed farmland, and disturbed conservation and bushland areas of southern Australia, where annual rainfall exceeds 200 mm. Horehound contains a bitter alkaloid, which makes it unpalatable for grazing livestock. Plants have opposite pairs of silvery olive near circular leaves (4 cm long). Covered in white cottony hairs, leaves are deeply veined, appear crinkled, and have bluntly toothed margins. Hairy stems are four sided and woody at the base. Highly aromatic, this much-branched weed (up to 75 cm) produces clusters of small white flowers (6 to 10 mm long) in summer. The fruit are brown burrs with small hooked spines (up to 2 mm) that readily attach to and disperse with stock, clothing, machinery, and vehicles. Mature plants can produce in excess of 20,000 seeds per year and seeds can remain viable within the soil for seven to 10 years (Blood, 2001).

Horehound was likely introduced to Australia in a shipment of botanical plants in 1798 (Frost 1993) and later promoted as a garden ornamental plant and medicinal herb. By 1848, horehound was naturalised in South Australia with its heaviest infestations recorded in the south-east of the state. Horehound's distribution extends further into north-western Victoria, New South Wales, Tasmania and sporadically in parts of south-east Queensland and Western Australia. It is also present in Victoria, Western Australia and south-eastern South Australia in semi-arid environments where it readily outcompetes native plant species.

Australia introduced four species of insects to test their potential as biocontrol agents for horehound. Two of these, the horehound plume moth (*Wheeleria spilodactylus*) and horehound clearwing moth (*Chamaesphecia mysiniformis*), were released and have established in the field (Weiss and Sagliocco, 2012).

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Horehound foliage (top) and flowers (bottom).

Recommendation

The combined effect of horehound plume moth and horehound clearwing moth is expected to reduce the spread and vigour of horehound in Australia. Effective control of horehound is best achieved using conventional control methods combined with biocontrol and with the presence of both moth species at your site.

Horehound plume moth
Wheeleria spilodactylus

First released in 1994 in Victoria and later redistributed in New South Wales, South Australia and Tasmania, the horehound plume moth, from southern France and Spain, is now widely established across the invaded range of horehound in Australia. At high infestation densities horehound plume moth larvae have been recorded to reduce plant biomass and lifespan (Weiss and Sagliocco, 2012).

Identification

Adult horehound plume moths are white (up to 10 mm long) and characterised by their resting posture. When at rest, their outstretched wings span between 20 and 25 mm and form a T-shape. Larvae are very hairy and similar in colour to the silvery green leaves of horehound. Pupae are silver green and transition to brownish-green with maturity. Damage is characterised by larvae feeding on leaves, initially along the leaf margins before skeletonising or consuming whole leaves.

Life cycle

Horehound plume moths can have up to four generations per year, with the summer generation lasting approximately 48 days. Adult females lay around 100 eggs singly or in groups of up to four on leaf undersides over a two-week period. After a few days to one week, eggs hatch and larvae begin feeding on the leaves, soft stems and shoot tips, before working their way down to feed on more mature foliage. Within a few weeks, they pupate within a silky cocoon on the upper leaf surface. Larvae from the autumn generation overwinter in leaf buds before pupating in spring.

Field collecting and rearing

The horehound plume moth is widely established and does not require redistribution. However, if the plume moth is not present at your site you can accelerate dispersal by redistributing these from a well-established site in spring and early summer when larvae and pupae are present on leaves. Rearing is unnecessary as larvae and pupae of the moth can easily be collected for redistribution. Initially look for leaves that have had their edges eaten or that have been skeletonised or mostly consumed. Thereafter, examine the tops of leaves more closely for well camouflaged larvae and pupae. Collect at least 200 cuttings, 10 to 20 cm in length, containing larvae and pupae and store them temporarily in a cool insulated box with ventilation to prevent foliage from drying out or insects over-heating (i.e. cool temperatures of around 15°C). Do not refrigerate larvae or pupae.

P. Sullivan

Horehound plume moth at rest.

S. Ivory, SARDI

Horehound plume moth larva.

How and when to release

As adults preferably lay on flowering horehound, release larvae and pupae directly onto actively growing and flowering horehound in the warmer months. To improve establishment, release larvae and pupae in batches of ideally 200 in high rainfall regions (i.e. rainfall of greater than 500 mm per year). Loosely place several of the larvae infested cuttings onto each plant at the nursery site near one another. Avoid releasing plant material that may contain seeds. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Within a few weeks of release, look for first instar larvae which are often found in the growing tips of the plant. Open the tips up and look for dark spots of insect excrement. Closer to a year after the first releases of the plume moth, confirm its presence by looking for larval damage through damaged leaf margins and leaf skeletonisation. Additionally, look for the presence of caterpillars, pupae and adults. Record presence or absence as per your monitoring guidelines (Appendix 3) and begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Horehound clearwing moth
Chamaesphecia mysiniformis

First released in 1997 in South Australia and Victoria, and later redistributed to New South Wales and Tasmania, the horehound clearwing moth, from Spain, has established. With only one generation per year, it can take ten or more years for populations to build up and disperse. Despite this, its impact, while localised, is significant with larvae killing most attacked plants (Weiss and Sagliocco, 2012).

Identification

Adult horehound clearwing moths are dark brown (up to 10 mm long), with fine white to yellow lines or markings across their abdomen. With a wingspan of around 12 to 14 mm, their wings have clear windows with dark brown edges. Larvae are cream

J. Weiss

Horehound clearwing moth.

P. Sullivan

Horehound clearwing moth larva.

P. Sullivan

Horehound clearwing moth pupa.

coloured with a dark brown head capsule. Damage is characterised by larvae tunnelling within the stems and roots.

Life cycle

Horehound clearwing moths have one generation per year. Adult females lay around 96 eggs at the base of plants over a one to two-week period in late spring (Sagliocco and Coupland, 1995). After hatching, larvae crawl to the base of plants where they bore a hole into the plant's roots and feed. Usually only one larva develops per root through the summer, autumn and winter period before they pupate within the root crown or lower stem, emerging in late spring. Prior to pupation, late instar larvae bore an exit hole in the root crown or base of the stem.

Field collecting, rearing and redistribution

Rearing and redistribution of the horehound clearwing moth is extremely difficult and requires considerable entomological expertise, equipment and a lot of time. Current best practice involves collecting rootstock containing pupae early in October and regular site visits to provide food for the moths in addition to releasing females from your enclosure. See Appendix 1 for 'field cage technique' for details on rearing and redistributing your moths. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Look for horehound clearwing moths by checking the root crowns or lower sections of the stems for small, slightly pinkish areas that surround the moth's emergence holes within a year of release. You can destructively sample rootstocks and lower stems to look for larval or pupal presence from autumn until spring by cutting the stems (described above). If stems are hollowed out and appear green, look further as larvae are likely to be present within. Record its presence or absence as per your monitoring guidelines (Appendix 3) and begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

If rearing and redistributing the horehound clearwing moth you will need to differentiate between female and male moths. Males' abdomens end with a fan-shaped tuft.

P. Sullivan

Female clearwing moth.

P. Sullivan

Male clearwing moth with fan-shaped tuft on end of abdomen.

Lantana

Lantana camara

Lantana is a woody, rambling, shallow-rooted perennial shrub thought to have originated from two or more lantana species from tropical America (Day *et al.*, 2003; Sanders, 2001). There are some 29 different forms or taxa naturalised in Australia, which vary in the form and colour of the flowers. Due to difficulties in distinguishing between each variety, six main flower colours differentiate varieties for convenience[1]. Colours include common pink, Hawaiian pink, pink-edged red, red, white, and orange. Lantana occurs in coastal and subcoastal regions from Torres Strait to the New South Wales-Victorian border. It also grows to a lesser extent in Western Australia and the Northern Territory (Day 2012a). Plants have four-angled stems and spearheaded bright green opposite leaves (up to 10 cm long), with serrated edges. Leaves are rough to touch and are fragrant when crushed. Flowering occurs year-round if there is adequate moisture in the soil. The brightly coloured flowers (about 2.5 cm in diameter) can deepen in colour with maturity. Round berries transition in

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Different lantana flowers.

R.G. Richardson

Lantana invading the Townsville Hinterland, Queensland.

[1] Due to a long history of cultivation, hybridisation, and invasiveness, the taxonomy of invasive *Lantana camara* is complex and unclear. Unravelling differences by physical appearance alone is impossible, but genetic research is assisting to unravel the mystery, and determine whether clear affinities between species exist or whether weedy forms of lantana in Australia result from one highly variable hybrid swarm. For biocontrol, pinpointing the native range of the target weed is imperative for facilitating the exploration for the best-adapted and most effective natural enemies. In the case of a hybrid form, weedy lantana possesses no native range as such, but tracing its ancestry assists biocontrol researchers to narrow down countries and species for conducting natural enemy surveys.

colour from green to black as they mature and are readily dispersed by animals, water, machinery and garden waste. Mature plants can produce up to 12,000 seeds every year and seeds can remain viable in the soil for several years under natural conditions. Lantana can reproduce vegetatively by stems touching the ground sending roots into the soil. The plant is also allelopathic, allowing it to form dense impenetrable thickets (CRC, 2003).

Introduced as a garden ornamental plant in the 1840s, first into South Australia and later into New South Wales, lantana by the late 1850s, had established outside of cultivation. Lantana was first considered as a pest in 1879 and by the1950s, it had naturalised over much of coastal and subcoastal areas of eastern Australia (Swarbrick, 1986). Its ability to release chemicals into the surrounding soil, thereby preventing native seedling germination, exacerbates its potential to spread in natural forests and agricultural systems. By 1999, lantana was listed as a Weed of National Significance due to its invasiveness, environmental and economic impacts, and potential to spread across northern Australia and west of the Great Dividing Range.

Since 1914, Australia has released 33 species of insects, a bud mite, and a pathogen from tropical America against lantana (Day, 2012a). Of these, 18 established. Together, these agents cause variable impacts on lantana, with damage highest in late summer or autumn. Four of the most damaging agents: the lace bug (*Teleonemia scrupulosa*), the leaf-mining hispine beetle (*Uroplata girardi*) the leaf-mining beetle (*Octotoma scabripennis*), and the stem-sucking bug (*Aconophora compressa*), are described in detail. All are widespread in the sub-tropical and temperate regions of eastern Australia and have demonstrated a substantial but seasonal impact on lantana (Day *et al.*, 2003). Following this all other agents that have established in Australia will be briefly described. We encourage you to report sightings and observations that may represent potential dispersal and impact to your local weed or biosecurity officer and via the Australian Biocontrol Hub.

Recommendation

Many of these biocontrol agents have been established for decades and are already widespread and occupying areas that are climatically suitable. Therefore, re-distribution of agents is not recommended. As damage by biocontrol agents is seasonal and will not permanently suppress lantana, integrated management taking advantage of when lantana is heavily damaged by the agents is strongly recommended. Refer to the Lantana Best Practice Manual and Decision Support Tool available via the Literature & Links tab at https://profiles.ala.org.au/opus/weeds-australia/profile/Lantana camara. Here you will find a variety of options for integrating conventional methods (e.g. manual, mechanical, chemical, use of fire, pasture improvement, grazing management and revegetation) with biocontrol in your patch.

The lace bug
Teleonemia scrupulosa

First released in 1936 in Queensland and widely redistributed throughout Queensland, New South Wales and Norfolk Island, the lace bug is one of the most widespread and seasonally damaging biocontrol agents for lantana in Australia (Day, 2012a). Sapsucking adults, and nymphs feeding in colonies, cause plant defoliation from late summer to autumn which substantially reduces flowering and seed production. Greater numbers occur on the white, red and pink edged red flowering varieties. The bug is more abundant and damaging in warm drier areas, such as exposed regions of subcoastal New South Wales, and southern and central Queensland, than areas with high rainfall.

Identification

The adult lace bug is 3 to 4 mm long with a mottled brown elongate-oval body with a slight expansion near their middle and an obscure dark brown "X" pattern on their forewings. Nymphs look completely different to adults, they are brownish-yellow and bear spines around their abdomen. Over the five growth stages (instars), nymphs get progressively larger; with wing buds appearing in the last two instars. Damage by both adults and nymphs feeding on leaf undersides is more prominent on the upper surface of leaves. Infested leaves show dark brown to black scorched or burnt areas, and with excessive feeding leaves turn yellow, curl and can become white prior to dropping from the plant. Lace bug damage is further confirmed by the presence of black droplets of excrement on the undersides of the damaged leaves.

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Adult lace bug.

K. Harley

Lace bug nymphs.

QDAF

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Scorched damage marks by the lace bug and lace bug plant damage.

Life cycle

The lace bug has 10 to 11 overlapping generations per year, with each taking about a month. Adult females will begin laying eggs within five to six days of becoming an adult. She partially inserts clusters of 10 to 30 eggs into the midrib and main veins on the underside of leaves in her three-month lifespan (Harley and Kassulke, 1971). Within eight days, eggs hatch and the new nymphs aggregate into colonies and begin feeding on leaf undersides, and newly opened buds and flowers. During this time, nymphs undergo five growth stages (instars) that can take approximately two weeks before they become active winged adults. Both adults and nymphs feed on cell contents using their piercing and sucking mouthparts on leaf undersides producing varnish-like spots and black droplets of excrement. Insects become less active and populations decrease over winter when it is cooler.

Monitoring for numbers and impact

Look for lace bug adults and nymphs on the underside of leaves on lantana. If present, record your sighting on the Australian Biocontrol Hub. Monitor for its presence and potential impact annually as per your guidelines (Appendix 3). When numbers begin to decrease at the end of autumn, consider implementing other control methods to help manage lantana. Consult the Lantana Best Practice Control Manual.

The leaf-mining hispine beetle
Uroplata girardi

First released in 1966 in Queensland and widely redistributed throughout Queensland, New South Wales and Norfolk Island, the leaf-mining hispine beetle is also one of the most widespread and seasonally damaging agents of lantana in Australia (Day, 2012a). Grazing by adults on the upper surfaces of leaves and leaf-mining by larvae can cause severe defoliation of lantana from late summer through to autumn which can substantially reduce flower and seed production. Found on all varieties of lantana, the leaf-mining beetle is more common in warm areas. Commonly found with another leaf-mining beetle *Octotoma scabripennis*, together their damage is similar and complementary, resulting in substantial defoliation of plants seasonally. Populations from both species, however, decrease in the dry winter months when plants become leafless, reducing their ability to suppress lantana permanently.

Identification

The leaf-mining beetle is around 8 mm long, with a shiny brown body and several large golden-brown spots on rectangular corrugated wing covers. Damage inflicted by adults appears as small scarification marks on the upper leaf tip's surface. In time, damage causes the leaf tips to curl and provide shelter for adults. While larvae are unseen, they form mines in the form of trails through the middle layers

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Adult leaf-mining hispine beetle.

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Mining by hispine beetle larvae.

of leaves. Two to three mines can be visible on the upper leaf surface, with each mine containing a single larva.

Life cycle

The leaf-mining beetle can produce up to three generations per year, with each taking around 31 to 52 days (Harley, 1969). Adult females lay eggs singly into leaf tissue, usually at the edge of an adult feeding scar, and cover it with frass (waste products). After emergence, larvae mine and feed on leaf tissue through the middle layer of leaves. Larvae complete three instars of development before pupating within the leaves, away from the centre vein. Adults can live for six to nine months. During winter adults may enter a period of inactivity in the leaf litter.

Monitoring for numbers and impact

Look for signs of the beetle either by checking younger leaves, especially where they curl, or by the extent of mining on the leaves. Heavily mined leaves indicate large populations of the beetle. If present, record your sighting on the Australian Biocontrol Hub. Monitor for its presence and potential impact annually as per your guidelines (Appendix 3).
As permanent suppression of lantana is unlikely through biocontrol, other control methods could be considered in late autumn when insect numbers are at the highest and damage is greatest. Consult the Lantana Best Practice Control Manual.

The leaf-mining beetle
Octotoma scabripennis

First released in 1966, the leaf-mining beetle is one of the most widespread and seasonally damaging biocontrol agents for lantana in Australia (Day, 2012a). Grazing by adults on the upper surfaces of leaves and leaf-mining by larvae, can cause severe defoliation of lantana from late summer to autumn, which can substantially reduce flower and seed production. Found on all varieties of lantana, the leaf-mining beetle is most abundant in subtropical, shady, wet coastal areas, particularly from Kempsey, New South Wales through to Bundaberg, Queensland. Commonly found with the other leaf-mining beetle *Uroplata girardi*, together their damage is similar and complementary, resulting in substantial defoliation of plants seasonally. Populations from both species, however, decrease in the dry winter months when plants become leafless, reducing their ability to suppress lantana permanently.

Identification

The leaf-mining beetle is around 8 mm long, with a dark rustic black body and metallic sheen. Damage inflicted by adults appears as small scarification marks on the upper surface of leaves. While larvae are unseen, they form mines through the middle leaf layers, causing dark blotches on the surface.

K. Hignell

Adult leaf-mining beetle.

QDAF

Scarification and mining damage by the leaf-mining beetle.

Life cycle

The leaf-mining beetle can have up to three generations per year, with each taking around 34 to 45 days (Harley, 1969). Adults can live for six to nine months. Females lay their eggs on the edge of adult feeding scars. After emergence, larvae mine and feed on leaf tissue through the middle layer of leaves. They go through three instars before pupating within the leaves, on the centre vein. During winter, adults may enter a period of inactivity, often hiding in the leaf litter (Day *et al.*, 2003).

Monitoring for numbers and impact

Look for signs of the leaf-mining beetle by checking younger leaves. Heavily mined leaves indicate large populations of the beetle. If present, record your sighting on the Australian Biocontrol Hub. Monitor for its presence and potential impact annually as per your guidelines (Appendix 3). As permanent suppression of lantana is unlikely through biocontrol, other control methods could be considered in late autumn when insect numbers are at the highest and damage is greatest. Consult the Lantana Best Practice Control Manual.

The stem-sucking bug
Aconophora compressa

First released in 1995, the stem-sucking bug is now widespread along coastal and subcoastal areas of eastern Australia from Sydney to Gladstone, as well as around Mackay and the Atherton Tableland. Sapsucking adults and nymphs feeding in colonies on the sap of woody stems leads to leaf drop and branch death resulting in reduced flowering and seed set in lantana, particularly from November through to February. Found on all varieties of lantana the stem sucking bug prefers open, cool dry conditions, (particularly between Kempsey, New South Wales and the Capricorn Coast of central Queensland), and tends not to establish in areas of high humidity.

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Adult stem-sucking bugs.

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Clusters of stem-sucking bug nymphs.

Identification

The stem-sucking bug is a light brown treehopper up to 8 mm long, with a thorn-shaped body and clear wings with dark longitudinal veins. Nymphs are white with black stripes. Damage inflicted by adults and nymphs feeding in groups leads to browning of the leaves and stems before leaf drop and branch death.

Life cycle

The stem-sucking bug has four to five generations per year, with each taking around 40 days. Adults live for up to six months and females lay eggs in batches of up to 65 on the stems of plants. Females guard their young against predators until mature, with nymphs taking from 28 to 42 days for their development across five instars. Populations increase over the winter period before dying off over the summer period in high temperatures (Day *et al.*, 2003).

WARNING!

This insect also attacks fiddlewood, *Citharexylum spinosum*, an exotic tree from the Caribbean often used as an ornamental plant. The insect does not kill fiddlewood trees, but high populations can cause leaf drop and a large production of honeydew. Fiddlewoods may need to be chemically treated with insecticides to provide control of the agent. Contact your local council for information.

QDAF

QDAF

Before control (top) and after control (bottom) by the stem-sucking bug.

QDAF

Stem-sucking bug damage.

Field collecting and rearing

Rearing is unnecessary. This insect is widespread, but it may still be spreading from areas where it has established. As populations can be highly variable throughout the year and within regions, it is possible to move the insect around to aid establishment in areas where it is not already present. Look for damaged lantana and the presence of clusters of insects along the stems, near the tips. Cut stems containing the insect and place them in a sealed container with ventilation prior to redistribution.

How and when to release

Place cut stems containing insects in new thickets of lantana. To increase the chance of establishment, 500 to 1000 individuals should be released. Place stems inside clumps of lantana so the insects do not get attacked by birds before they have a chance to move off onto the plant. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Within one year of release at your nursery site, look for adults and nymphs feeding in colonies or signs of damaged leaves and stems that turn brown with feeding. Record presence or absence as per your monitoring guidelines (Appendix 3). Monitor annually.

Other lantana agents that have established in temperate Australia

- The bud mite, *Aceria lantanae*, is well established in northern Queensland, but with limited establishment in south-eastern Queensland. The bud mite establishes on many varieties of lantana but prefers red or pink-edged red flowering varieties. Mites feed on new flower-buds causing galls of up to 20 mm in diameter which ultimately leads to reduced flowering and seed productivity. Its impact post release is still unknown.

- The flower-feeding moth, *Lantanophaga pusillidactyla*, while accidentally introduced, is one of the most widespread biocontrol agents for lantana in Australia. Found on all varieties of lantana, the moth tolerates a range of climatic conditions and is found from south of Sydney through to far north Queensland. While common, impact is minimal. Adults feed on nectar whereas larvae feed within the flower heads and receptacle.

- The flower and bud-feeding moth, *Crocidosema lantana*, is widespread throughout the invasive range of lantana in Australia but is more common in warmer coastal areas occurring from Ulladulla in southern New South Wales through to Cooktown in far north Queensland. While it is found on all varieties of lantana, it is seasonally abundant and does not appear to have a significant impact on lantana despite larval feeding. Larvae feed on shoot tips and flower heads to reduce flowering and seed productivity.

- The leaf-feeding moth, *Hypena laceratalis*, is common in the subtropical and tropical regions of eastern Australia, particularly from Kempsey New South Wales through to Cow Bay in far

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Fresh flower gall damage by the bud mite, Aceria lantanae.

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Flower-feeding moth, Lantanophaga pusillidactyla: adult (left) and larva (right).

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Flower and bud-feeding moth, Crocidosema lantana: adult (left) and larva (right).

north Queensland. It is found on most varieties of lantana. Damage by larvae is predominately seasonal and localised, having an overall minimal impact on lantana. Adults feed on the nectar of flowers while larvae feed on the lower and middle parts of leaves, leaving the upper leaf surface scarred, with a window-pane effect.

QDAF

Leaf-feeding moth, Hypena laceratalis: adult (left) and larva.

- The leaf-feeding moth, *Neogalea sunia*, is common and widespread in drier areas of northern New South Wales through to the subcoastal regions of southern Queensland and is found on most varieties of lantana. Impact is highly seasonal and substantial plant defoliation may occur when combined with other biocontrol agents. Adults feed on nectar while larvae feed on leaves and flowers.

QDAF

Leaf-feeding moth, Neogalea sunia: adult (left) and larva.

- The leaf-feeding moth, *Salbia haemorrhoidalis*, is abundant in tropical areas and subtropical regions from Coffs Harbour on the mid-north coast of New South Wales to Cow Bay in far north Queensland and is found on most varieties of lantana. Damage is moderate from late summer and autumn and best combined with other biocontrol agents to cause severe defoliation of leaves. Adults feed on flowers while larvae feed within folded leaves, which they fasten together with silk.

QDAF

Leaf-feeding moth, Salbia haemorrhoidalis: adult (left), larva.

- The leaf-mining hispine beetle, *Octotoma championi*, has established in north Queensland and south of Sydney in low numbers on pink and pink-edged red flowering varieties of lantana. Impact is slight, as populations are somewhat low for complete effectiveness, but most damage occurs in late summer through to autumn. Adults feed on the upper leaf surface while larvae mine within the leaf. Adults are generally smaller than *Octotoma scabripennis*.

QDAF

Leaf-mining hispine beetle, Octotoma championi: adult (left) and its damage (right).

- The leaf-mining fly, *Calycomyza lantanae*, is widespread from Cooktown Queensland to Kempsey New South Wales and is found on all varieties of lantana. Populations are slow to build up in more temperate regions and as such, result in minimal damage compared to the tropics. Adults feed on flowers while larvae mine within the leaf forming blotches.

- The rust, *Prospodium tuberculatum*, is widespread in eastern Australia and is highly specific, occurring only on the pink variety of lantana. Establishment is greatest in wetter mountain regions of northern New South Wales and southern Queensland, but the rust can establish in the central regions of New South Wales. Damage is greatest in the summer months coinciding with higher summer rainfall. The rust causes a leaf infection in the form of dark, purple-brown lesions that can be irregular in shape. Pustules on the underside of leaves are raised and look like tiny coffee granules. Damage is highly seasonal but high infections can lead to leaf chlorosis and premature abscission.

- The seed-feeding fly, *Ophiomyia lantanae*, is widespread in eastern Australia and can be found on all varieties of lantana. Due to the enormous quantities of fruit lantana produces, the impact of this agent is slight. Greatest damage occurs in late summer through to autumn, particularly in moist warm areas where lantana flowers readily. Adults feed on nectar whereas larvae feed on the fleshy pulp of seeds. Birds tend not to eat infected berries, thus reducing spread.

For further information on many of these agents, refer to the Queensland Department of Agriculture and Fisheries fact sheets:

https://www.daf.qld.gov.au/__data/assets/pdf_file/0009/62010/lantana.pdf

QDAF

Leaf-mining fly, Calycomyza lantanae: adult (left) and its damage (right).

NSW DPI

Lantana rust, Prospodium tuberculatum.

QDAF

The seed-feeding fly, Ophiomyia lantanae: adult (left) and its damage (right).

https://www.business.qld.gov.au/industries/farms-fishing-forestry/agriculture/land-management/health-pests-weeds-diseases/weeds-diseases/invasive-plants/restricted/lantana

If any of these agents are present at your site, record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for their presence annually as per your monitoring guidelines (Appendix 3).

Madeira vine
Anredera cordifolia

Madeira vine is a vigorous perennial climber or scrambling shrub native to South America (Palmer and Senaratne, 2012). It typically grows in riparian vegetation, rainforest edges and tall open forests in wetter temperate, sub-tropical and tropical regions of coastal Australia, but as its range expands, it is has been found to penetrate into dryer inland areas. Plants have fleshy, heart-shaped (cordate) leaves (2 to 15 cm long) and slender hairless green or reddish young stems (up to 30 m long) that become woody with age. Also commonly known as lambs tail, the aromatic, white, or cream-colored drooping flower spikes (up to 30 cm) resemble a lamb's tail over the summer and autumn flowering period. Plants spread predominately through the production of thousands of greyish brown or greenish warty aerial and underground stem tubers (up to 10 cm), which are easily dispersed in water, and garden or contaminated waste. Tuber densities are high (up to 1500 per m^2) and can remain viable for up to 15 years. In Australia, viable seeds are generally not produced (Vivian-Smith *et al.*, 2007).

NSW DPI

Madeira vine infestation.

NSW DPI

Aromatic drooping flower spikes of Madeira vine.

NSW DPI

Warty aerial stem tubers.

Madeira vine was introduced as a garden ornamental plant in the early 1900s (Floyd, 1989), and was first reported as naturalised in New South Wales in the 1960s. Deemed as problematic by the 1980s over large areas of New South Wales and Queensland, the irreversible damage caused by Madeira vine, led to its later categorisation as a 'transformer' species. Its climbing habit, ability to establish under closed canopies and fast growth rate (exceeding one metre per week) can result in the smothering of forest canopies which inevitably leads to canopy collapse of mature forest trees and a complete restructuring of communities. Currently regarded as one of the five most invasive plants in south-eastern Queensland, Madeira vine was listed as a Weed of National Significance due to its environmental impacts, invasiveness, potential to spread, and difficulty to control.

Madeira vine

Australia introduced two species of biocontrol agents to test their potential to control Madeira vine from a collaborative biocontrol program between South Africa and Australia (Palmer and Senaratne, 2012). One of these, the Madeira vine leaf-feeding beetle (*Plectonycha correntina)* was released and is established at various sites across New South Wales and Queensland.

Recommendation

Control of Madeira vine is a long-term process which requires regular follow-up until all tubers and bulbils are removed or killed. Integrated control of Madeira vine is recommended using biocontrol, physical removal and chemical control.

Madeira vine leaf-feeding beetle
Plectonycha correntina

First released in 2011 in south-eastern Queensland, the Madeira vine leaf-feeding beetle is still undergoing redistribution in Queensland and New South Wales. Both adults and larvae of this insect are leaf feeders, with larvae being particularly damaging. Commensurate damage aims to reduce the leaf area, thereby reducing the plant's rate of photosynthesis, and forcing it to draw upon its stored resources (tubers), which will eventually deplete. Establishment is currently variable, but when numbers build up, larvae and adults can be quite damaging, but the impacts are also highly variable and driven by microclimate. As a result, further redistributions to build populations up are required in addition to utilising a range of other management techniques integrated with biocontrol.

Identification

The adult leaf-feeding beetle is approximately 5 mm long and identified by its orange to brown body and sometimes-absent 14 black spots and orange legs. Clear windowpane scars on the top of leaves and shot holes occur from adults feeding on the underside of leaves. Larvae are more effective in reducing the leaf area than adults. The larvae emerge as small, white then butter-yellow grubs with black heads (up to 4 mm long) and become covered in a protective black, gelatinous substance upon feeding. Chewed leaf margins are commonly observed as the larval slimes travel from leaf to leaf devouring entire leaves as they go. Groups of tiny cylindrically-shaped eggs (0.8 mm long) are cream-yellow and commonly laid in two rows, in groups of eight to 15 eggs, on the underside of leaves (Palmer and Senaratne, 2012).

K. O'Donnell

Adult leaf-feeding beetle.

L. Snow

Shot hole damage and windowpane scars from feeding by adult leaf-feeding beetle.

Life cycle

The leaf-beetle has several generations per year, with each lasting up to eight weeks. Their lifespan ranges from 20 to 130 days. Highly fecund females lay approximately 550 to 800 eggs in batches of one to 35 on the underside of leaves (Snow *et al.*, 2012). Newly hatched larvae remain on the underside of the leaves to feed voraciously, and once they start, they become covered in a gelatinous black coating to protect them against predators. Older larvae feed on leaf margins and begin to migrate to the lower sections of the plant before shedding their gelatinous cover and burrowing into the soil to pupate and emerge as adults approximately 20 days later. Like larvae, adults also feed on the foliage and are found on the underside of leaves.

K. O'Donnell

L. Snow

Leaf-feeding beetle larvae (top) progressing to being covered in a protective gelatinous substance (bottom).

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Collecting adult beetles and insects ready for transport.

Field collecting and rearing

Collect adult beetles and larvae for redistribution from September to March. Adults are best collected from the leaf undersides in the middle of the day when they are most active. Place a vial, small container or tray directly behind and below the beetle, and gently touch it so that it will fall backwards into the container. Larvae can be collected throughout the day by carefully removing individual leaves containing the larval slimes. Prior to redistribution, adults and larvae can be stored temporarily (at around 20°C for a few days) in sealed containers containing some madeira vine leaves and covered with either a lid with small air holes or insect mesh for ventilation.

Alternatively, beetles can be reared in insect proof cages containing potted plants over an approximately eight to twelve-week period. On

average, six potted plants can fit in a 1200 mm length × 700 mm width × 330 mm depth cage lined with insect mesh. This type of cage can support the progeny of up to 30 adults (at temperatures in the mid to high 20°C range). To facilitate pupation, a layer of potting mix (3 to 5 cm in depth) is required across the bottom of the cage. After a period of three months, each cage can produce up to 1000 adults. These must be removed immediately for redistribution or placed in new cages with new food.

To maintain a good supply of food for beetles, host plants need to be prepared prior to setting up the rearing cage. Allow host plants to grow enough foliage by providing plenty of water and apply one treatment of liquid fertiliser if required. Once the host plants have grown to a height of about 30 cm (usually two to three weeks in summer) and contain a good amount of thick lush foliage, it is time to prepare the rearing cage. Place a 3 to 5 cm thick layer of potting mix on the bottom of the cage. Place the pots/trays inside the cage and on top of the layer of potting mix, then add insects and secure the cage to prevent them from escaping.

For the first four weeks or so, continue watering the base of the plants. Avoid wetting foliage as this can encourage leaf spot or other diseases on the host plants. You will notice an increase in the development of larval slimes. Once larvae have defoliated the host plants they will travel to the base of the plants and prepare to pupate. Keep soil conditions moist, but not wet, for adult emergence. Upon emergence, adult insects may be collected and used for field redistribution or establishing additional rearing cages. Keep cages stocked with fresh plant material to maximise production.

How and when to release

Release beetles (adults and larvae) directly onto healthy plants as soon as possible. To assist establishment, select sites with a northern aspect (in full sun) and avoid flood or frost prone sites. Aim to release about 400 beetles for each site by making a small nest of attached leaves within a healthy Madeira vine thicket directly above the ground. Open the container and pour the contents of the container, including adult insects, larvae and foliage onto a small area of the vine. The beetles will seek out their own preferred location from there. Release adults and larvae at any time, except during winter. Record release information as per the weed biocontrol release form (Appendix 2) and submit a copy to the local weed or biosecurity officer.

Monitoring establishment and dispersal

On the underside of leaves look for presence of adult and larval leaf-beetles and their feeding damage (windowpane scars and shot holes (adult) and chewed edges (larvae)) at the nursery site within one year of release. Once located, look more closely, especially on the lower side of leaves, for egg clusters. Record their presence or absence as per the monitoring guidelines (Appendix 3) and if present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the guidelines. Monitor annually.

Mistflower

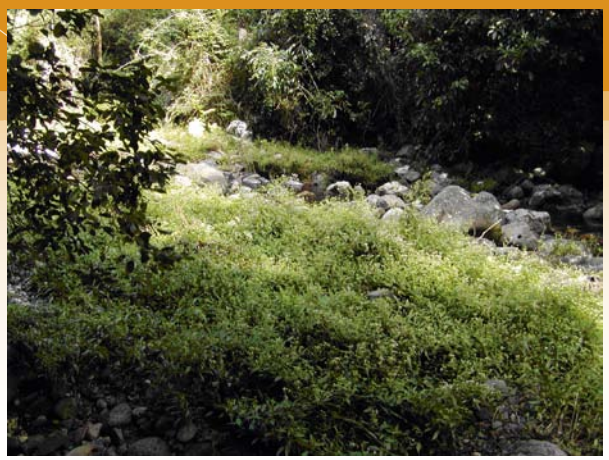
Ageratina riparia

Mistflower is a low growing (up to 1 m), scrambling perennial herb native to Mexico and Central America (Schooler *et al.*, 2012). It is most abundant in the high rainfall (>1700 mm per annum) forested coastal regions of temperate to tropical regions of eastern Australia. Plants have narrow oppositely arranged leaves (up to 7 cm long) with toothed margins, and small white flowers that cluster together at branch tips. Flowering occurs en masse from late winter through to late spring before forming thousands of small wind-dispersed seeds allowing the plant to colonise habitats upstream and upslope. As seeds float, mistflower is readily dispersed by water, but also by animals, on clothing, by vehicles or with machinery.

R. Holtkamp

Flowers and foliage of mistflower.

Introduced as a garden ornamental plant in 1875 in New South Wales, and recorded in the botanic gardens of Adelaide, Melbourne, Sydney and Brisbane, mistflower quickly escaped cultivation and spread north up into Queensland where it was first recorded naturalised in 1930 (Schooler *et al.* 2012). Mistflower is now recognised for its aggressive growth habits and is listed within the top 25 most invasive weeds for Queensland's southeast. Unlike



R. Holtkamp

Mistflower invading a riparian understory.

most weeds, mistflower can spread along riparian corridors into pristine catchment headwaters, where it forms canopies over headwater streams. It is shade tolerant and quickly dominates by forming a mat of layered and interwoven stems that exclude native plants and animals reliant upon these riparian corridors.

Australia imported two species of insects, including a gall fly (*Procecidochares alani*) in 1985 and a plume moth (*Hellinsia beneficus*) between 1985 and 1988, from existing biocontrol programs against mistflower in Hawaii (Schooler *et al.*, 2012). Of these, only the gall fly was released in 1986 and has established. In 2010, the white smut fungus (*Entyloma ageratinae*) was recorded on mistflower near Lamington National Park in south-eastern Queensland. It was likely to have been introduced unintentionally from New Zealand, where there is an existing biocontrol program against mistflower, and made its way to Australia on unclean hiking equipment. Highly successful, it has since been widely redistributed and is now widespread across the invaded range of mistflower in Australia (Schooler *et al.,* 2012).

Recommendation

When looking for agents in the field, keep in mind that the white smut fungus prefers wetter, cooler areas and affects the lower leaves first, whereas the gall fly is found in drier, warmer areas and affects the upper parts of the plant.

Mistflower gall fly
Procecidochares alani

First released in Queensland in 1986, the mistflower gall fly is now widespread throughout the invaded range of mistflower (Schooler *et al*., 2012). Plant growth is retarded by commonly seen gall development. However, the mistflower gall fly has proven ineffective in controlling mistflower, possibly due to native parasitism.

Identification

Adult gall flies have boldly patterned wings (brown patterns with see through patches in between) and a wingspan of approximately 8 mm. Larvae are cream-coloured (1 to 2 mm long) and their feeding activity induces the formation of galls on the growing tips. The galls are the most easily identifiable feature of gall fly presence on affected plants. Galls are the same colour as the stem and can be as large as 25 mm long and 10 mm diameter (Schooler *et al*., 2012). Larvae complete their development in the gall and prepare emergence holes (transparent windows) in the cuticle of the gall shortly before pupation. Pupae have a brown pupal case.

F. Kynd

Adult mistflower gall fly with characteristic wing patterns.

S. Thorpe

Mistflower gall showing adult fly emergence holes.

Life cycle

The mistflower gall fly has multiple generations per year, with each generation taking approximately six weeks depending on temperature and plant quality. Adult gall flies live for about two weeks and are active during the day (diurnal) but do not feed (Schooler *et al*., 2012). Females deposit eggs on the top (apex and auxiliary) buds of the plant. Eggs hatch in three to five days and larvae feed within the buds inducing gall formation. Galls may contain up to 15 cream-coloured larvae, that once fully developed, pupate within the gall. At the end of pupation, adults escape from the galls through thin tissue left behind by the feeding larvae, leaving a noticeable adult fly-sized hole. Gall flies are known to breed continually over the warmer months of the year. The average number of generations per year in the field is not known, but is likely to be three or more depending on conditions. Impact is low to moderate, probably due to parasitism of the larvae and pupae (Schooler *et al*., 2012).

Field collecting and rearing

Rearing is unnecessary. The gall fly is widespread throughout the distribution of mistflower in Australia and generally does not require redistribution. However, should mistflower populations be located where no signs of the gall fly can be detected, then cuttings containing galls without emergence holes (minimum of 100) can be collected from established sites during summer and autumn (see Appendix 1 for cuttings technique).

How and when to release

Place the cuttings containing galls on the ground within the mistflower infestation at the new site. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

The easiest way to check for gall fly establishment, within a year of your release, is to look for galls during summer and autumn. Also look out for adult flies which are active during the day throughout the warmer months. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

Recommendation

Release the gall fly at warmer, drier sites, whereas the white smut fungus should preferably be released at sites which receive regular rainfall and are generally cooler and wetter.

L. Morin

Left – mistflower growing in Kangaroo Valley in southern New South Wales prior to flowering. Right – same site showing native plant regeneration after mistflower was killed by the white smut fungus.

White smut fungus
Entyloma ageratinae

First recorded on mistflower in Lamington National Park (south-eastern Queensland) in 2010, the white smut fungus provides good control of mistflower (Schooler *et al.*, 2012). The origin of this white smut fungus infection is thought to be the result of an accidental introduction from New Zealand. Redistribution of the white smut fungus actively occurred after confirmation of its host specificity on mistflower. It is now widely established across the invaded range of mistflower in Australia (Morin *et al.*, 2012). The fungus causes rapid, major defoliation that provides complete control of mistflower particularly in humid and wet areas. Reduced impact is observed in areas where rainfall and humidity are low.

Identification

Symptoms of the white smut fungus are predominately seen when the daytime temperature is between 10 and 20°C, where there is high humidity and when there has been enough rain to promote active plant growth. The white smut fungus appears as cottony white spores on the lower side of the leaves, and later red lesions on the top of the leaves that turn black and merge together with time.

L. Morin

Mistflower killed by the white smut fungus.

As white smut fungus destroys plant tissue, the leaves wither, die and fall to the ground. Severely diseased plants do not flower and thus do not set seed. Mistflower does regrow from stems or roots of infected plants, but reinfection of regrowth is rapid, and the plants die.

Life cycle

The white smut fungus needs high humidity and mild temperatures of between 10 and 20°C to infect mistflower leaves. Lack of humidity is thought to limit its success in areas with lower rainfall. Small lesions appear on the lower surface of leaves seven to 14 days after infection. These lesions produce copious amounts of spores that have a white woolly appearance. As the infection progresses, angular reddish-brown lesions appear on the upper surface of the leaves and the infection sites become necrotic. Eventually the lesions coalesce, and the leaves die and fall off. Plant death from severe infections occurs after several months.

Field collecting and rearing

Rearing is unnecessary. The white smut fungus is distributed across the invasive range of mistflower in Australia and generally does not require redistribution. However, should mistflower populations be located where no signs of the white smut fungus can be detected, redistribution can be achieved by translocation of infected potted plants.

Note: Do not dig up and pot white smut fungus infected mistflower plants from an established site and transfer them to a release site far away, as this constitutes a biosecurity risk. Instead, grow mistflower plants in pots with a commercially prepared medium and then place these at an established site until white smut fungus symptoms appear on plants. Then move the pots to the intended release site.

L. Morin

White fruiting bodies of white smut fungus are found on the lower leaf surface of mistflower leaves 7 to 10 days after infection.

L. Morin

Rust-coloured lesions appear on the top of mistflower leaves as the tissue turns necrotic due to the infection of the white smut fungus on the lower leaf surface.

How and when to release

Release the white smut fungus by placing infected potted plants at suitable sites during wet, milder months when local humidity is higher. Aim for 10 infected plants per site. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Follow-up monitoring of release sites should take place a month after inoculation to check for signs of infection. Look initially for woolly white spores on the lower surface of leaves. For longer established sites, defoliated plants should be evident or look rather unhealthy (with retained leaves) with reddish-brown lesions that turn black on their upper surfaces. If there are brownish red lesions on the upper surface of leaves, without the woolly white spores below, then the infection is not white smut fungus but a species of *Phoma*. The best time to look for establishment is three to four weeks after good rainfall has occurred and when the daytime temperatures are mild (around 20°C). If white smut fungus is present, begin monitoring for dispersal at incremental distances away from the site of inoculation as per your monitoring guidelines (Appendix 3). Monitor annually.

Noogoora burr complex

Xanthium chinense and *Xanthium orientale*

Noogoora burr (formerly *Xanthium occidentale*) is an annual, erect herb native to the southern United States of America (USA), the West Indies and Mexico (van Klinken and Morin, 2012). Recent DNA barcoding and next-generation sequencing has condensed four species in the Noogoora burr complex present in Australia down to two genetic groups (Charles *et al.*, 2019). The first group called *Xanthium chinense* is made up of *X. occidentale* (Noogoora burr) and *Xanthium orientale* (California burr); the second group called *X. orientale* encompasses *Xanthium italicum* (Hunter burr) and South American burr, *Xanthium cavanillesii*. These names were chosen because they matched vouchered specimens in a review by Tomasello (2018). The Charles *et al.* (2019) study also found many hybrids of the two species groups, which means field identification of species in the Noogoora burr complex is not possible based on its physical appearance alone. There are no native *Xanthium* species in Australia.

Noogoora burr is the widest spread of the naturalised *Xanthium* species in Australia and is thought to have been introduced with imported cotton seed in the 1860s (Parsons and Cuthbertson, 2001). By the 1950s Noogoora burr was a major weed in Australia. It is toxic to livestock, reduces stock carrying capacity, contaminates wool, and is expensive to control. It invades cultivated, grazing, and undeveloped areas of Australia's temperate, sub-tropical and tropical regions. Noogoora burr favours fertile soils that are subject to summer rainfall, flooding, or irrigation (Parsons and Cuthbertson, 2001). Plants grow between 1 and 2.5 m high and can be single stemmed or have a much-branched spreading habit. Stems are hairy and have purple/green mottling. Leaves are are grapevine-like and are arranged alternately on the stems. Each leaf can be up to

J. Gasparotto

Small Noogoora burr plant.

B. Auld and R. Medd

Noogoora burr plant with immature burrs.

S. Johnston

Noogoora burr plant after leaf drop showing mature burrs.

15 cm in diameter and has three to five lobes. The flowers are inconspicuous. The resulting burr is a conspicuous, woody fruit that is armed with many hooked spines and two terminal beaks.

From the 1930s, three insect species were studied to test their potential against Noogoora burr. All were approved for release in Australia, including a North American seed fly (*Euaresta aequalis*) and two stem-boring beetles; one from North America (*Mecas saturina*) and one from India (*Nupserha vexator*). While all agents established, none have significantly reduced Noogoora burr infestations (van Klinken and Morin, 2012). Due to their limited impact, these agents will not be discussed further in this section (but see photos for identification below). During this time, a polyphagous stem-galling moth (*Epiblemma strenuna*) released against parthenium weed (*Parthenium hysterophorus*) was found to have some impact against the Noogoora burr complex (Dhileepan and McFadyen, 2012) and a rust (*Puccinia xanthii*) was recorded on Noogoora burr as a result of an unauthorised introduction (Alcorn, 1975).

P. Murray

Noogoora burr seed fly with distinctive wings.

M. Quinn

Noogoora burr stem-boring beetle from Northern America (Mecas saturina) affects plants in the field but cannot keep up with the compensatory growth of the plant.

Cerambycoidea Forum

Noogoora burr stem-boring beetle from India (Nupserha vexator) like its North American relative also does not overcome Noogoora burr's compensatory growth when attacked by this insect.

You may see verticillium wilt (*Alternaria zinnia*) and a form of powdery mildew (undetermined species) on the Noogoora burr complex. These fungi are not host specific and carry the risk of infecting several crops in Australia including cotton (Charles *et al.*, 2019).

K. Kirby

Verticillium wilt-affected Noogoora plants (left) and healthy cotton plants (right). This demonstrates the impact this disease can have on Noogoora plants, however note, the plants on the left were still able to produce burrs.

G. Charles

A Noogoora burr plant with its leaves covered in powdery mildew.

Stem-galling moth
Epiblema strenuana

The polyphagous stem-galling moth (*Epiblema strenuana*), released against parthenium weed (*Parthenium hysterophorus*) in 1982, has been found to damage young Noogoora burr plants, but with minimal impact to adult plants (Dhileepan and McFadyen, 2012). Larvae feed initially within the leaf and later within the growing shoot. This induces the stem to form an elongated gall. The stem-galling moth is widely established across Australia and reduces Noogoora burr plant vigour but provides limited control. Annual ragweed (*Ambrosia artemisiifolia*) is also attacked by this moth.

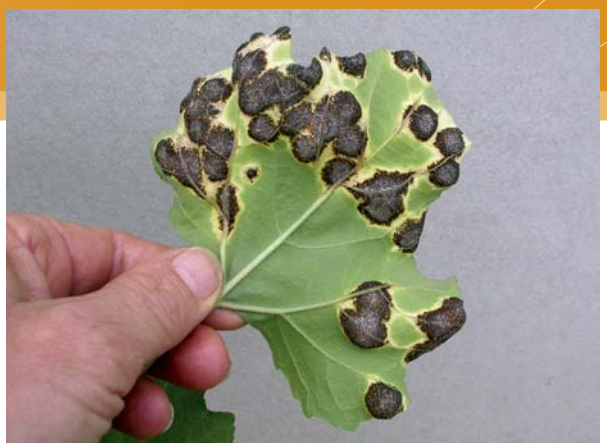
L.Morin

Noogoora burr stem-galling moth (Epiblema strenuana).

P. Sullivan

Noogoora burr stem-galling moth larvae weaken but do not kill Noogoora burr plants.

Noogoora burr



L. Morin

Noogoora burr rust symptoms on a mature Noogoora burr leaf.

Noogoora burr rust
Puccinia xanthii

The Noogoora burr rust, originally from America, was first detected in Australia in 1975. The rust spread quickly (both naturally and through human-mediated dispersal) and has successfully controlled Noogoora burr across large parts of its Australian distribution (van Klinken and Morin, 2012). All species of the Noogoora burr complex are highly susceptible to the rust, especially young plants which are severely impacted by the disease. The Noogoora burr rust has been less effective for plants growing in arid areas and the tropics where there are defined wet and dry seasons.

Identification

Noogoora burr rust fruiting bodies can be found on the lower leaf surface, as well as on stems and petioles (Morin *et al.*, 1992). The spore producing structures (sori) present as dark brown raised spots of up to 10 mm in diameter. When humidity is high, the sori contain many tiny white specks. When the spores germinate, the sori change to a white-grey. The Noogoora burr rust depletes plant resources, resulting in leaf drop of heavily infected leaves.

Life cycle

Puccinia xanthii is a microcyclic rust fungus, which means its life cycle is much simpler than many other rusts which have many life stages (Hanlin, 1994). Plants infected with the Noogoora burr rust show symptoms five days after inoculation in the form of dark brown sori (also known as telia), which are raised groups of teliospores that are not deciduous (i.e. remain attached to the plant when mature). Teliospores readily germinate under moist conditions. The rust fungus requires a dew period of 24 hours to maximise infection, with the ideal temperature for infection being 25°C (Morin *et al.*, 1993). Darkness stimulates teliospore germination (Morin *et al.*, 1992). Germinated teliospores produce small, hyaline (almost transparent) basidiospores that are dispersed by wind. Once basidiospores land on a susceptible host plant, they germinate and locally colonise *Xanthium* species plant tissue. Telia develop from these infection sites, so completing the life cycle. Several generations can occur per year, especially under humid, warm conditions. The spores overwinter on the dead leaves of Noogoora burr.

Field collecting and rearing

The Noogoora burr rust is widespread, and little is to be gained by redistributing it. Where plants are not infected, climatic conditions are likely to be suboptimal and too dry for it to thrive.

Monitoring establishment and dispersal

Look for distinctive 5 to10 mm diameter raised brown spots on the underside of leaves approximately one week after rain. Avoid monitoring when the maximum and minimum temperatures in the week post-rain have exceeded 40°C or were below 10°C. If present, report your sighting on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Paterson's curse
Echium plantagineum

Paterson's curse (also known as Salvation Jane) is an erect, coarsely haired, annual herb native to the Mediterranean Region (Sheppard and Smyth, 2012). It is present in all Australian states and territories. Plants grow up to 1.2 m in height and have trumpet-shaped, 2 to 3 cm long flowers that are usually bluish-purple, but can occasionally be pink or white. Large plants can produce up to 10,000 seeds (Sheppard and Smyth, 2012). These can germinate at any time through the year, but most germination occurs following the first and subsequent rainfall events in autumn and early winter. Plants grow as a rosette until spring, after which one or more flower-producing stems arise from the base. Paterson's curse thrives on a wide range of soils in warm temperate regions that have dominant winter rainfall. Plants usually die back in summer, but some plants persist for a second year (Sheppard and Smyth, 2012).

Paterson's curse was introduced to Australia as an ornamental plant and was first recorded growing at Camden, New South Wales in 1843 (Parsons and Cuthbertson, 2001). By 1918 it was considered a serious weed. It is a strong competitor in winter crops, pastures, roadsides and areas that are disturbed, degraded, neglected or fallow. In southern Australia it can dominate annual and perennial pastures from autumn until summer. Paterson's curse is poisonous causing cumulative liver damage that often leads to death, particularly with horses and other non-ruminant stock.

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Paterson's curse rosette. Note leaf shape and distinct branched veins.

J. Edwards

Paterson's curse flowers.

P. Sullivan

Infestation of Paterson's curse in a pasture situation.

From 1972, seven biocontrol agents from the Mediterranean region were introduced to Australia to test their potential against Paterson's curse (Sheppard and Smyth, 2012). Seven were approved for release with six, the leaf-mining moth (*Dialectica scalariella*), crown weevil (*Mogulones larvatus*), root weevil (*Mogulones geographicus*), flea beetle (*Longitarsus echii*), stem beetle (*Phytoecia coerulescens*) and pollen beetle (*Meligethes planisculus*) establishing in the field. These agents have reduced seed production, seed banks, plant density and plant vigour, and this has led to a significant reduction in the importance of Paterson's curse as a weed.

Note: The biocontrol agents for Paterson's curse also control the closely related weed known as viper's bugloss, *Echium vulgare*. Both weeds overlap in their distribution, with Paterson's curse being more abundant in warmer regions while viper's bugloss is more abundant in the cooler tableland districts.

P. Sullivan

Roadside population of viper's bugloss.

Recommendation

It is thought that the 'Portuguese strain' of the crown weevil and the flea beetle have the most impact on the two *Echium* spp., with the root weevil and the leaf-mining moth contributing less to control. The stem- and pollen-feeding beetles have minimal impact on the two weed species. All agents act synergistically for the effective control of *Echium* weeds throughout southern Australia with plants often dying before they flower due to their synergistic impact. Because of this, and to ensure best practice management of Paterson's curse, you first need to establish what biocontrol agents are present at your site. If only one species of agent is present, you may wish to introduce other agents to your site depending upon site conditions, or integrate biocontrol with other management practices (e.g. chemical, mechanical, grazing management).

Paterson's curse leaf-mining moth
Dialectica scalariella

The Paterson's curse leaf-mining moth, originally collected in the Mediterranean region, was first released in Australia in 1988 (Sheppard and Smyth, 2012). It is currently abundant and widespread, but it seldom reaches the high population levels needed for control, due to the limitation of food material over summer, low winter temperatures and high rates of parasitism. Larvae feed within the upper and lower leaf tissue forming mines in the leaves. Occasionally, with high levels of feeding, plant death occurs especially when plants are stressed (e.g. during drought).

Identification

The adult moths are approximately 5 mm long, thin and identified by their silvery-white and gold wing patterns. Moths can be found resting at an inclined, 45 degree angle to the leaf. As the adult moth can be easily confused with the Australian native species *Dialectica aemula*, identifying the biocontrol agent may be difficult. The cocoons of both species are, however, easily distinguishable by shape and colour. The inner (contains pupa) and outer cocoon of the Paterson's curse leaf-mining moth is white and similar in size, whereas the inner cocoon of the native has a yellow tinge, is elongate and narrow and relatively small compared to outer cocoon (Kumata and Horak 1997). Adults are non-feeding. Damage typically inflicted is by larval feeding where they tunnel within the leaf causing characteristic blister or blotch-like mine effects visible on the underside of the leaf's surface.

Life cycle

The leaf-mining moth has many generations per year, dependent on temperature, with the fastest generation occurring in two to three weeks in favourable warm conditions. On average 135 eggs are laid over a three-week period; usually on the lower side of leaves (Dodd and Woods, 1989).

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Paterson's curse leaf-mining moth.

J. Hosking

Paterson's curse leaf-mining moth damage.

If the plants are flowering, then eggs are also laid on the small leaves that grow on the stem itself. Newly hatched larvae tunnel into the middle of the leaf to feed and develop through five growth stages (instars). Larvae feed within the leaf forming serpentine, then blotch-like mines, before mature larvae (around 5 mm long) spin a doubled layered silken cocoon around themselves to pupate between the epidermal layers of the leaf. Emergence occurs after a few days in summer or several weeks in winter (Dodd and Woods, 1989).

Field collecting and rearing

Rearing is unnecessary. The moth is widespread throughout the distribution of Paterson's curse. As this moth has a low impact on the weed (compared to other agents), and because it is widespread throughout the distribution of Paterson's curse, field collecting for redistribution is not recommended.

Monitoring establishment and dispersal

Larvae are easy to detect throughout the year. In the early feeding stages, larvae form dark serpentine mines under the cuticle of the leaf, especially on the lower leaf surface. Blotch mines result from the feeding of older larvae and can be seen on the underside of leaves. Look also for the small white cocoons in leaves with old mines and adult moths (which can be observed sitting at an inclined, 45 degree angle to the leaves). If present, record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your monitoring guidelines (Appendix 3).

Paterson's curse crown weevil
Mogulones larvatus

The Paterson's curse crown weevil, originally from Europe, was first released in Australia in the early 1990s (Sheppard and Smyth, 2012). They were widely released across southern Australia and are currently abundant and widespread. Plants attacked by the crown weevil often die prior to flowering, which has resulted in a reduced Paterson's curse seed bank. Plants that have been attacked and survive are usually stunted and produce few seeds.

Crown weevils released in Australia were originally collected near Montpellier in France. They have a peak post-aestivation (summer or dry season dormancy) emergence in March. Many areas invaded by Paterson's curse in Australia are hot and dry and have unreliable autumnal rainfall which is problematic for the crown weevil's survival. If good autumn rains have not occurred by the beginning of April, when 90% of this 'French' strain of weevil has emerged from aestivation, there will be few germinated Paterson's curse rosettes for the weevils to feed on resulting in starvation and a population decrease. To address this, a second population of crown weevil (the 'Portuguese' strain) was collected from a hot dry area in Portugal and released in the early 2000s. Testing of the 'Portuguese' strain showed that it has a peak post-aestivation emergence in April which is five weeks later than that of the 'French' strain (P. Wilson, unpublished data). There is a greater likelihood of good autumn rains by early/mid-May when 90% of the 'Portuguese' strain would have emerged. This later post-aestivation emergence of the 'Portuguese' strain of crown weevil has enabled their populations to increase in those areas of Australia that are hotter, dryer or have unreliable autumn rainfall. Higher crown weevil populations have occurred across much of Australia since the 'Portuguese' strain was released.

Recommendation

To differentiate between plants impacted by the crown and root weevils, use the following signs:

- Plants infested with the crown weevil have shot hole damage on the leaves with blackened petioles and a dark necrotic discharge from the crown.
- Plants infested with root weevil have shot hole damage on the leaves without the blackened petioles and a dark necrotic discharge from the crown.

Identification

Adult crown weevils are 3.5 to 4 mm long and dark with light patterns along their sides and in the middle of their back. When they are disturbed, they tuck their legs under their body, resembling a bird dropping for camouflage. Adults create 'shot hole' feeding damage in the leaf tissue. The white-coloured larvae can be found feeding within the

Table 1. Emergence of *Mogulones* spp. from aestivation in Australia (P. Wilson, unpublished data)

Percentage emergence	*Mogulones larvatus* ('French' strain)	*Mogulones larvatus* ('Portuguese' strain)	*Mogulones geographicus*
50	Mid-March	Mid-April	Early-May
90	Early-April	Early/mid-May	Mid-May

plant crown, upper root, stems and petioles and are approximately three times longer than they are wide. It is hard to distinguish between crown and root weevil larvae; however, crown weevil larvae tend to feed higher in the plant in the petioles and plant crown than root weevil larvae which feed mainly in the root cortex.

S. Ivory, SARDI

Paterson's curse crown weevil.

A. McConnachie

Paterson's curse crown weevil feeding damage.

Life cycle

The crown weevil has one generation per year with adult females laying on average 450 eggs from autumn to spring after rains stimulate Paterson's curse germination (Sheppard and Smyth, 2012). After hatching, larvae feed inside the leaf petiole (stalk) and as they mature, they mine towards the root crown to feed. Attacked plants are easily identified through blackened petioles and a dark discharge from the crown. Larval feeding damages the meristem of the rosette, and one to two larvae alone can kill a rosette. Pupation occurs in the soil. Adults emerge from their pupal case in spring and feed on pollen and other plant parts to build up their fat reserves before moving into the soil and leaf litter where they become dormant to escape the high summer heat. Autumn rain stimulates their activity to coincide with plant growth. Only healthy adults survive this period of summer dormancy (aestivation). Because larval feeding occurs mainly above ground, populations of this agent are most effective under high rainfall and low grazing pressure situations.

Field collecting and rearing

Rearing is unnecessary. The crown weevil is widely distributed across the range invaded by Paterson's curse in Australia and generally does not require redistribution. However, should Paterson's curse populations be located where no signs of crown weevil damage can be detected, then adults can be collected from established sites in spring by targeting the flowers with a sweep net or beating tray (see Appendix 1 for techniques).

How and when to release

Release collected crown weevils directly onto healthy Paterson's curse plants as soon as possible. Select release plants with an overall diameter of >10 cm. Aim for a minimum of 500 adults per site. It is preferable to release in ungrazed areas. Record release information as per your weed biocontrol

release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Look for crown weevil damage from autumn through to spring by taking note of blackened petioles and a dark necrotic discharge from the crown. Plants with leaves showing circular to oval 'shot hole' damage (approximately 3 mm in diameter) are likely to have been fed on by adult crown and/or root weevils. Look for adult weevils under leaves or in debris near the root crown. Adults are normally seen from autumn through until late spring. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

P. Sullivan

Paterson's curse crown weevil damage impact on rosettes.

Paterson's curse root weevil
Mogulones geographicus

First released in Tarcutta, New South Wales in 1993, the Paterson's curse root weevil is the most common and damaging root herbivore found in its native European range (Sheppard and Smyth, 2012). As part of a national redistribution program, they were widely released across the range invaded by Paterson's curse in Australia where they have established widely and are now common. Although individual plant death has not been attributed solely to this weevil (unlike that observed through crown weevil impact), significant levels of plant damage are observed in the field.

Identification

Adult Paterson's curse root weevils are 4 to 5 mm long and mottled dark and light brown, with a fine cream-coloured patterning along their body and a distinctive curved snout. When disturbed, like the crown weevils, they tuck their legs under their body to camouflage themselves. Adults cause distinctive feeding scars on the leaf tissue or circular to oval 'shot hole' feeding damage (similar to the crown weevil). The white larvae can be found feeding within the roots of the plant and are approximately three times longer than they are wide. It is hard to distinguish between the Paterson's curse crown and root weevil larvae; however, the root weevil larvae tend to feed lower in the root of the plant (especially in the top 5 cm of the root).

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Paterson's curse root weevil.

P. Sullivan

Paterson's curse root weevil and leaf feeding scars.

Life cycle

In Europe, the Paterson's curse root weevil has one generation per year with adult females laying on average 250 eggs from autumn to spring after rains stimulate Paterson's curse germination (Sheppard and Smyth, 2012). It has a similar life cycle to the crown weevil, but a later post-summer dormancy emergence occurring in May that is advantageous during dry autumns (Table 1). After hatching, larvae feed in the lower root cortex and, as a result, are partially protected from damage through stock grazing. Pupation occurs in the soil. Adults emerge from their pupal case in spring and feed on pollen and other plant parts to build up their fat reserves before aestivating during the summer/early autumn period. Only healthy adults survive aestivation. Prior to reaching dense local populations, adults usually disperse from release sites (Sheppard and Smyth, 2012).

Field collecting and rearing

Rearing is unnecessary. The root weevil is widely distributed across the range invaded by Paterson's curse in Australia and generally does not require redistribution. However, should Paterson's curse populations be located where no signs of root weevil damage can be detected, then adults can be collected from established sites in spring by targeting the flowers with a sweep net or beating tray (see Appendix 1 for techniques).

How and when to release

Release collected root weevils onto healthy Paterson's curse plants as soon as possible. Select release plants with an overall diameter of more than 10 cm. Aim for a minimum of 500 adults per site. It is preferable to release in ungrazed areas. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Look for root weevil damage from autumn through to spring by looking for plants with feeding scars or circular to oval 'shot hole' feeding damage (approximately 3 mm in diameter). Look for adult root weevils under leaves or in debris near the rosette during autumn and spring. Identification of root weevil larvae can be achieved by digging up plants and examining the lower root cortex for wheat grain-sized larvae. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

Paterson's curse flea beetle
Longitarsus echii

First released in Australia in 1996, the Paterson's curse flea beetle from Europe has the highest establishment and plant attack rates out of all the Paterson's curse biocontrol agents (Sheppard and Smyth, 2012). A national redistribution program widely released the flea beetle across regions invaded by Paterson's curse in Australia. It has established widely and is now common. If enough larvae are present on a plant, the entire root system will be eaten, and the plant will die. The flea beetle provides excellent control with plant death often occurring prior to flowering. This is the most effective agent for areas with dry and unreliable rainfall and appears to be relatively less affected by grazing, compared to the two weevil species.

Identification

Adult flea beetles are approximately 3 mm long and are identified by their shiny, metallic-black/blue-green sheen. As their common name implies, they are called flea beetles due to their large hind legs which are adapted for jumping. Their modified hind legs give their overall body an arrow-head shape. Larvae are long and thin (six times longer than they

are wide) and white. Circular shot holes in leaves are the damage typically inflicted by adult flea beetles.

P. Sullivan

Adult Paterson's curse flea beetles and feeding shot holes.

Life cycle

In Europe, the flea beetle completes one generation per year, with adult females laying on average 250 eggs directly on the tap root in winter and spring after heavy rains stimulate Paterson's curse germination (Sheppard and Smyth, 2012). They share a similar life cycle to the crown and root weevils, with the exception that adult flea beetles emerge from aestivation mid-winter, whereas the weevil adults emerge from aestivation early to mid-autumn. Rainfall triggers adult emergence from their earthen chambers (up to 20 cm below ground), where-after they feed and then lay eggs. The thin larvae hatch after two to three weeks and feed on and mine the tap root and secondary roots and may even attack the underside of leaf petioles that are prostrate on the ground (Sheppard and Smyth, 2012). They tend to feed lower down in the root system than the root weevil larvae. When feeding is complete, the larvae exit the plant and pupate in the soil. Adult flea beetles aestivate over the summer period and emerge mid-winter, when it is more likely that Paterson's curse seeds will have germinated.

Recommendation

To differentiate between the Paterson's curse weevil larvae and the flea beetle larvae, use the following rule of thumb:

- Crown and root larvae are approximately three times longer than they are wide.
- Flea beetle larvae are approximately six times longer than they are wide.

P. Sullivan

Paterson's curse crown and root showing heavy attack from crown weevil (larvae are fatter) and flea beetle (larvae are thinner).

Field collecting and rearing

Rearing is unnecessary. The Paterson's curse flea beetle is widely distributed across the range invaded by Paterson's curse in Australia and should generally not require redistribution. However, should Paterson's curse populations be located where no signs of flea beetle damage can be detected, then adults can be collected from established sites in winter by using a sweep net (see Appendix 1 for technique) just above Paterson's curse rosettes. Collecting the flea beetles from early to mid-winter will result in capturing females that have laid fewer eggs and are more fertile. Greater numbers will be collected on sunny days

(preferably during the middle of the day when plants are dry).

How and when to release

Release collected flea beetles directly onto healthy plants as soon as possible. Aim for 250 adults per release. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Look for tell-tale signs in the form of 1 to 1.5 mm skeletonised, circular feeding 'shot' holes on leaves. Alternatively use a sweep net approach, or stealthily look for the adults feeding on the top of leaves (especially on sunny days). An Australian native copper-coloured flea beetle species can also be found feeding on Paterson's curse, so ensure that you do not confuse the two. If the Paterson's curse flea beetle is present, begin monitoring its dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

Paterson's curse stem beetle
Phytoecia coerulescens

The Paterson's curse stem beetle from Europe was released in Australia in 1995 and is widespread but uncommon (Sheppard and Smyth, 2012). This longicorn beetle feeds on the stems and upper roots of the plant, but it has never been observed in high numbers and its impact is minimal (Sheppard and Smyth, 2012). The damage occurs too late in the season to have an impact, except on thinner stemmed plants where the vascular tissues are destroyed, and the plants die.

Identification

Adult beetles are 15 to 25 mm long and are usually brown; however, this may vary from grey, to yellow green, to grass green or to steel blue. They have highly mobile antennae which are almost as long as

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Paterson's curse stem beetle.

their bodies (lying slightly above and alongside it). Larval feeding has a greater impact on plants than adult feeding, and can be observed by making a transverse cut through the mid- to lower stem and looking for evidence of tunnelling.

Life cycle

In Australia, and like its native range in Europe, the stem beetle has one generation per year (Sheppard and Smyth, 2012). Adult beetles emerge in spring and lay eggs into the lower section of the developing flower stems. The larvae then bore up the main plant stem before changing direction to bore into the main root. When ready to pupate, mature larvae construct a cocoon within the stem at ground level and remain dormant until the following spring when they pupate. The stem beetle survives well on thicker stemmed plants, so viper's bugloss is often a preferred host.

Field collecting and rearing

Rearing is unnecessary. The stem beetle is not recommended for redistribution. The stem beetle has never been found in high numbers, so collecting sufficient numbers for redistribution is not possible.

How and when to release

The agent is not available for release.

Monitoring establishment and dispersal

Look for the distinctive beetles on the larger plant stems during spring. If present, record your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Paterson's curse pollen beetle
Meligethes planisculus

First released in Australia in 1996, the Paterson's curse pollen beetle from Europe is common in cool, high rainfall areas (Sheppard and Smyth, 2012). To date, the pollen beetle has not reached population densities high enough to significantly limit the production of Paterson's curse seeds.

Identification

The adult beetles are 2 to 2.5 mm long and black. They have slightly forward-pointing, club-shaped antennae which extend out to the edge of the body. Adults feed mostly in open flowers. Larvae are small (up to 4 mm long) and white, and feed initially in flower buds and later on young flowers (Sheppard and Smyth, 2012).

Life cycle

The pollen beetle generally completes one generation per year, however, in Australia it can have a second generation if new adults emerge before day length starts to increase (Sheppard and Smyth, 2012). The adult beetles overwinter in leaf litter or soil. They become active in spring and start to feed, mate and lay eggs usually in the terminal, pre-flowering buds. Upon hatching, a larva will mine into the flower bud to feed on the anthers, pollen and immature seeds before feeding on the young flowers. When development is complete, the larvae drop to the soil to pupate. Adults emerge 10 days later and feed on pollen and the developing seed in open flowers.

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Adult Paterson's curse pollen beetle.

Field collecting and rearing

The pollen beetle is not recommended for redistribution. Even though it is currently established at 45% of release sites, it has not reached the high population densities needed to limit the seeding of Paterson's curse (Sheppard and Smyth, 2012).

How and when to release

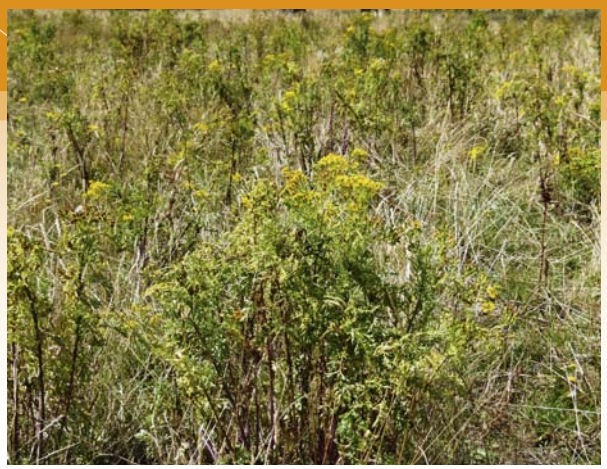
The agent is not available for release.

Monitoring establishment and dispersal

Look for the pollen beetle during the peak flowering period in spring by placing a beating tray underneath the flowers and tapping them. If present, report your sighting to your local weed or biosecurity officer and on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Ragwort
Jacobaea vulgaris

Ragwort is an erect, biennial or perennial herb native to Europe and western Asia (Ireson and McLaren, 2012). It has become a serious problem in parts of Victoria and Tasmania where annual rainfall exceeds 750 mm. Isolated infestations also occur throughout humid temperate regions of New South Wales, South Australia and Western Australia. Plants have deeply divided leaves (up to 35 cm long), single or multiple stems commonly 45 to 60 cm in height and clusters of bright yellow flowers (approximately 2.5 cm in diameter). Plants have numerous fleshy roots about 15 cm long and many fibrous roots that extend deeper into the soil.



P. Sullivan

Ragwort infestation.

Ragwort is a serious agricultural weed and is poisonous to most stock, causing cumulative chronic liver damage and fatality. Sheep, however, have a high tolerance to the alkaloids and are often used to control ragwort on infested properties (Parsons and Cuthbertson, 2001). Ragwort is not a strong

P. Sullivan

Ragwort flowers and foliage.

competitor and as such is not a problem in lush well managed pastures. For overgrazed or damaged pastures, it competes strongly once established to reduce pasture productivity and the value of agricultural land. Producing tens of thousands of seeds, combined with its capacity to regenerate from root or crown fragments, enables ragwort to rapidly spread.

Since 1930, Australia introduced seven species of insects from Europe to test their potential as biocontrol agents for ragwort. All seven were released but only five of these (including two flea beetles and three moths) have established. Numerous releases of the cinnabar moth, *Tyria jacobaeae* were made over a 64-year period starting in 1930, however, the moth has only survived at one site on the Mornington Peninsula in Victoria (Ireson and McLaren, 2012). As it is unlikely to become more widely established or play a significant role in ragwort control, provision of further information on this moth is considered unnecessary.

Recommendation

Effective control of ragwort is best achieved using conventional control methods combined with biocontrol and the presence of complementary species at your site. Except for the flea beetles whose dispersal you can accelerate, all other agents (plume moth, stem and crown-boring moth) are widespread and occupying areas that are climatically suitable. Therefore, redistribution is not recommended.

Ragwort flea beetle
Longitarsus flavicornis

The ragwort flea beetle, *Longitarsus flavicornis*, from France, was first released in Victoria and Tasmania in 1979 (Ireson and McLaren, 2012). It is common and widespread in ragwort infested areas of Tasmania, and its impact has been excellent, where ragwort densities have been reduced by up to 95% (Ireson *et al.*, 1991). Populations of the flea beetle have been restricted in areas where there is frequent pasture flooding and poor drainage, or in areas where incompatible management strategies are utilised. In Victoria, *L. flavicornis* has only established in high rainfall areas (above 500 m) and its dispersal has been slow.

Identification

Adult flea beetles are light brown and are approximately 3 mm long. They are called flea beetles because their large hind legs are adapted for jumping. Their whitish larvae grow up to 5 mm in length and have a head capsule that transitions from dark brown to reddish-brown. Adults feed on the rosette leaves producing small 'shot-holes'.

P. Sullivan

Ragwort flea beetle.

Life cycle

Ragwort flea beetles have only one generation per year. Adult flea beetles commence laying eggs in summer and laying can continue through autumn and winter, mainly in the soil around the base of plants. The larvae tunnel into leaf petioles and down the stem to feed on the root crown and roots from autumn until spring. The larvae pupate in the soil and emerge as adults in late spring.

Field collecting and redistribution

This agent does not need redistribution in Tasmania; however, there may be some sites in Victoria where successful establishment would be beneficial. A vacuum machine is the best way to collect this agent (see Ireson *et al.*, 2000 for technique). Collections can be made from early January until mid-April. Store agents only temporarily (at cool temperatures in a large insulated plastic box using an ice brick) and release insects directly onto healthy ragwort populations ideally within 24 hours of collection. Try to release a minimum of 300 ragwort flea beetles. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Within one year of release, look for the distinctive feeding 'shot-holes' in the leaves. The best time to look for adults is in autumn, preferably early in the morning, late in the afternoon or on cloudy days. Larvae can be found from August to October by pulling out plants and inspecting the root crown and roots for the small creamy white larvae. Record the presence or absence of the flea beetle as per your monitoring guidelines (Appendix 3). Monitor annually.

Ragwort flea beetle
Longitarsus jacobaeae

The flea beetle, *Longitarsus jacobaeae*, from Italy, was first released in Victoria in 1987 (Ireson and McLaren, 2012). This Italian biotype was considered to be better adapted to dryer and lower-altitude conditions than *L. flavicornis*. It has an aestivating adult stage over the hot summer period, with oviposition commencing in autumn to ensure the survival of eggs as opposed to the summer oviposition of *L. flavicornis*. Unexpectedly, its impact on ragwort in Victoria has been minimal and shows poor dispersal between sites. In Tasmania, its distribution overlaps well with that of *L. flavicornis*. Together they have reduced ragwort densities by up to 95%, although much of this impact has been attributed to *L. flavicornis* (Ireson *et al.*, 1991).

W. Chatterton

W. Chatterton

A ragwort site in Cradoc, Tasmania before (1987) (top) and after (1995) (bottom) introduction of ragwort flea beetle.

Identification

Longitarsus jacobaeae and *L. flavicornis* are very similar in appearance and it is not possible to separate the two species in the field.

Field collecting and monitoring suggestions

Collect, redistribute and monitor this species in the same way as *L. flavicornis*.

Ragwort stem and crown-boring moth *Cochylis atricapitana*

The ragwort stem and crown-boring moth, *Cochylis atricapitana*, from Spain, was released in Victoria in 1987 (Ireson and McLaren, 2012). It is well established in Victoria and Tasmania and continues to spread naturally through the invaded range of ragwort.

Identification

Adult moths are up to 10 mm long and are mottled cream in colour, with three darker bands (each approximately 1 mm wide) running across the middle and ends of the forewings. The larvae are

P. Sullivan

Damage caused by ragwort stem and crown-boring moth larvae.

Australian lepidoptera website

Ragwort stem and crown-boring moth adult.

creamy-white and grow up to 10 mm long and 2 mm wide. Larvae bore into the leaf and flower buds, crowns and stems causing significant plant damage, reducing their size and survival. Damage is characterised by blackened tissue on the young, central shoots, the root crown or the flower buds or compensatory multiple stem growth. In severely infested rosettes, larvae destroy the central crown killing the plant. Some plants survive and regrow, but with reduced foliage, and do not flower that season.

Life cycle

The ragwort stem and crown-boring moth is thought to have three generations per year in Victoria (McLaren, 1992) and two generations in Tasmania (Ireson and McLaren, 2012). Adult moths lay eggs on the underside of leaves along the leaf veins. After hatching, young larvae feed on the soft tissue in leaves and buds and older larvae mine into the stems and root crown. Larvae overwinter in old plant material and usually pupate inside the stems.

Redistribution

Ragwort stem and crown-boring moth is widespread, well established and does not require redistribution.

Monitoring establishment and dispersal

Larval damage is observed as blackened tissue on the young central shoots, the root crown or the flower buds. Plants that have been attacked by larvae often compensate by producing multiple stems. If larval damage is present, then examine the stems or root crown for the creamy-white coloured larvae. Alternatively look for moths at dawn and dusk from May to June or August. The ragwort stem and crown-boring moth should not be confused with the native, blue stem-borer moth, *Patagoniodes farinaria*, whose larvae have bluish-grey stripes along the length of their body. If the agent is present, report your sighting on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Ragwort plume moth
Platyptilia isodactyla

The ragwort plume moth, *Platyptilia isodactyla*, from Spain was first released in Victoria in 1999 (Ireson and McLaren, 2012). It is well established in Victoria and Tasmania and continues to spread unaided.

Identification

Adult ragwort plume moths have a body that is beige-coloured. Their light-brown wings have small dark-brown patches on them. The moth has a characteristic resting posture with the body and fully outstretched wings forming a 'T' shape. Adults are approximately 9 mm long with a wingspan of around 21 mm. Larvae are up to 12 mm long and transition from ivory to dark green as they mature. Larvae cause severe damage by tunnelling in the petioles, stems and root crowns, resulting in reduced plant vigour and reduced numbers of flowers and seeds (Ireson and McLaren, 2012).

W. Chatterton

Ragwort plume moth.

Life cycle

The ragwort plume moth has two generations per year. Adult moths lay an average of 100 eggs during their approximate 12-day lifespan. Eggs are usually laid on the underside of leaves. Newly hatched larvae mine down the petioles, through the stem and into the root crown. Pupation lasts approximately one week and usually takes place in the stem or root crown. Young larvae overwinter in ragwort plants (Ireson and McLaren, 2012).

Redistribution

Ragwort plume moth is widespread and well established throughout the range of ragwort and does not require redistribution.

Monitoring establishment and dispersal

Ragwort plume moth is nocturnal so look for the distinctive resting 'T' shape of the adult moth at night during spring and autumn. Alternatively look for larval feeding. Larvae eject their frass (waste products) from a small hole in the stem and this debris accumulates on silken webbing spun around the hole by the larva. If present, report your sighting on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Salvinia

Salvinia molesta

Salvinia is a free-floating, mat-forming, perennial fern (non-flowering) native to south-eastern Brazil (Julien, 2012a). It thrives on still or slow-moving, freshwater bodies in eastern and northern Australia. Infestations can double in area in under two weeks. Plants have light green, round to oval leaves (fronds), with three growth stages of development. Young leaves float flat on the water. In the second stage, leaves are slightly cupped and in the third stage the leaves are tightly folded and densely packed. The floating leaves appear as opposite pairs and are covered in waxy hairs. A third leaf is modified, submerged and more root-like. Salvinia grows up to 2 cm in height from the water surface with rhizomes extending up to 30 cm. Reproduction is solely by vegetative growth. Daughter plants form from broken rhizome pieces bearing buds. In Australia it appears that the spore sacs are either empty or sterile (Julien, 2012a).

Introduced as an ornamental plant for fishponds and aquariums in the 1950s, salvinia quickly became problematic in aquatic ecosystems and was widespread throughout the tropical and subtropical regions of Australia by the 1970s (Room and Julien, 1995). Dense floating mats limit sunlight, reduce available oxygen, and create anaerobic conditions that are destructive to all other aquatic life. In 1999 salvinia was listed as a Weed of National Significance due to its invasiveness and negative impacts on the environment and human activity.

Australia introduced three species of insects from South America to test their potential as biocontrol agents for salvinia. Two agents (the weevil, *Cyrtobagous salviniae*, and the moth *Samea multiplicalis*), were released with only the weevil proving to be effective against salvinia (Julien, 2012a).

P. Sullivan

Salvinia plants with leaves covered in waxy hairs or trichomes.

R. Coventry

R. Coventry

Before salvinia control (top) on the Hawkesbury river New South Wales and after control by the salvinia weevil (bottom).

Recommendation

Biocontrol efforts should focus solely on using the weevil *Cyrtobagous salviniae*. Thick mats of salvinia may need to be thinned, either mechanically or with herbicide, prior to biocontrol. The salvinia weevil is an excellent agent that can effectively control salvinia in tropical, subtropical and warmer temperate areas within one to three years at some sites. In cooler regions, it can take several years to achieve weevil establishment and even longer for control. Site specific characteristics (including temperature, shade, nutrient concentration, waterbody size and type) influence the level of weevil establishment and control. Repeated releases may be required. It is best to release salvinia weevils in spring. Keep an eye on your site through continual monitoring and re-introduce more weevils when necessary.

Salvinia weevil
Cyrtobagous salviniae

The salvinia weevil (*Cyrtobagous salviniae*) from Brazil provides excellent control. First released at Lake Moondarra, Mount Isa Queensland in 1980, it reduced 50,000 tonnes of salvinia to less than a tonne within 15 months (Room *et al.*, 1981). Since then, the weevil has been widely redistributed throughout coastal and sub-coastal eastern Australia and the Northern Territory with control occurring in under three years in warm tropical sites. Redistribution is often required because of site specific characteristics and poor dispersal by the weevil.

Identification

Adult weevils are approximately 2 mm long and black (Julien, 2012a). Newly emerged adults are light brown then darken to turn black within about five days. Adult weevils damage leaves and buds. Damaged leaves have distinguishing shot holes, and the buds turn brown and rot. The thin cream-coloured larvae (approximately 4 mm long) are more damaging than adults as they feed internally within rhizomes and buds, which deforms and stunts salvinia growth.

P. Sullivan

Adult salvinia weevil.

L. Postle, NSW DPI

Salvinia weevil larva.

P. Sullivan, NSW DPI

Browning by weevil feeding.

P. Sullivan, NSW DPI

Weevil feeding holes.

Life cycle

The salvinia weevil has multiple generations per year; the number is dependent on temperature and food availability. Weevils can live for about six months. Adult females lay more than 300 single eggs within the leaves and rhizomes (Room, 1990). Eggs hatch after approximately 10 days. The newly emerged larvae feed on the outer leaf surface initially before tunnelling into the plant to feed within rhizomes and buds. Larvae complete their development over five to seven instars. Pupation takes place in a cocoon attached to the roots about 2 cm below the water surface.

Field collecting and rearing

Rearing salvinia weevils is time consuming but productive if a continuous supply of weevils is needed for your site. Mass-rearing centres may have a readily available supply of the salvinia weevil, so check with your local weed or biosecurity officer before collecting the agent from known release sites.

The weevil is generally active all year round in the sub-tropics and tropics. However, it is best to collect from November through to March when it is most active. Contact your local weed or biosecurity officer for suitable sites from which to collect the weevil. To collect the agent, first look for signs that it is present including adult weevils crawling over the salvinia or brown plants. To collect adults, submerge infected salvinia under wire mesh that has holes small enough (approximately 1 cm^2 is suitable) to submerge the salvinia but large enough to allow the weevils to pass through and float to the surface. The weevils will continue to float to the surface for up to two days. Allow a few, small, floating salvinia plants to act as an attractant for the weevils so that they can be easily collected using forceps or small nets (see Appendix 1 for further details). Ideally, a minimum of 200 adults is required for your release site.

T. Brown

Submerging salvinia to collect the weevil.

Recommendation

Be careful when working with biocontrol agents and salvinia, as plants can easily break into pieces and create new infestations downstream.

Traditionally, releases of the salvinia weevil were conducted by releasing plant material infested with larvae. However, while larvae are most damaging, it is preferable to collect and release only adult weevils to minimise the spread of contaminated material to new areas (see page 10 on practising good hygiene).

Prior to redistribution, adults can be stored temporarily (at cool temperatures using an ice brick) in sealed containers containing some leaf material. Cover either with a lid with small air holes or insect mesh for ventilation (i.e. for a few days at around 15°C).

How and when to release

Ideally release between 200 and 500 weevils (more is better) in full sun directly onto healthy plants from spring to summer. To assist with nursery site establishment, release weevils in a protected part of the salvinia mat, e.g. a small bay away from the main waterway channel. This gives the weevils an opportunity to establish large populations without being washed downstream. Record release information as per your Weed Biocontrol Release Form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for weevil presence by examining the plants for bud damage. Damaged leaves will turn brown or black from weevils feeding at the nursery site within one year of release. The extent of bud damage correlates well with the adult weevil population size and salvinia decline. The more damage you see indicates that the weevil population may be succeeding in controlling salvinia. Alternatively, adult weevils can be counted on plants (they are often found hiding around the bud area). If weevils are present, begin monitoring dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor regularly over summer to see if the weevils are actively feeding and if the salvinia is gradually turning brown. If there is insufficient change, more weevils may be required.

Scotch broom

Cytisus scoparius var. *scoparius*

Scotch broom (also known as English broom) is an erect, perennial shrub native to Europe (Parsons and Cuthbertson, 2001). It is estimated to infest in excess of 230,000 ha throughout Australia. Scotch broom is a thicket-forming, woody shrub up to 4 m in height which has brownish-green, ridged stems. Broom plants have leaves comprised of three leaflets (that often fall off during summer), bright-yellow, pea-shaped flowers and brown-black, flattened seedpods. In recent years, two red- and yellow-flowered hybrids known as *Cytisus scoparius* cultivar 'Andreanus' and *Cytisus scoparius* cultivar 'Andreanus aureus' have also established field populations (Hosking *et al.*, 2012). In 2012 Scotch broom was listed as a Weed of National Significance due to its invasiveness, environmental and economic impacts and potential to spread.

Scotch broom was originally introduced to New South Wales as an ornamental species in the early 1800s (Parsons and Cuthbertson, 2001). By the late 1800s it was recognised as a serious weed. It invades native bushland, watercourse margins, roadsides, neglected areas and cattle grazing properties in cool climate, high rainfall areas of south-eastern Australia.

P. Sullivan

Scotch broom flowers.

P. Sullivan

Scotch broom infestation.

Scotch broom can form dense thickets that prevents the re-establishment of native seedlings. Seed dispersal occurs through movement in soil and water, and on machinery, footwear, stock and wildlife. Pods burst open (dehiscence) and shoot seeds several metres away. This facilitates the rapid thickening of infestations and drives spread, especially along water courses. Scotch broom seeds have a hard coat that can delay germination for months or years, allowing large seed banks to develop. Seed can remain viable in the soil for many years, often germinating in spring and autumn of years following fire or soil disturbance.

From 1993, three insects and a mite from Europe were introduced to Australia to test their potential against Scotch broom (Hosking *et al.*, 2012). These included a twig mining moth (*Leucoptera spartifoliella*), a psyllid (*Artainilla spartiophila*), a bruchid beetle (*Bruchidius villosus*), and mite (*Aceria genistae*). All agents were released and have established causing some damage to Scotch broom in parts of southern New South Wales, Victoria and Tasmania.

Scotch broom gall mite
Aceria genistae

The Scotch broom gall mite, from Europe, was released in south-eastern Australia from 2008 to 2010 (Hosking *et al.*, 2012). Scotch broom gall mites infest dormant vegetative buds in autumn causing them to become galls in spring. Galls drain Scotch broom's reserves. This results in the development of fewer shoots, flowers and seeds, and when heavily attacked shrub death can occur after several years (e.g. six to seven years in Tasmania). Galls also provide shelter and protect mites from being fed on by predatory mites. The efficacy of Scotch broom gall mite populations is reduced in colder areas.

Identification

Adult Scotch broom gall mites are so small that it is extremely hard to see them with the naked eye. They are translucent, creamy-white to pale orange and cigar-shaped. The best way to detect them is to look for the 5 to 30 mm diameter, furry, whitish galls that form as a result of their feeding.

Life cycle

The Scotch broom gall mite has multiple generations per year. Eggs are laid initially on or inside buds and, if present, within existing galls. Young Scotch broom gall mites develop and feed within the gall. In late summer and autumn, the galls may start to whither and when this happens the mites migrate to new stem buds for the winter. Scotch broom gall mites are also wind dispersed.

Recommendation

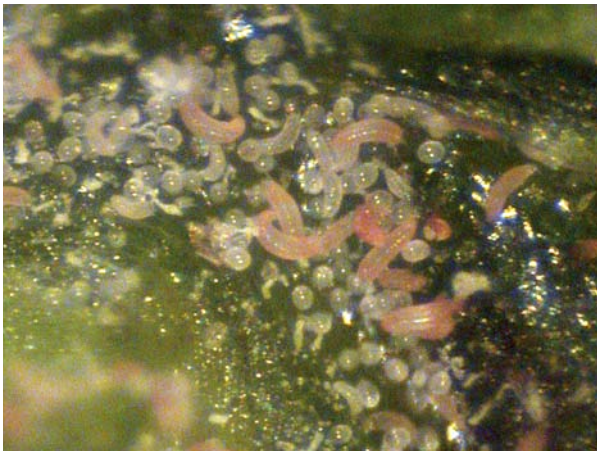
Scotch broom gall mite currently offers the best biocontrol solution for Scotch broom. Under favourable conditions shrubs may die due to the gall mites' impact. The efficacy of the mite may be climatically limited in cooler areas.

P. Sullivan

Furry whitish gall caused by the Scotch broom gall mite.

P. Sullivan

Large gall caused by the Scotch broom gall mite.



W. Chatterton

Scotch broom gall mite adults (orange), larvae and eggs.

P Sullivan

Scotch broom plant heavily attacked by gall mite.

Field collecting and rearing

Scotch broom gall mites can be easily redistributed by transferring cuttings (in late autumn or early spring) of gall-covered stems to uninfected shrubs (refer to Appendix 1 for technique). Cuttings should be about 20 cm long and contain at least three, fresh, green galls per cutting. Store cuttings temporarily in a cool insulated box with ventilation to prevent foliage from drying out or insects over-heating (i.e. cool temperatures of around 15°C). Do not refrigerate. Redistribute cuttings as soon as possible.

P Sullivan

Distributing Scotch broom gall mite to a new plant via a cutting.

How and when to release

Attach one mite-infested cutting per healthy Scotch broom bush. Gather 6 to 12 stems containing fresh buds from the release bush, and using these surround the galled cutting. Secure this arrangement using a piece of tie wire. The tie wire can be left in place because the mites will migrate from the drying gall onto new buds, begin feeding and start gall formation. The formation of galls may take up to a year before they can be seen. Tie high visibility tape near the wrapped stems to assist with future monitoring. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Wait at least one year from the transfer of cuttings to examine the plants for establishment of Scotch broom mite, which is indicated by the presence of fuzzy, whitish galls. Record the presence or absence of the mite as per your monitoring guidelines (Appendix 3). Monitor annually.

Other agents that have been released

- Scotch broom twig mining moth (*Leucoptera spartifoliella*) was first released in New South Wales in 1993 and shortly thereafter in Victoria, South Australia and Tasmania (Hosking *et al.*, 2012). It has had minimal impact on Scotch broom, possibly due to parasitism.
- Scotch broom psyllid (*Artainilla spartiophila*) was first released from 1994 in New South Wales, Victoria and South Australia but its establishment was poor, and it has not been seen since 2010 (Hosking *et al.*, 2012).
- Scotch broom seed bruchid (*Bruchidius villosus*), a beetle, was released in New Soufth Wales and Victoria from 1995 to 1998 and established at several sites (Hosking *et al.*, 2012). Its impact in Australia is not known, however, seed predation levels of up to 84% have been recorded in New Zealand.

Tasmanian Institute of Agriculture

Scotch broom twig mining moth.

© D. Ouvrard/Psyl'list/NHM-London

Scotch broom psyllid.

U. Schmidt

Scotch broom seed bruchid.

St John's wort

Hypericum perforatum

St John's wort is a toxic, perennial herb native to Europe (Briese and Cullen, 2012). It was introduced to Australia in the mid to late nineteenth century and has since invaded over 400,000 ha, predominately in south-eastern Australia but also in parts of South Australia and Western Australia. It has two growth forms; in autumn and winter it grows as a prostrate rosette, while in spring and summer, it produces erect, woody, flowering stems (Campbell *et al.*, 1995). Rosettes with an overall diameter of 60 cm, have a central woody crown, with soft, spindly stems and a dense mat of bright, elongated leaves. A stout, main root is associated with each rosette which can grow to a depth of one metre. Rhizomes occur just below the soil surface from which aerial growth develops each year. Flowering stems (up to 1.2 m tall), have opposite leaves (1.5 to 3 cm long) and golden-yellow flowers (approximately 2 cm in diameter) borne in terminal clusters. Plants can produce up to 30,000 seeds annually. Seeds can remain viable in the soil for 20 years (Briese and Cullen, 2012). Several varieties of St John's wort exist in Australia; the more common narrow-leaf variety, a broad-leaf variety and several varieties with a mix of these characteristics.

Introduced to Australia for ornamental and medicinal purposes during the mid to late 1800s (Harris and Gill, 1997), St John's wort, by 1917, was recognised for its negative impact on the grazing industry and to the environment. As a strong competitor, St John's wort readily invades poorly managed grazing land, open woodland, roadsides and neglected areas in humid and sub-humid temperate regions throughout the year; especially drier sites at elevations between 500 and 1000 m. Poisonous to livestock, St John's wort causes photosensitisation, depression, loss of condition, abortion, infection and resultant death (Bourke, 1997).

R.G. Richardson

St John's wort flowers and foliage.

J. Hosking

St John's wort rosette producing new plants from lateral roots.

St John's wort

From 1928, 15 biocontrol agents from Europe were studied to test their potential against St John's wort (Briese and Cullen, 2012). Eleven were approved for release in Australia, with six, the leaf-feeding beetles (*Chrysolina quadrigemina* and *Chrysolina hyperici*), eriophyid mite (*Aculus hyperici*), root-feeding beetle (*Agrilus hyperici*), phloem-feeding aphid (*Aphis chloris*) and gall-forming fly (*Zeuxidiplosis giardia*), establishing in the field. Of these, the two chrysomelid beetles and the mite provide a degree of intermittent control at a localised level because the strong root reserves of St John's wort enable fast regrowth. Difficult to control, St John's wort requires a persistent integrated management program.

Recommendation

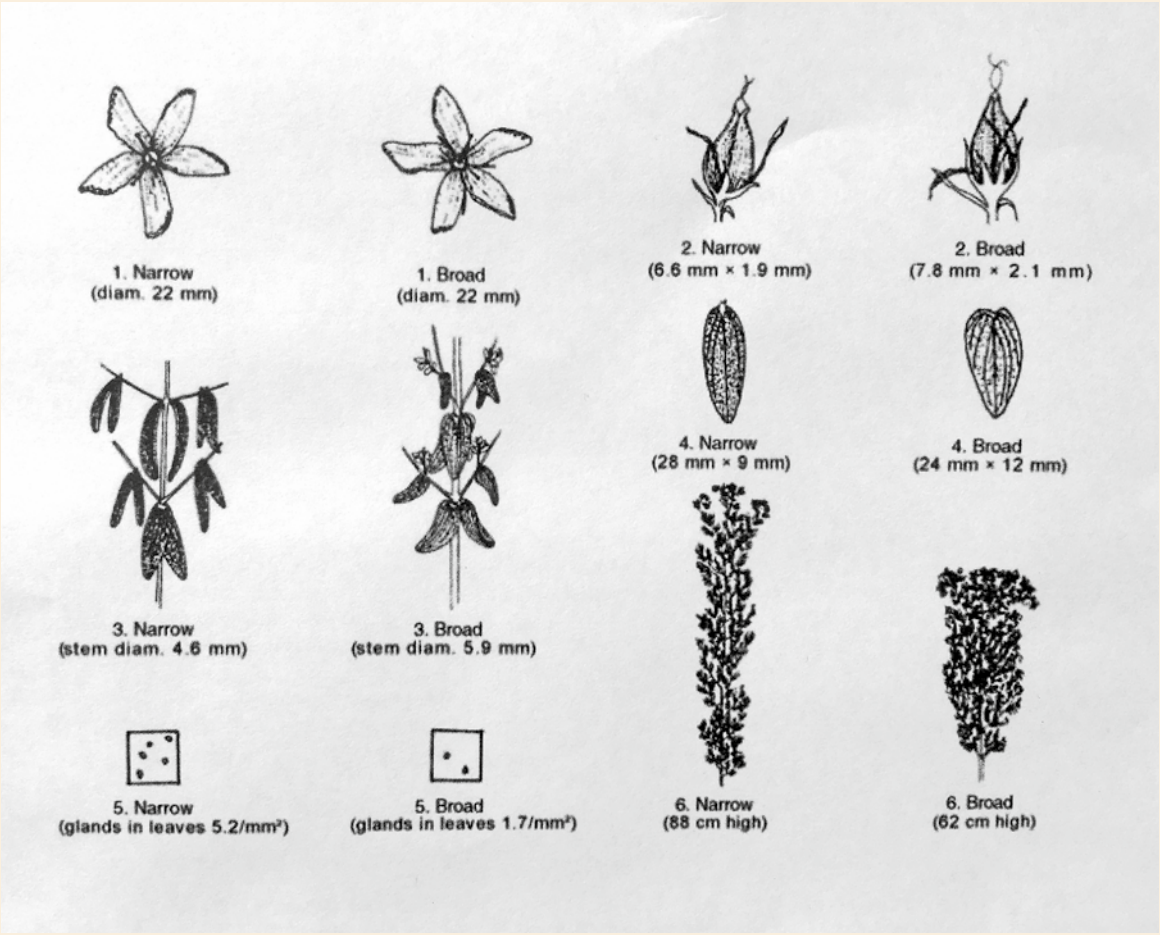
Use an integrated approach to manage St John's wort, including biocontrol, good pasture management and judicious chemical control.

M. Campbell

Invasion of St John's wort in a pasture situation.

As there are two different varieties of St John's wort which can influence your biocontrol program or wider integrated management program, it is important to differentiate between them. For example, while the chrysomelid beetles are effective against both forms of St John's wort, the St John's wort mite is only effective against the narrow-leaf variety.

To differentiate between the narrow- and broad-leaf varieties of St John's wort, measure the leaves at the sixth node (bump) on the flowering stem when the plant is growing well in spring. The narrow-leaf variety has leaves 7 to 9 mm wide whereas the broad-leaf strain has leaves 10 to 12 mm wide.



J. Peacock

Illustration comparing parts of narrow-leaf and broad-leaf varieties of St John's wort (downloaded from NSW WeedWise website https://weeds.dpi.nsw.gov.au/Weeds/Details/135).

St John's wort chrysomelid beetles *Chrysolina quadrigemina* and *C. hyperici*

The St John's wort chrysomelid beetles *C. quadrigemina* and *C. hyperici* are discussed concurrently as they share similar biologies, behaviour, modes of action and occasionally occur in mixed species populations (Briese and Cullen, 2012).

Chrysolina quadrigemina from England was first released in south-eastern Australia from 1930 to 1937. In parallel, *C. hyperici* (also from England) was released from 1930 to 1934 (Wilson and Campbell, 1943). Both species established, however, *C. quadrigemina* populations started to outcompete those of *C. hyperici* since the former had earlier autumnal activity. To rectify this imbalance, new populations of both species were re-collected from summer rainfall regions of France and released from 1980 to 1981 (Briese and Cullen, 2012). Unfortunately, no change in effectiveness was observed. Both species are active in spring when they sweep through areas invaded by St John's wort, feeding on the flowering stems. This gives temporary relief, but unfortunately only a few of the defoliated plants will die and most plants will regenerate following good rainfall or in the following year. Usually several years will lapse (between two and 10 years) before the beetles occur in the same area again.

Identification

Chrysolina quadrigemina adults are 6 to 7 mm long and have a metallic blue, green, purple or brown appearance. *Chrysolina hyperici* is slightly smaller at 5 to 6 mm long and is metallic bronze in colour. The larvae of both species are orange in their early stages, and then turn a pinkish-grey as they mature (Briese and Cullen, 2012).

Adult feeding damage is characterised by defoliation of the flower stems during spring, while larval feeding damage is notable as defoliation of the rosettes during winter (*C. quadrigemina*) and spring (*C. hyperici*).

A. McConnachie

The St John's wort beetles.

H. Egon

Chrysolina hyperici.

Life cycle

The St John's wort chrysomelid beetles have one generation per year (Briese and Cullen, 2012). The adults of both species undergo a summer resting period (aestivation) in the plant leaf litter. In autumn, females resume activity to feed and lay eggs (*C. quadrigemina* does so several weeks earlier than *C. hyperici*). More than 1000 eggs can be deposited by each female, either individually or in small clusters, on the underside of leaves or on rosette leaf buds during autumn (Briese and Cullen, 2012). *Chrysolina quadrigemina* eggs hatch within three weeks, while those of *C. hyperici* overwinter only to hatch in spring. The young, orange-coloured larvae transition to

pinkish-grey as they mature. They feed at night and rest concealed in the leaf litter beneath the plant during the day. Mature larvae pupate in globular soil cells (5 cm below the surface) and after two to three weeks, adults emerge in mid to late spring before undergoing aestivation to escape summer heat.

Field collecting and rearing

Rearing is unnecessary. The beetles are widely distributed across the range invaded by St John's wort in Australia and generally do not require redistribution. However, due to the boom/bust outbreaks of the beetles they may have a seasonal occurrence at your site. Should St John's wort populations be located where no signs of beetle damage can be detected, then adults can be collected from established sites in spring by targeting the flowers with a sweep net or beating tray (see Appendix 1 for techniques).

How and when to release

Release collected beetles directly onto healthy St John's wort plants as soon as possible. Select release plants in full sun, with high population densities. Aim for ideally 500 adults per site. Release in ungrazed areas so that adults and larvae are not trampled. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring suggestions

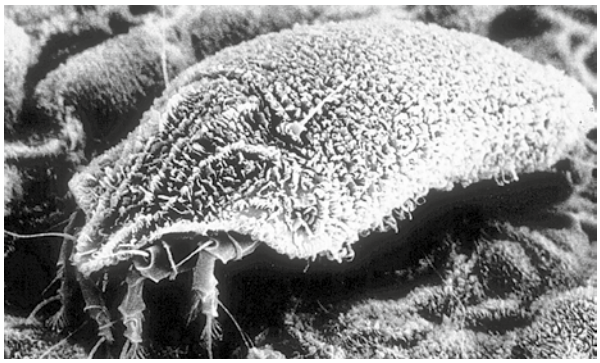
Look for adult beetles or defoliated, reddish flower stems during spring. Alternatively, inspect rosettes for defoliated feeding damage or larvae within the leaf litter in winter for *C. quadrigemina* or during spring for *C. hyperici*. Keep in mind that *C. quadrigemina* usually moves through a site along a feeding front in large numbers, and then may be absent from an area for several years. If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Monitor annually.

St John's wort mite
Aculus hyperici

First released in 1990 in south-eastern Australia, the St John's wort mite, from southern France (Briese and Cullen, 2012) is now widely established following a national distribution program (Jupp, 1996). Mites deplete root reserves over a two to three-year period by feeding on cells and sap in the growing points of St John's wort. As a result, shoots become dwarfed, and the distance between shoots on the stems becomes reduced resulting in plant stunting and death. With time, plants become less competitive, with infected sites showing a decline in plant density. Some varieties of St John's wort have been found to be resistant to mite attack, with the narrow-leaf form appearing to be most susceptible (Naughton and Bourke, 2007).

Identification

Adult female St John's wort mites are invisible to the naked eye (approximately 0.15 mm long and 0.05 mm wide) (Briese and Cullen, 2012). They are cream-coloured and have a soft, flattened, unsegmented body with two pairs of legs. Nymphs look like the adults, only smaller. Mite damage is characterised by a stunting and twisting of leaves and stems with a dust-coated appearance.



CSIRO

Adult St John's wort mite.

Life cycle

The St John's wort mite has multiple generations per year (Briese and Cullen 2012). Females lay up to 40 eggs each in the soft leaf tissue of the growing points. The eggs are minute, having a diameter of only 0.05 mm. After hatching, the mite nymphs pass through two nymphal instars (growth stages) before transitioning to adulthood. All life stages are present through the year. The mite is wind dispersed and new colonies have been found to disperse up to 50 km within one year (unpublished data).

Field collecting and rearing

The St John's wort mite is difficult to rear, but reasonably easy to redistribute via transfer of infected shoots. To do this look for signs of damage on shoots indicating mite activity. Collect at least 15 (more is better) shoot cuttings (around 30 cm long) containing mites in spring or autumn when conditions are mild. Prior to redistribution, cuttings containing mites can be stored temporarily (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at around 15°C).

How and when to release

Attach several mite infested stems to healthy narrow-leaf plants at your release site. Use a tie wire to secure infested cuttings to healthy plants in an upright position against a star picket to prevent the plant collapsing. The mites will move across to the new host as the infected leaf dries out. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring suggestions

Look for mite activity by observing stunted leaves and stems in narrow-leaf populations of St John's wort the following spring and autumn. Confirmation of mite establishment can be carried out under a microscope or hand lens. Record the presence and absence of the mite as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Other agents that have established

Several other agents have been released and established in Australia as part of the St John's wort biocontrol program. Their usefulness as effective biocontrol solutions are limited for various reasons, and are briefly detailed below (Briese and Cullen, 2012):

- The root beetle, *Agrilus hypericin* from southern France, was first released in 1939 (with supplemental introductions in 1984 and 1989 in New South Wales and South Australia). It is currently only thought to persist at one isolated locality near Tuena, New South Wales.
- The aphid, *Aphis chloris*, from southern France was released in 1986 and is relatively widespread through the invaded range, although not common. Populations appear to go through seasonal cycles of growth and dispersal but are often not high enough to provide adequate impact (Briese and Jupp, 1995).
- The gall fly, *Zeuxidiplosis giardi*, from France, was first released in 1953 and is widespread, though not very common. The high numbers required to cause significant damage have not been observed in the field.

Thistles

There are several introduced thistle-type genera occurring in Australia, with some of the most problematic species occurring within the genera *Carduus, Onopordum* and *Cirsium*. All comprise of species originating from the Mediterranean, Europe, Asia Minor and North Africa. Biocontrol has been a highly successful integrated management tool used in managing thistles in Australia, of which biocontrol programs against eight of these species including, nodding thistle (*Carduus nutans*), slender thistle (*Carduus pycnocephalus*), winged slender thistle (*Carduus tenuiflorus*), spear thistle (*Cirsium vulgare*), Scotch thistle (*Onopordum acanthium*), Illyrian thistle (*Onopordium illyricum*), and stemless thistle (*Onopordum acaulon*) are described within this section.

Nodding thistle
Carduus nutans

Nodding thistle is an erect (up to 120 cm in height), annual or biennial, spiny herb native to Eurasia and North Africa (Parsons and Cuthbertson, 2001). Two subspecies occur in Australia but only *Carduus nutans* subsp. *nutans* is problematic, especially in New South Wales where it invades open areas of the tableland districts. It is also present in small areas of the Australian Capital Territory, Victoria and Tasmania. Plants have deeply divided, grey-green leaves, large, pink, drooping flower heads (approximately 8 cm in diameter) surrounded by purplish spiny bracts and a deep taproot. Prolific seed production occurs as plants can germinate throughout the year with adequate moisture (Popay and Medd, 1995).

Introduced into Australia, likely as a seed contaminant from New Zealand in the 1940s (Parsons and Cuthbertson, 2001), nodding thistle was soon recognised as a serious pastoral weed in New South Wales. Its aggressive, competitive nature creates dense patches which discourage grazing by livestock.

NSW DPI

Nodding thistle flower head.

NSW DPI

Nodding thistle infestation.

Difficult to control, nodding thistle requires a persistent integrated management program.

Three species of insects from Europe were introduced to test their potential as biocontrol agents for nodding thistle. All three agents, including the receptacle weevil (*Rhinocyllus conicus*), seed fly (*Urophora solstitialis*) and rosette weevil (*Trichosirocalus horridus*), have been released and demonstrate considerable success in reducing seed production, seed banks and plant density (Cullen and Sheppard, 2012).

Recommendation

All biocontrol agents released against nodding thistle are widespread and redistribution is generally not necessary. However, if nodding thistle seed fly and rosette weevil are not present at your site, you can accelerate dispersal by redistributing these from a well-established site. Combined, they coexist well in the field and their activity is complementary or synergistic against nodding thistle.

Nodding thistle receptacle weevil
Rhinocyllus conicus

First released between 1988 and 1990 throughout the tablelands of New South Wales, the receptacle weevil, from three different regions within Europe and later imported from New Zealand, is now widespread throughout the invasive range of nodding thistle in Australia (Cullen and Sheppard, 2012). High larval populations occur early in the flowering season, destroying most of the early-season seed but are less effective later in the season. A maximum reduction of 36% in seasonal seed productivity has been observed (Woodburn and Cullen, 1993), which is insufficient in controlling nodding thistle due to high mid and late season seed productivity. As such, other agents are required to complement the receptable weevil's activity.

Different ecotypes of the receptacle weevil develop on four thistle genera (*Carduus*, *Cirsium*, *Silybum* and *Onopordum*). Although, the nodding thistle receptacle weevil prefers nodding thistle (*Carduus nutans*) they will attack other thistles to varying degrees including spear thistle (*Cirsium arvense*) as described below.

Identification

Adult weevils grow up to 7 mm long and are dark brown to black with a yellowish mottled coat of hairs and long rostrum (snout). Inconsequential damage by adults can be identified by small round holes in the leaves and/or small brown scars. The larvae are white and feed inside the receptacle forming a gall-like callus causing much greater damage than adult feeding. Keep in mind that this callus can easily be confused with the galls developed by seed fly larvae (Groenteman, 2008a).

P. Sullivan

Receptacle weevil.

Life cycle

The receptacle weevil usually has one generation each year but can have a second generation when very early emerging adults produce eggs in the second half of the flowering season. Adult females deposit up to 200 eggs on developing flower heads once buds have appeared on thistles during spring (>20 eggs can be found on a single flower bud). After hatching, larvae tunnel into the flower head, and feed on the receptacle for about four weeks before pupating within the receptacle. Within a few weeks, newly emerged adults feed briefly on leaves before undergoing aestivation (summer dormancy)

in the soil until the next spring. Adults can live up to 15 months (Groenteman, 2008a).

Field collecting and rearing

Rearing is unnecessary. Nodding thistle receptacle weevil is widespread throughout the distribution of nodding thistle in Australia and does not require redistribution.

Monitoring establishment and dispersal

Look for adult receptacle weevils on the flower heads in spring and early summer. While larvae and pupae can be found within thistle flower heads from early summer through to late winter they can be easily misidentified as larvae and pupae of the seed fly. If present, record your sighting on the Australian Biocontrol Hub and begin monitoring agent dispersal as per your monitoring guidelines (Appendix 3). Monitor annually.

Nodding thistle seed fly
Urophora solstitialis

First released in 1991 throughout the tablelands of New South Wales and later redistributed, nodding thistle seed fly from Europe is now widespread throughout the invasive range of nodding thistle in Australia. A single larva can destroy one seed and prevent development of a further six (Sheppard *et al.*, 1994). Additionally, gall formation exhausts the plant nutrient reserves and reduces the plant's overall vigour. The existence of a second generation of flies in the latter half of the flowering season helps to reduce seed production in nodding thistle after larval activity of the receptacle weevil has ceased (Cullen and Sheppard, 2012).

Identification

Adult seed flies grow between 5 and 8 mm long, and are black with distinctive black stripes on their long and clear wings. Larvae are barrel-shaped and creamy-white with a black posterior. Damage by

P. Sullivan

Nodding thistle seed fly.

seed fly larvae is distinguished by gall formation. As larvae mine through the florets their feeding makes the plant produce gall tissue around each larva, which in time can fuse together to form a larger gall containing as many as 25 insects (Groenteman, 2008b).

Life cycle

The seed fly has one generation per year and a partial second generation occurs where larvae that develop early in the season go on to produce a second generation later in the season. During spring, adult females lay up to 100 eggs singly into developing flower heads. After hatching, the larvae feed inside the flower head inducing the plant to divert its nutrients intended for seed production into gall formation around the larvae. Mature larvae go on to pupate within the galled flower head. New adults emerge to begin a second generation, providing suitable flower heads are available. These second-generation larvae overwinter in the galls and emerge

Seed flies can be adversely affected by nodding thistle receptacle weevils in the early part of the flowering season. This is because their food source for larval development may be limited due to severe flower attack by the receptacle weevil. Populations of seed fly therefore tend to build up and provide greater impact against nodding thistle when larvae of the receptacle weevil are scarce, usually in the mid-to-late flowering season.

as new adults the following spring (Cullen and Sheppard, 2012).

Field collecting and rearing

Rearing is unnecessary. The seed fly is widespread and should not need redistribution. However, should your site show no evidence of seed fly damage, you can accelerate dispersal by collecting at least 20 (preferably more) galled thistle heads during winter and redistributing them as soon as possible to a new nursery site. Ensure that you do not deplete your collection site of infested gall flower heads. Infested thistle heads can be stored in containers with ventilation temporarily prior to redistribution (i.e. a few days at cool temperatures of around 15°C).

How and when to release

Twenty or more galled thistle heads should be enclosed in a small, fine chicken wire cage during winter and early spring (large enough to get flies out in spring but not too large so that the flower heads fall). The cage will help to protect galls from being damaged by livestock until adult flies emerge. Cages should be suspended approximately one metre off the ground (e.g. attached to a star picket) and near actively growing nodding thistles. Avoid full sun as larvae can be easily killed. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for seed fly activity within one year of release. Adult seed flies can readily be observed on the flower heads throughout the flowering season, especially in summer and autumn. Larvae and pupae can be found from summer until winter inside multi-chambered galls within thistle heads. Keep in mind that they can easily be mistaken for larvae and pupae of the receptacle weevil. Record agent establishment and dispersal for each nursery site as per your monitoring guidelines (Appendix 3). This should be repeated yearly in spring or early summer.

Nodding thistle rosette weevil
Trichosirocalus horridus

First released in 1993 throughout the tablelands of New South Wales and later redistributed, the nodding thistle rosette weevil (from an existing biocontrol program against nodding thistle in New Zealand) is now widely established across the invasive range of nodding thistle in Australia (Cullen and Sheppard, 2012). Larval feeding damages the central growth point of the rosette stimulating the formation of sub-crowns and, if these are attacked, the formation of additional sub-crowns which reduce the vigour of the plant, and sometimes leads to plant death. Larval feeding also stimulates the production of softer spineless rosette leaves that are more readily grazed by stock. A reduction of nodding thistle seed production was estimated at 72% by the rosette weevils' impact, making this the most effective agent established for control of nodding thistle (Cullen and Sheppard, 2012).

Identification

Adult rosette weevils are 3 to 4 mm long and transition from reddish-brown to dark brown as they age. Larvae are barrel-shaped and creamy-white. Adult feeding on leaves causes distinctive 'shot holes'. Larvae either mine down the petiole to the meristem,

L. Kok

Nodding thistle rosette weevil.

or crawl to the crown, where they initially feed on the unexpanded leaves surrounding the meristem before moving on to feed on the meristem itself and leaves that surround it. As larvae feed, they exude a black, tar-like substance to the surface. The main veins of the surrounding leaves take on a reddish colour as a result of their feeding.

Life cycle

Nodding thistle rosette weevil has one generation per year. Adult females lay up to 800 eggs singly on the lower side of rosette leaves and in the pockets of the leaf's midribs and petioles (leaf stalk) from autumn through to spring. After hatching, (which can take a week to a month depending upon temperature), the larvae exit the plant and move down and within the rosette (crown) to feed before pupating in late winter to early spring. Adults emerge during spring to feed extensively on plant leaves for several weeks before moving into the soil and leaf litter where they hibernate to escape the high summer heat until early autumn (Woodburn,1997; Cullen and Sheppard, 2012).

Field collecting and rearing

Rearing is challenging and unnecessary. The rosette weevil is widespread and should not need redistribution. However, dispersal of the weevil can take a long time. Should your site show no evidence of rosette weevil damage, you can accelerate dispersal by collecting adult weevils from well-established sites in autumn and winter and moving them to new areas. You can easily collect weevils using an aspirator (see Appendix 1 for technique). Rosette weevils can be temporarily stored in containers with ventilation prior to redistribution (i.e. a few days at cool temperatures of around 15°C).

How and when to release

Release, ideally, at least 300 adults directly onto healthy thistles at the new nursery site in autumn as soon as possible after collection. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for rosette weevil activity in winter and early spring within one year of release. Look for leaves with a mined midrib or those that have a reddish colour at their base. Also look for the black discharge from the crown and the tell-tale physiological changes to the rosettes such as multiple sub-crowns and or softer spineless rosette leaves. Alternatively, look on the rosettes for adult weevils or 'shot holes' during autumn and spring. Record the presence or absence of the weevil as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the provided guidelines. Monitor annually in spring or early summer.

Slender thistles
Carduus pycnocephalus and *Carduus tenuiflorus*

The two closely related species, slender thistle (*Carduus pycnocephalus*) and winged slender thistle (*Carduus tenuiflorus*), are discussed concurrently, with differences specified where required. Both are erect and slender annual herbs (up to 2 m in height) native to Eurasia and northern Africa (Parsons and Cuthbertson, 2001). In Australia they have a similar distribution. They readily invade improved pastures and neglected areas as well as natural ecosystems of subhumid, and warm-temperate regions especially in areas with winter dominant rainfall (>500 mm per annum). Both species have single or multiple stems that are strongly ribbed with spiny wings. In winged slender thistle, the wings extend up to the base of the flower head, whereas slender thistle usually does not have wings up to the base of the flower head. Slender thistle has green stems but winged slender thistle often have red or purple stems. Further, both plants have pink or purple flowers, approximately 1 cm in diameter that are borne in clusters of three to eight flowers (winged slender thistle) or three to four flowers (slender thistle).

Both thistles were introduced to Australia over 100 years ago from Europe, the details of how are unknown. Recognised as naturalised in Victoria in the 1880s the slender thistles now occur in much of eastern and southern Australia up into south-eastern Queensland (Parsons and Cuthbertson, 2001). Both species are recognised as contaminants of wool with infestations significantly reducing pastoral productivity. Difficult to control, slender thistles require a persistent integrated management program.

Introduced over 50 years ago, an isolate of the rust fungus *Puccinia cardui-pycnocephali* was tested and found to be ineffective in reducing seed productivity of slender thistles in Australia. However, two strains of the rust from Europe were identified as promising for thistle control and were introduced to test their potential as biocontrol agents for slender thistles. Both strains established well and are now widespread across the invasive range of slender thistles in Australia (Chaboudez *et al.*, 1993; Groves and Sheppard, 2012).

R.G. Richardson

Slender thistle.

R. Holtkamp

Winged slender thistle.

The nodding thistle receptacle weevil, *Rhinocyllus conicus,* has been observed on flower heads of slender thistle but its impact as a biocontrol agent is not known. Monitor for its presence on slender thistles and report sightings to the Australian Biocontrol Hub.

P. Sullivan

Nodding thistle receptacle weevil.

Slender thistle rust fungus
Puccinia cardui-pycnocephali

First released in 1993, two strains of the rust fungus from Mediterranean Europe are capable of significantly reducing plant growth and seed productivity in slender thistles across southern Australia (Burdon *et al.*, 2000). Although a strain of the rust fungus was already widespread in Australia, the two strains from the Mediterranean were shown to be more aggressive than the Australian strain which provided little impact on slender thistle infestations (Chaboudez *et al.*, 1993). These two strains of rust fungus affect the two species of slender thistles differently. The strain from Italy is more infectious on slender thistle (*C. pynocephalus*) whereas the strain from France is more infectious on winged slender thistle (*C. tenuiflorus*). Heavy rust infection can cause the leaves and stems to dry up which leads to a significant reduction in plant growth and seed productivity. Both strains are widespread throughout the invaded range of slender thistles in Australia; and control appears to be more pronounced in wetter summers. The rust does not require redistribution (Groves and Sheppard, 2012).

Identification

Symptoms of slender thistle rust fungus can be seen predominately in autumn and spring, coinciding with new plant growth. Infection occurs on the lower leaf surface and on flowering stems and presents as brown necrotic spots or pustules. Sporing bodies of the rust often develop next to the leaf veins and a yellow ring can develop around the active sporing body where leaf tissue has been damaged (Faithfull *et al.*, 1998).

Life cycle

Several generations of the rust can occur each year under optimal conditions. Rust infections start on new seedlings from autumn and build up over spring. To infect the plant, spores enter the leaf through open stomata (breathing pores) on the leaf's underside. Within two to three weeks, tens of thousands of new spores are produced, which are easily transported by wind and water to infect new plants or sites of infection (Faithfull *et al.*, 1998).

J. Kruse

Slender thistle rust fungus.

Field collecting and redistribution

Both rust strains are widespread and unlikely to need redistribution. However, use the 'spore water' method in late autumn or spring if redistribution is warranted for your site (see Appendix 1 for spore water technique). Keep in mind that control is more effective in wet summers. Where plants are not infected, conditions may be suboptimal and too dry for the rust to thrive. Speak to your local weed or biosecurity officer to assist you in making this assessment.

Monitoring establishment and dispersal

Look for the rust from autumn through to spring. Leaves and stems will appear to be withered and have rust pustules (particularly on the leaf undersides). If present, report your sighting on the Australian Biocontrol Hub. Monitor for its presence annually as per your guidelines (Appendix 3).

Spear thistle
Cirsium vulgare

Spear thistle is an annual, biennial or short-lived spiny perennial herb (up to 1.5 m tall) native to Eurasia and North Africa (Parsons and Cuthbertson, 2001). Plants first form a basal rosette of leaves, before becoming upright and erect (up to 1.5 m tall), with a deep taproot and much branched spiny stems with a covering of woolly white hairs. Leaves are dark green (up to 35 cm long) and become deeply lobed and armed with spines along their margins with age. Flowering by the purple and globular flower heads (3 to 5 cm in diameter and enclosed in numerous spiny bracts) occurs year-round but is most common from spring through to autumn. The flattened seeds are grey or light brown and topped with a ring of feathery bristles to aid in dispersal. Up to 200 flowering heads and 8000 seeds can be produced by a single plant, with most producing around 100 seeds on average.

First recorded in Tasmania in the 1830s, and recognised by the 1850s as a serious weed of pasture and cereal crops throughout the southern states of Australia (Parsons and Cuthbertson, 2001), today few habitats in temperate Australia are free of spear thistle. Unpalatable and spiny, dense infestations of spear thistle impede stock movement and grazing, are a devaluing component in wool, and are a major weed in rice rotations. Difficult to control, spear thistle requires a persistent integrated management program.

Three insect species were introduced from Europe to test their potential as biocontrol agents for spear thistle. All three agents, the receptacle weevil (*Rhinocyllus conicus*), the gall fly (*Urophora stylata*) and the rosette weevil (*Trichosirocalus horridus*) were released and have established (Sagliocco *et al.*, 2012).

NSW DPI

Spear thistle flower head.

P. Sullivan

Spear thistle infestation.

Recommendation

Information pertaining to the extended distribution and impact of biocontrol agents against spear thistle are largely unknown. Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting biocontrol agents for redistribution purposes.

Spear thistle receptacle weevil
Rhinocyllus conicus

First released in 1990 in Victoria, the receptacle weevil (also released on nodding thistle) from western France did not persist on spear thistle and was re-released in 1994. Although it has now established in isolated areas of Victoria, information pertaining to its extended distribution and impact are largely unknown. Although adults can fly, the weevil can take a long time to disperse to new areas (Sagliocco *et al.*, 2012). Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting for redistribution purposes.

See nodding thistle section on page 142 for details on the receptacle weevil's biology.

Field collecting and rearing

Rearing is unnecessary. Redistribution from sites where receptacle weevils are well-established is possible but will need to be sourced from Victoria. Adult weevils can be sourced during spring by using the beating method, an aspirator (see Appendix 1 for technique) or even a modified garden leaf vacuum machine whereby the insect is not sucked through but collected using a bag in the mouth of the vacuum. Repeat this method across many plants so that enough numbers can be collected, ideally between ten and twenty adults or more where possible. Collect on warm sunny days when insects are most active. Prior to redistribution, weevils can be stored temporarily with a small amount of foliage (at cool temperatures) in sealed containers with small air holes for ventilation (i.e. for a few days at cool temperatures of around 15°C).

How and when to release

It is integral that thistles at your release site have plenty of developing flower buds for adults to deposit their eggs and for larvae to feed. To achieve good establishment, it is best to place insects into a fine mesh cage or tent erected over a dense patch of thistles. After two weeks remove the cage to allow the adults to disperse to other thistles at your release site. However, if a cage is unavailable insects may be released without one. Simply empty the container of weevils onto healthy spear thistles with plenty of developing flower buds. Spread the insects evenly around a number of plants in close proximity so one plant is not overloaded but the insects are close enough to find each other again for mating purposes. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your weed or biosecurity officer.

Monitoring establishment and dispersal

Look for adult receptacle weevils on flower heads in spring and early summer within a year of release. Keep in mind that while larvae and pupae can be found within the flower heads from early summer through to late winter they can be easily misidentified as larvae and pupae of the seed fly. Record the presence and absence of the weevil as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Spear thistle gall fly
Urophora stylata

First released in Victoria in 1993, the spear thistle gall fly, sourced from western France, was later redistributed to New South Wales and Tasmania, and thereafter, naturally dispersed to South Australia. It has firmly established, but impact is variable (Sagliocco *et al.*, 2012). While up to a 32% reduction in seed per flower head has been observed, it is insufficient to reduce plant population density (Winston *et al.*, 2014). As a result, other agents are required to complement this fly's activity, along with other control methods.

Identification

Adult spear thistle gall flies are small (around 5 mm long), with brown and yellow bodies and clear wings with striking dark bands that extend beyond their body. Larvae are small, barrel-shaped and creamy-white with a black posterior. Larval feeding within the flower head, induces plant swelling to develop a hard pea to marble-sized gall, that is initially green turning brown with maturity (Bruzzese *et al.*, 1998).

Life cycle

The spear thistle gall fly has one generation per year and a partial second generation, when early developing larvae go on to produce a second generation later in the season. Adult females lay their eggs between the bracts of developing flower buds or between the florets in spring. After hatching, single or multiple larvae move down the florets and feed on developing seed to create a swelling made of hard tissue called a gall. Here they overwinter as larvae before they pupate within and emerge in spring. Providing suitable flower heads are available, new adults can emerge mid-summer to begin a partial second generation. This second generation of larvae overwinter in the galls, emerging as new adults the following spring (Bruzzese *et al.*, 1998).

Field collecting and rearing

Rearing is unnecessary. Collect at least twenty (preferably more) galled thistle heads during winter and early spring and redistribute them as soon as possible to a new nursery site. Ensure that you do not deplete your collection site of infested gall flower heads. Prior to redistribution, infested thistle heads can be stored in containers with ventilation temporarily (i.e. a few days at cool temperatures of around 15°C).

How and when to release

To protect galls from livestock damage until adult flies emerge, twenty or more galled thistle heads are best enclosed in a small, fine chicken wire cage

S. Ivory, SARDI

Spear thistle gall fly.

P. Sullivan

Spear thistle gall fly larva.

during winter and early spring (large enough to let flies out in spring but not too large so that the flower heads fall). Suspend your cage approximately 1 m in height off the ground (e.g. attached to a star picket) and near actively growing spear thistles. Avoid full sun as larvae can be easily killed. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for spear thistle gall fly activity within one year of release. Adult flies can readily be observed on flower heads throughout the flowering season, in spring and especially in summer. Alternatively, during winter you can also look for the presence of the spear thistle gall fly by looking for galls. To confirm this, wear thick gloves and press the dry thistle heads gently between your thumb and fingers to feel for the pea to marble-sized hard gall. Keep in mind larvae of the gall fly can easily be mistaken for larvae and pupae of the receptacle weevil. Record agent establishment and dispersal for each nursery site as per your monitoring guidelines (Appendix 3). Monitor annually.

Spear thistle rosette weevil
Trichosirocalus horridus

First released in 1996 in eastern Australia, the spear thistle rosette weevil was imported from an existing biocontrol program against nodding thistle in New Zealand. In New Zealand they recognised that the weevil was able to establish on spear thistle in the absence of nodding thistle populations. In Australia, adult and larval feeding are expected to stunt plant growth and delay flowering. While established, information pertaining to its extended distribution and impact are largely unknown (Sagliocco *et al.*, 2012). Although adults can fly, the weevil can take a long time to disperse to new areas. Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting for redistribution purposes.

See nodding thistle section on page 144 for details on the rosette weevil's biology.

Field collecting and rearing

Rearing is challenging and unnecessary. Redistribution from sites where rosette weevils are well-established may be possible if found during autumn and winter. You can collect weevils using an aspirator (see Appendix 1 for technique), by inspecting rosettes for the presence of adult weevils, or using a modified garden leaf vacuum machine whereby the insect is not sucked through but collected using a bag in the mouth of the vacuum. Prior to redistribution, rosette weevils can be stored temporarily in containers with ventilation (i.e. a few days at cool temperatures of around 15°C).

How and when to release

Ideally release at least 300 adults directly onto flowering healthy plants in autumn at the new nursery site as soon as possible after collection. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for rosette weevil activity in autumn and spring within one year of release. Look on the rosettes for adult weevils or signs of adult feeding that create 'shot-holes' in the leaves during autumn and spring. Record agent establishment and dispersal for each nursery site as per your monitoring guidelines (Appendix 3). Monitor annually.

Thistles

Onopordum thistles

Onopordum thistles are annual, biennial or occasionally perennial herbs native to Eurasia (Parsons and Cuthbertson, 2001). Their taxonomy is complex owing to the existence of intermediate forms resulting from hybridisation. Species include the two closely related species, Scotch thistle (*Onopordum acanthium*) and Illyrian thistle (*Onopordum Illyricum*) and their hybrids (referred to as the Scotch/Illyrian thistle complex) and stemless thistle (*Onopordum acaulon*). All are covered in dense, woolly hairs that give them a whitish to bluey green appearance with Scotch thistle being more silver in appearance. Leaves and winged stems are toothed with each ending in a stout rigid spine. Large purple flowers are all surrounded by numerous spined bracts that are often purple. Scotch and Illyrian thistles have single winged erect stems (up to 3 m tall) and are major weeds of pastures, crops, disturbed sites, waste areas, gardens and roadsides in mostly temperate regions where annual rainfall is usually greater than 500 mm. Stemless thistles are prostrate and occur in warm-temperate regions where rainfall is usually less than 450 mm. They commonly occur in pastures, along roadsides, and in neglected areas.

Likely introduced as ornamental plants in the mid-1800s, *Onopordum* thistles are all serious agricultural and environmental invaders. Recognised as naturalised throughout much of south-eastern Australia, they outcompete pastures, are largely unpalatable to stock and as such restrict movement and stock carrying capacity. Prolific seeders, once established they are difficult to control.

Australia introduced eight species of insects from Eurasia to test their potential as biocontrol agents for *Onopordum* thistles. Seven of these were released, with four including the seed-head weevil (*Larinus latus*), the stem-boring weevil (*Lixus cardui*), the crown weevil (*Trichosirocalus briesei*) and the crown moth (*Eublemma amoena*) establishing in the field (Briese 2012b). All agents are effective on *Onopordum* thistles except for the stem weevil which does not survive on stemless thistles.

P. Sullivan

Scotch thistle flowers and foliage.

P. Sullivan

Scotch thistle infestation.

Recommendation

All agents impact *Onopordum* thistles except for the stem-boring weevil (*Lixus cardui*), which does not survive on stemless thistle. Impact by biocontrol agents on stemless thistles is largely unknown. Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting and redistribution purposes.

NSW DPI

Illyrian thistle flowers and foliage.

NSW DPI

Illyrian thistle infestation.

J. Dellow

Stemless thistle.

Scotch thistle seed-head weevil
Larinus latus

First released in 1992 and later widely redistributed for Scotch/Illyrian thistles complex, the Scotch thistle seed-head weevil from Europe is now abundant and widespread. A single larva can destroy all the seed within a flower head and subsequently, seed productivity has been found to be reduced at some sites by more than 80%. Other agents complement the seed head weevil's activity, leading to substantial control of the long-lived seedbank (Briese 2012b).

Identification

Adult seed-head weevils are large (up to 25 mm long), with a large characteristic rostrum (snout) typical of all weevils. They transition from yellowish, green-brown to black as they mature. Larvae are white and develop internally within the flower heads where they feed on receptacle tissue and developing seeds. Adult feeding on stems leads to scaring and 'shot holes' (up to 4 mm in diameter) appear on leaves. Damage caused by larval feeding is difficult to assess as they remain inside the flower head until adult emergence following pupation (Briese, 2012b).

Life cycle

Seed-head weevils have one generation per year. Adult females lay up to 70 eggs in their lifetime with each laid singly in the upper stem or flower head and protected by faecal matter. Females lay their eggs from late spring through to early summer or as long as the flowers stay healthy. After hatching, larvae tunnel into the flower head where they feed for up to six weeks on surrounding tissue and developing seed before pupating within the flower head and emerging, usually in late summer. They seek protected overwintering sites until mid to late spring when they become active and mate (Pettit and Briese, 2000; Briese, 2012b).

Field collecting and rearing

The seed-head weevil is widespread and should not need redistribution. However, dispersal of the weevil can take a long time. Should your site show no evidence of seed-head weevil damage, you can accelerate dispersal by collecting adult weevils from well-established sites in spring and moving them to new areas. You can easily collect the weevils using the beating method (see Appendix 1 for technique). You could potentially use a modified garden leaf vacuum machine whereby the insect is not sucked through but collected using a bag in the mouth of the vacuum. Prior to redistribution, seed-head weevils can be stored temporarily in containers with ventilation (i.e. a few days at cool temperatures of around 15°C).

R. Holtkamp P. Sullivan

Scotch thistle seed-head weevil adult (left), larvae (right).

How and when to release

Ideally release between 60 and 150 seed-head weevil adults directly onto flowering plants at the new nursery site as soon as possible and before the end of spring. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for seed-head weevil activity when plants are flowering (spring and summer) and within one year of release. Adults are easy to see on flower heads. Alternatively, if plants are not flowering, adult feeding may be detected by the shot holes they create in leaves. Pupae can be observed by breaking open flower heads in mid to late summer. Record the presence and absence of the weevil as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Onopordum stem-boring weevil
Lixus cardui

First released in 1993 in New South Wales and Victoria, and later redistributed to Tasmania, the stem-boring weevil from southern France is now the most widely established of the *Onopordum* biocontrol agents in south-eastern Australia (Breise, 2012b). Heavy attack by both adult and larval feeding reduces plant vigour and seed productivity. Weakening the plant, the stem-boring weevil substantially makes way for the seed-head weevil to later complement attack and provide substantial control of the Scotch/Illyrian thistle complex (Swirepik *et al.*, 2008).

Identification

Adult stem-boring weevils are elongate and thin in appearance (up to 15 mm long and 5 mm wide). With yellow mottling along their backs, they transition from a green-brown appearance to black as they mature. Larvae are white with a brown head capsule. Adults feed voraciously on the plant to create 'shot holes' in leaves (up to 8 mm in diameter) or complete defoliation when abundant. Larvae bore into stems where they feed on structural tissue (Briese, 2012b).

P. Sullivan

Onopordum stem-boring weevil.

P. Sullivan

Onopordum stem-boring weevil feeding damage.

Life cycle

Stem-boring weevils have one generation per year. As soon as thistles start to produce flowering stems, adult females lay their eggs within the developing stems. They protect their eggs with a visible plug of plant frass (waste products). After hatching, larvae bore deep within plant stems, and feed on structural tissue. Up to 100 larvae can develop within a single stem which substantially weakens the plants. After pupating within stems, new adults remain in dead stems over summer and winter before emerging in spring (Briese, 2012b).

Field collecting and rearing

Rearing is unnecessary. The stem-boring weevil is widespread and should not need redistribution. However, should your site show no evidence of stem-boring weevil damage, you can accelerate dispersal by collecting adult weevils from well-established sites in autumn and winter when they are hibernating within the dead stems of thistles. Identify and collect stems with bore holes, as these contain adult weevils. Collect as many stems as possible to maximise the number of adults for release. Store these stems in containers with ventilation (at cool temperatures of around 15°C) for redistribution in late winter.

How and when to release

Attach several weevil infested stems to healthy thistles in late winter. Each stem may contain around 30 to 40 adults. Open a few stems to check for healthy adult activity, being careful not to damage adult weevils. Ideally release 100 to 200 adults at a site. Using a tie wire, secure infested stems to healthy thistles in an upright position against a star picket to prevent them collapsing. Potentially protect them from livestock grazing by releasing agents in fenced off areas. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for stem-boring weevil activity during the autumn and winter when stems are dead and showing signs of stem boring weevil activity through the presence of many holes. Alternatively, look for signs of damage appearing as shot holes or complete plant defoliation and adult activity during late spring and summer and within one year of release. Record the presence or absence of the weevil as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the provided guidelines. Monitor annually.

Scotch thistle crown weevil
Trichosirocalus briesei

First released in 1997 in New South Wales, Scotch thistle crown weevil, sourced from northern Spain, was later redistributed to a limited number of sites in south-eastern Australia. While established, it is isolated to parts of the tableland's districts within New South Wales where its impact is localised. Larvae can kill small rosettes and reduce the size and vigour of large plants (Briese, 2012b). Information pertaining to its extended distribution and impact however are largely unknown. Although adults can fly, the weevil can take a long time to disperse to new areas. Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting for redistribution purposes.

Identification

Adult crown weevils are small (between 3 and 5 mm long), mottled dark brown with a characteristic long rostrum (snout). Larvae are white and develop internally within the rosette crown. Adult damage is characterised by chew holes in the rosette leaves. Damage by larvae feeding within the rosette crown and surrounding petioles causes a black discharge visible at the crowns surface (Briese, 2012b).

CSIRO

Scotch thistle crown weevil.

Life cycle

Crown weevils have one generation per year. Adult females lay several hundred eggs from autumn to spring in the midribs on the undersides of rosette leaves. After hatching, larvae develop over three growth stages (instars) by tunneling down the midrib to the crown of the plant where they feed on the base of the petiole (leaf stalk) and on plant tissue. When mature, larvae migrate to the soil where they pupate and emerge in late spring briefly to feed on foliage before undergoing aestivation (summer dormancy) until the following autumn (Briese *et al.*, 2002; Briese, 2012b).

Field collecting and rearing

Rearing is challenging and unnecessary. Should your site show no evidence of crown weevil damage, you can accelerate dispersal by collecting adult weevils from well-established sites in autumn and moving them to new areas. You can easily collect the weevils using an aspirator (see Appendix 1 for technique) where much time may be spent inspecting rosettes for the presence of adult weevils, or use a modified garden leaf vacuum machine whereby the insect is not sucked through but collected using a bag in the mouth of the vacuum. Prior to redistribution, crown weevils can be stored temporarily in containers with ventilation (i.e. a few days at cool temperatures of around 15°C).

How and when to release

Ideally release at least 100 adult crown weevils directly onto healthy rosettes at the new nursery site as soon as possible after collecting in autumn. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for crown weevil activity during winter to early spring within a year of release. This may be evidenced by feeding holes in leaves, mining presence in the

leaf midrib or a black discharge from the crown. Additionally, adults can be found externally on the rosettes from autumn through to spring. Record the presence or absence of the weevil as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines. Monitor annually.

Scotch thistle crown moth
Eublemma amoena

First released in 1998 in New South Wales, the crown moth, sourced from southern France, is established but restricted in its current range to the southern tablelands and south-western slopes of New South Wales (Breise, 2012b). Impact is localised and largely unknown, however larval feeding can kill small rosettes and reduce the size, vigour and seed production of larger plants. The summer generation is the quickest to develop and subsequently has the largest impact over the season. Impact is complemented by the crown weevil. Before the major onset of flowering, the crown weevil can reduce plant size leaving the crown moth to have a substantial impact on *Onopordum* thistles over the summer period (Swirepik and Woodburn, 2002). Monitoring of established sites throughout Australia is required to identify locations suitable for harvesting for redistribution purposes.

Identification

Adult crown moths are white to light tan (up to 15 mm long), with two dark coloured bands across their wings. Larvae are greenish brown with a black head capsule. Larval feeding causes the leaves to deform, curl upwards and die and may lead to the stunting of plants and the death of smaller rosettes (Dellow and Holtkamp, 2005).

P. Sullivan

Scotch thistle crown moth.

J. Lester

Scotch thistle rosette under attack by the crown moth.

Life cycle

Crown moths have three generations per year, commencing in early spring when adults emerge from pupal cells in thistle rosettes, one in summer and an overwintering generation as mature larvae within a cocoon. Adult females lay blue-green eggs singly on leaves in mid to late spring. After hatching, larvae feed within the petioles (leaf stalks) of rosette and stem leaves, on crown tissue and by boring into the plant root before undergoing pupation and emergence from rosettes (Dellow and Holtkamp, 2005; Briese, 2012b).

Field collecting and rearing

Rearing is unnecessary. As collections may be laborious and time-consuming aim to collect adults during the summer when the moth is most abundant. Collect moths using a sweep-net or aspirator (see Appendix 1 for technique) and store them only temporarily in a container with ventilation prior to redistribution (i.e. a few days at cool temperatures of around 15°C).

How and when to release

Ideally release at least 200 crown moth adults during summer directly onto the leaves of healthy thistles at the new nursery site as soon as possible after collecting. Record your release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for crown moth activity during spring and summer within a year of release. Adult moths may be observed on leaves and larval activity is evidenced by curled rosette leaves, dead leaves on rosettes and flowering stems, and damage to the centre of the rosette crown causing crown deformity. You can also dissect some stems to look for larvae. Record the presence and absence of crown moth as per your monitoring guidelines (Appendix 3). If present, begin monitoring agent dispersal at incremental distances away from each nursery site as per the provided guidelines. Monitor annually.

Water hyacinth

Pontederia crassipes

Water hyacinth is a free-floating, mat-forming perennial aquatic herb native to Brazil's Amazon basin (Julien, 2012b). It thrives on still or slow-moving freshwater bodies and muddy shores along the east coast of Australia, particularly north of Sydney. This floating aquatic plant has mauve flowers, shiny round leaves, and reproduces through clonal growth and viable seeds. Due to the stoloniferous production of ramets (daughter plants), water hyacinth forms large aggregates of plants in the form of floating mats (Julien, 2012b). When growing under crowded conditions, petioles (leaf stalks) elongate (up to 1 m) and when nutrient levels are elevated, the leaves may reach dinner-plate size. On the invading front of a mat, or for plants growing on the edge in uncrowded conditions, the petioles are short and bulbous (to 30 cm) and produce small kidney-shaped leaves (Julien, 2012b). Between 6 and 10 petioles are arranged along each rhizome, which supports the stolon development as well as a feathery, fibrous, submerged root system (Coetzee *et al.*, 2009). Flowers are light purple with darker blue/purple and yellow centres, and are 4 to 6 cm long and 3.5 to 5 cm wide. Flowers occur as dense spikes above plant.

Introduced to Australia as an ornamental pond plant in the 1890s (Parsons and Cuthbertson, 2001), water hyacinth was well established by 1900 (Wright and Purcell, 1995). This aquatic invader rapidly degrades waterways due to its enormous reproductive potential. Floating mats can double in size in as little as two weeks due to new daughter plants forming quickly on the ends of stolons (Penfound and Earle, 1948; Pieterse, 1978) and also because plants can rapidly regenerate from a long-lived seedbank (up to 28 years; Sullivan and Wood, 2012). Dense mats prevent light penetration into the water which in turn creates anaerobic conditions that negatively impact aquatic biodiversity. Due to its invasiveness and environmental, economic and cultural impacts, water hyacinth was listed as a Weed of National Significance in 2012.

P. Sullivan

Water hyacinth invasion in Australia.

P. Sullivan

Water hyacinth flower.

Australia introduced four species of insects from South America (two weevils and two moths) to test their potential as biocontrol agents for water hyacinth. Two of the four agents released (the weevil species, *Neochetina bruchi* and *Neochetina eichhorniae*) cause considerable damage to water hyacinth and can control it over a number of years in tropical and subtropical regions but are less effective in more temperate areas. In contrast, both species of moth have a limited impact on water hyacinth when used in isolation, but one of these species (*Niphograpta albiguttalis*), when used in combination with the two weevil species, can assist with the management of water hyacinth. The second moth (*Xubida infusellus*) is known to have established at one site at Loganholme in south-eastern Queensland. It has a limited impact and will not be discussed further in this section.

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Water hyacinth in Papua New Guinea: before biocontrol (top) and after biocontrol (bottom).

Recommendation

The two weevils *Neochetina eichhorniae* and *Neochetina bruchi* should be used in combination as they coexist well in the field and their damage is synergistic.

Water hyacinth weevils *Neochetina eichhorniae* and *Neochetina bruchi*

The water hyacinth weevils *Neochetina eichhorniae* and *Neochetina bruchi* are discussed concurrently as they have similar biologies and impact.

Neochetina eichhorniae from Uruguay was first released in Australia in 1975 (Julien, 2012b). Fifteen years later, *N. bruchi* (also from Uruguay) was released in south-east Queensland, Australia. Both weevils continue to be used in combination for the effective control of water hyacinth in tropical and subtropical regions of Australia. Adult feeding damage reduces the photosynthetic ability of the leaves, while internal feeding by the creamy white larvae exposes the leaves and petioles to pathogens and waterlogging which cause the petiole bases to rot and collapse, eventuating in plant death. Moderate to severe weevil infestations cause plants to be shorter with smaller leaves, fewer daughter plants and flowers, lower tissue nutrient content, and reduced overall vigour than uninfested or lightly infested plants (Center and Van, 1989).

Identification

Neochetina eichhorniae adults are 4 to 5 mm long and dark grey, while *N. bruchi* adults are 5 to 6 mm long and brown/grey. The adults of both species are characterised by two small, elongated black marks along the centre of their backs, with *N. eichhorniae* having longer marks than *N. bruchi*. Distinguishing between species can become increasingly difficult as they age.

P. Sullivan

Water hyacinth weevils:
Left – Neochetina bruchi (larger, light brown/grey, and shorter centrally located marks on back).
Right – Neochetina eichhorniae (smaller, dark grey, and longer centrally located marks on back).

P. Sullivan

Adult water hyacinth weevils and leaf feeding scars.

USDA

Water hyacinth weevil larva and petiole damage.

Adult weevil feeding damage on water hyacinth is characterised by square feeding scars on leaves and upper petioles (leaf stalks). The feeding scars are often used in field assessments to determine the presence and density of adults. Larval feeding is identified through the browning of petioles and curling of leaves and results in more damage to water hyacinth plants than adult feeding.

Life cycle

The water hyacinth weevils have multiple generations per year, with up to four generations occurring in tropical regions. Female weevils lay up to 400 eggs cyclically over a life span of up to 300 days (Center, 1994). Eggs are either laid singly within the leaf or petiole (*N. eichhorniae*) or several at each oviposition site (*N. bruchi*), and hatch 7 to 10 days later at temperatures of around 24°C (Center *et al.*, 2002). Larvae tunnel within the plant and feed in the petiole and crown. After completing three instars (lasting 35 to 40 days), larvae exit the plants and pupate in the upper root area, within a cocoon made of root hairs.

Field collecting and rearing

Rearing water hyacinth weevils is time consuming but productive if a continuous supply of weevils is required for your site. Mass-rearing centres may be able to supply you with release consignments of the weevils, so check with your local weed or biosecurity officer for availability and suppliers first.

Alternatively, adult weevils can be easily hand-collected from the field in summer. During the day, weevils can be found hiding in small spaces toward the plant crown, such as at the base of petioles and in newly formed leaves. Adults can also be collected by the submergence method (refer to Appendix 1). After infected plants are submerged for half an hour (just below the water surface), adults can readily be collected as they float to the water surface. Ideally, a minimum of 200 weevils is required for your release

site. Prior to redistribution, adults can be stored temporarily (at cool temperatures using an ice brick) in sealed containers containing some leaf material and covered either with a lid with small air holes or insect mesh for ventilation (i.e. for a few days at around 15°C).

P. Sullivan

Equipment required to collect adult water hyacinth weevils through submergence.

Recommendation

Be careful when working with water hyacinth. New infestations can easily occur (both locally and downstream) when daughter plants detach.

It is preferable to collect and release only adult weevils to minimise the spread of contaminated material to new areas (see page 10 on practising good hygiene).

How and when to release

Ideally release between 200 and 500 adult weevils in full sun on healthy water hyacinth plants from spring to summer (this is ideal as populations build up quickly in the warmer months before decreasing in winter). Record release information as per your Weed Biocontrol Release Form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for weevil presence by examining the plants for the characteristic adult feeding scars on the leaves and petioles. Old feeding scars are brown while new feeding scars are green. The number of feeding scars is positively correlated with the size of the adult weevil population. If weevils are observed at the release site, begin monitoring for dispersal at incremental distances away from your nursery site as per your monitoring guidelines (Appendix 3). Monitor annually ideally at the end of summer.

Water hyacinth moth
Niphograpta albiguttalis

First released in Australia in 1977, the water hyacinth moth, *Niphograpta albiguttalis* (formerly *Sameodes albiguttalis*), from Argentina (Julien, 2012b), is now widely established, although populations are patchy. When feeding, larvae tunnel inside the petioles of water hyacinth preventing growth and sometimes killing the leaf buds. Larvae prefer to feed on young, bulbous plants that are typically found in new infestations or along the edge of water hyacinth mats. Feeding damage by the moth larvae therefore helps to reduce the dispersal of water hyacinth downstream from an infestation. By itself, *N. albiguttalis* does not control water hyacinth. However, it can help contribute to the management of water hyacinth when used in combination with the water hyacinth weevils.

M. Julien

Adult water hyacinth moth.

Identification

Adult moths are 6 to 10 mm long and have a 17 to 25 mm wingspan. Their colour varies from golden-yellow to charcoal-grey, with brown, black and white markings. The larvae are very small, ranging from 1 mm in the first instar (growth stage) to 5 mm in later instars (ARC PPRI, 2015a). Larval feeding causes small, dark spots to appear on the surface of the petioles. Severe damage causes leaves and petioles to wilt, turn brown and rot.

Life cycle

In Argentina, the water hyacinth moth completes up to five generations per year (DeLoach and Cordo, 1978). In Australia, the generation time can be as short as four weeks. Adults are nocturnal and only live for four to nine days (ARC PPRI, 2015a). During this time, they do not feed and are found resting on the under surface of leaves during the day. Females lay an average of 300 eggs which are deposited in leaf tissue (DeLoach and Cordo, 1978) either singly or in small groups. Eggs are often deposited in existing leaf injuries, abrasions or even in the feeding scars of the water hyacinth weevils. The eggs hatch 5 to 10 days after oviposition, and first instar larvae feed within the leaf tissue. After a couple of days, the larvae bore into the petioles and leaf buds. Late instar larvae burrow and feed in the crown of the plant, which prevents any further growth. Late instar larvae exit the last feeding area and bore into a relatively undamaged petiole where they pupate in a white cocoon (ARC PPRI, 2015a).

Redistribution

This moth is widespread and does not require redistribution.

Monitoring establishment and dispersal

Look for water hyacinth moth presence along the edge of water hyacinth mats. During the day, inspect for adult moths on the underside of leaves. Evidence of larval presence include small dark spots caused by feeding within the petioles of young, bulbous plants. If present, record your sighting on the Australian biocontrol Hub and begin monitoring agent dispersal as per your monitoring guidelines (Appendix 3). Monitor annually.

Water lettuce

Pistia stratiotes

Water lettuce is a free-floating, perennial aquatic herb thought to be native to the tropical regions of America, Asia, Malesia, Africa, and the Northern Territory of Australia (Gillett *et al.*, 1988; Parsons and Cuthbertson, 2001). However, considering there is a large diversity of natural enemies associated with water lettuce in South America, it is most likely that this geographic region constitutes its native range (Waterhouse, 1994). Water lettuce is frost sensitive, but thrives in tropical and subtropical freshwater lakes, dams and slow-flowing streams of the Northern Territory, Queensland, New South Wales and Western Australia (Parsons and Cuthbertson, 2001). The entire plant resembles a small, floating, open head of lettuce. Plants have wedge-shaped, pale green, spongy leaves up to 15 cm long, which are covered in water-repellent hairs. Root systems are unbranched and feathery, up to 80 cm long. Flowers are inconspicuous and whitish-green (up to 15 mm long). Water lettuce reproduces both from seed and vegetatively from daughter plants.

The introduction history of water lettuce is unknown, but it was first recorded in the Northern Territory in 1887 (Parsons and Cuthbertson, 2001). This aquatic invader quickly degrades ecosystems, causing environmental (e.g. creation of anaerobic conditions which in turn affect biodiversity), economic (e.g. hydroelectric flows are reduced) and cultural problems (e.g. interfering with fisheries) with its rapid growth, mat-forming habit and ability to rapidly disperse. Reproduction is both through the production of daughter plants (vegetative) and viable seeds (sexual). The daughter plants are connected to the parent plants through stolons. When these stolons break, the daughter plants are freed from the parent mass, enabling them to disperse and form new colonies (Parsons and Cuthbertson, 2001).

P. Sullivan

Water lettuce plants.

Water lettuce seeds contain an air chamber which allows them to initially float (and disperse in the water currents) before sinking to the bottom of the waterbody. Seeds germinate in the sediment in early summer when water temperatures rise above 20°C. As they grow, they become buoyant and float to the surface.

Australia introduced one species of insect (the water lettuce weevil, *Neohydronomus affinis*) from Brazil in 1981 for consideration as a potential biocontrol agent (Harley *et al.*, 1990). The water lettuce weevil was deemed host specific and approved for release in Australia in 1982 around the outskirts of Brisbane (Day, 2012b). High populations of the weevil cause considerable damage and can control water lettuce within several years in tropical and subtropical regions.

A. McConnachie

Infestation of water lettuce in Maitland, New South Wales.

Recommendation

Use the weevil (*Neohydronomus affinis*) to control water lettuce in contained (e.g. dams and ponds) populations, in tropical and sub-tropical regions.

Localised eradication of small isolated water lettuce infestations is sometimes attempted, using non-biocontrol methods. In these situations, it is recommended to use biocontrol to initially reduce the infestation and minimise the cost of eradication; but only if there is minimal chance that the water lettuce will disperse and create new infestations further downstream. Use herbicides and/or mechanical control if there is a reasonable chance that the infestation will disperse to new areas.

Water lettuce weevil
Neohydronomus affinis

The water lettuce weevil (*Neohydronomus affinis*) from Brazil was first released in Brisbane, Queensland in 1982 (Harley *et al.*, 1990). The water lettuce weevil was widely released and redistributed throughout the invasive range of water lettuce; with the exception of New South Wales, where water lettuce was an eradication target up until 2016. High weevil populations of around 130 individuals per plant have been found to severely damage water lettuce resulting in plant death. Moreover, within 12 to 18 months of release, the water lettuce weevil was found to have reduced *Pistia stratiotes* infestations by more than 40% (Harley *et al.*, 1990). Complete control of floating mats was observed to occur over a period of one-and-half years in tropical areas and more than two years in subtropical areas (Day, 2012b).

Identification

Neohydronomus affinis adult weevils are small (about 1.8 mm long) and their colour varies from brown to bluish-grey (ARC-PPRI, 2015b). They are covered in dense scales and have a distinctive 'chevron-like' pattern as a result of bare patches where the scales have rubbed off. Adults feed externally on the leaves creating tiny shot holes that may extend all through the leaf tissue. They are found on the underside of leaves, amongst the hair, above the water line. The yellow-coloured larvae are usually not externally visible as they tunnel within the leaves. On occasion, however, they can be seen externally on the underside of leaves and between the leaf ribs.

Life cycle

The water lettuce weevil has multiple generations per year, which are largely dependent on the temperatures experienced at each release site (i.e. warmer sites experience more generations than cooler sites). Females lay eggs singly on the upper leaf surface in shallow pits, under the epidermis of young leaves (ARC-PPRI, 2015b). The eggs are covered in a black substance and hatch in three to four days (Harley *et al.*, 1990). Larvae feed inside the petioles and spongy leaf tissue while adults feed externally on the leaves. Larval stage duration is 11 to 14 days, followed by pupation in small chambers lasting four to five days (ARC-PPRI, 2015b). In

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Adult water lettuce weevils.

summer, the entire life cycle is completed in 25 to 30 days.

Field collecting and rearing

Small-scale rearing of weevils is undesirable as water lettuce is intensively managed in temperate Australia (especially in New South Wales) and the creation of satellite populations of the weed target for these activities is discouraged. Mass-rearing centres may be able to supply you with release consignments of the weevil, so check with your local weed or biosecurity officer for suppliers and availability first.

Alternatively, adults can be easily hand collected from the field in summer. Use the submergence method (refer to Appendix 1). After infected plants are submerged for half an hour (just below the water surface), adults can readily be collected as they float to the water surface. This process is likely to continue for up to a day. Ideally, a minimum of 200 weevils is required for your release site. Prior to redistribution, adults can be stored temporarily (at cool temperatures using an ice brick) in sealed containers containing some leaf material inside and cover either with a lid with small air holes or insect mesh for ventilation (i.e. for a few days at around 15°C).

Recommendation

Be careful when working with water lettuce. New infestations can easily occur (both locally and downstream) when daughter plants detach.

It is preferable to collect and release only adult weevils to minimise the spread of contaminated material to new areas (see page 10 on practising good hygiene).

QDAF

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Fixed photo points of a water lettuce site before (top) and after (bottom) biocontrol using the weevil, in Park Ridge, Queensland.

How and when to release

Ideally release a minimum of 200 adult weevils in full sun on healthy water lettuce plants from spring to summer (best as populations build up quickly in the warmer months before decreasing in winter). To assist with establishment, release weevils in small bays away from the main waterway channel. This gives the weevils the best opportunity to establish decent-sized populations, with minimal chance of being washed downstream. Record release information as per your Weed Biocontrol Release Form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for weevil presence by examining the plants for signs of feeding damage (small adult shot holes in the leaves or signs of yellowing in the leaf tissue) three months post release. If weevils are observed at the release site, begin monitoring for dispersal at incremental distances away from your nursery site as per your monitoring guidelines (Appendix 3). Monitor annually, ideally at the end of summer.

Weedy *Sporobolus* grasses

A group of five introduced perennial unpalatable grasses of the *Sporobolus* genus are collectively known in Australia as the weedy *Sporobolus* grasses (WSG). Their common name, scientific name and geographical origin are: giant Parramatta grass (GPG), *Sporobolus fertilis*, native to Asia and some Pacific islands; Parramatta grass (PG), *Sporobolus africanus*, native to Africa; giant rat's tail grass (GRT) (species 1), *Sporobolus pyramidalis*, native to Africa and Yemen; GRT (species 2), *Sporobolus natalensis*, native to central and southern Africa; and American rat tail grass, *Sporobolus jaquemontii*, native to Mexico, Central and South America (Simon and Jacobs, 1999; Palmer, 2012).

Weedy *Sporobolus* grasses all have robust, well-rooted tussocks with tough leaf blades. *Sporobolus jaquemontii* grows up to 75 cm in height; *S. africanus* grows up to 100 cm in height, *S. natalensis* and *S. fertilis* grow up to 170 cm in height and *S. pyramidalis* grows up to 200 cm in height (Bray and Officer, 2007). Weedy *Sporobolus* grasses have leaf blades that are long (8 to 70 cm long) and narrow (0.5 to 1 cm in width). Their seed heads are usually thin and elongated resembling a 'rat's tail' like spike (8 to 80 cm long). The seed heads of GRT grass may branch to form elongated pyramidal shapes when mature.

Weedy *Sporobolus* grasses plants are usually more robust than native Australian *Sporobolus* grasses, with larger seed heads that contain more seed. While all *Sporobolus* grasses may look very similar, all WSG have leaf margins that are hairless at the leaf base. Weedy Sporobolus grasses establish in areas of at least 500 mm of annual rainfall but thrive in areas receiving greater than 700 mm. PG and GPG grow in the coastal and sub-coastal regions of Queensland, New South Wales, Victoria, South Australia and south-west Western Australia. The three GRT grasses have established in the coastal and sub-coastal areas of tropical and subtropical Queensland and New South Wales.

D. Officer

Dense giant Parramatta grass in a control area of a wick-wiping trial, showing mature and immature seed heads.

D. Officer

A paddock with patchy giant Parramatta grass that has dropped its seeds and hayed off, which makes it very easy to identify this grass during winter.

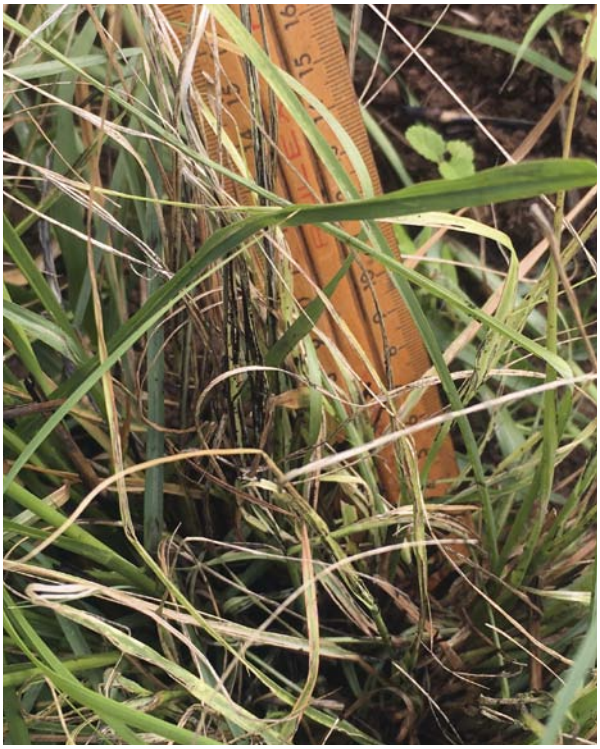
D. Officer

Dense infestation of giant rat's tail grass showing the characteristic pyramid-shaped seed head.

weedy *Sporobolus* grasses

Weedy *Sporobolus* grasses were introduced to Australia as early as the 1800s (Palmer, 2012). The methods of introduction for most species are unknown, however GPG is thought to have been introduced through contaminated hay in packing equipment, and GRT was most probably introduced as a contaminant in seed imported from Africa for pasture improvement (Bray and Officer, 2007). WSG have major economic impacts on pastoral industries because they are low in energy and protein and are poorly digested. Cattle, in grazing pastures dominated by WSG, often grow poorly and cannot maintain weight. The WSG are highly invasive in part because their seeds stick to anything that brushes past the plant when the seed head is wet from rain or dew. Because the seed is easily spread, WSG now dominate large areas of native and improved pastures in eastern Australia.

No biocontrol agents have been officially released for WSG. Several biological control agents from southern Africa including a leaf smut (*Ustilago sporoboli-indici*) and stem wasps (*Tetramesa* spp.) were identified in South Africa as potential candidates. These potential agents were never imported because they were expected to impact on native *Sporobolus* grasses (Yobo *et al*., 2009) or in the stem wasps' case were not able to be reared in quarantine. Unexpectedly, the leaf smut was discovered in Australia in 2017 in an area from Bundaberg to Gympie in Queensland (Vitelli *et al*., 2017). Anecdotal evidence has shown a significant reduction in the number of GRT inflorescences present on plants with leaf smut which means seed production is curtailed. Further research is required to determine the impact of leaf smut against WSG and native *Sporobolus* species under Australian conditions. This research will need to determine what conditions suit infection and what management strategies are required to maximise the value of the leaf smut as a biocontrol agent (while minimising its impact on off-target plant species).



D. Officer

A giant rats tail plant (Sporobolus natalensis) infected with leaf smut (Ustilago sporoboli-indici). Note the characteristic sooty black spores.

Although no agents have been imported for WSG biocontrol, the effects of a native crown rot fungus, *Nigrospora oryzae* (Ramasamy *et al*., 2008) has been shown to be effective in killing PG and GPG in some situations (Officer, 2012). Many pasture areas on the north and mid-north coast of New South Wales, that were dominated by GPG in the early 2000s, now have infestation levels below the threshold for grazing profitability.

Recommendation

Integrated control using *Nigrospora* crown rot together with existing weed control strategies can be effective for controlling the two Parramatta grasses (PG and GPG), but control takes time.

Nigrospora crown rot fungus
Nigrospora oryzae

The *Nigrospora* crown rot fungus is native to Australia. It kills PG and GPG, and infects the three GRT grass species (Officer, 2012) but has no impact on their plant health (Fletcher and Leemon, 2015). The speed with which the infection spreads is affected by climatic conditions and plant health. *Nigrospora* crown rot can reduce the size of GPG tussocks by up to 78% over a 15-month period and reduce the number of tussocks by 64% within 12 months (Officer, 2012). The impact of *Nigrospora* crown rot on the GRT grasses has been highly variable, ranging between negligible to significant. Reasons for this variability are not well understood.

Identification

Spores are pale yellow to yellowish orange when young and olive brown to black when mature. They are single celled and invisible to the naked eye (being 0.01 to 0.016 mm in diameter) but are easily seen under a 20 × magnification light microscope. Crown rot causes leaves and tillers (new grass shoots) to turn orange and the crown end of the diseased tiller to blacken and die. These same stunted and shrivelled diseased tillers are easily pulled away from the crown of the tussock. Four weeks after the orange colouring first appears, diseased tillers become a pale straw colour.

Life cycle

Nigrospora crown rot fungus is found in and on a range of plants species both living and dead. *Nigrospora* crown rot fungus only affects young tillers which often turn orange 7 to 10 days after the first good rainfall event in spring. Diseased tillers then shrivel and hay off as they age but retain a black colour at the crown (unlike the crown end of healthy tillers which are pale white). The disease will continue to proliferate through individual tussocks until late summer if there continues to be good rainfall. Over time, large tussocks shrink in size and are replaced by

D. Officer

The pale orange leaves of young giant Parramatta grass tillers with active Nigrospora crown rot.

D. Officer

An individual diseased tiller of giant Parramatta grass showing pale orange of active disease and black necrosis at the crown.

D. Officer

The short, shrivelled and hayed-off tillers in this picture have died due to Nigrospora crown rot infection four weeks previously.

smaller individuals which eventually succumb to the disease. During autumn and winter the symptoms of the disease tend to disappear and reappear the following spring.

Field collecting and rearing

Transplanting diseased plants is the best way to introduce *Nigrospora* crown rot to new sites. Contact your local weeds professional (e.g. council weeds officer) for information on locations of *Nigrospora* crown rot, infected plants. Diseased plants can be found from late spring until early autumn by looking for orange-coloured young leaves. Transplantation should be done when the disease is active in one or more tillers of a plant. The spores of *Nigrospora* crown rot mainly disperse overland in water after rain and via animal and vehicle movement, however, they can also travel short distances in the air. There is no state-based rearing of *Nigrospora* crown rot fungus.

How and when to release

Place diseased plants in areas invaded by WSG every 10 to 20 m along ridgelines and high areas to help facilitate the downhill movement of spores through water flow. Alternatively, transplant diseased plants along frequently used stock paths and high traffic areas to facilitate stock dispersal of the spores in WSG infested areas. The ridgeline and high areas tend to have shallow soil with poor water holding capacity. As insurance for successful inoculation, also plant around one quarter of the diseased plants among healthy WSG in deeper soils. Diseased plants and 5 to 10 cm of their surrounding soil can be transplanted into healthy populations, but only after considering the risk that they may be carrying other biosecurity matter such as soil-borne pathogens or other weed seeds. Always seek advice from your local council weeds officer before transplanting WSG. Stomp around the transplanted, diseased plant to exclude air and ensure good root ball-to-soil contact.

Recommendation

Practise hygiene

Relocate diseased plants locally to reduce the chance of moving weeds, arthropods and pathogens other than *Nigrospora* crown rot to a new location. Due to the biosecurity risk, always seek advice from your local weeds officer before transplanting weedy *Sporobolus* grasses.

Before transplanting, wait until both the source and recipient areas have received a good soaking of rain and are likely to remain damp at least for the next month. Successful transplanting will not take if you transfer diseased plants that are water stressed into a disease-free area suffering from drought. Record release information as per your weed biocontrol release form (Appendix 2) and submit a copy to your local weed or biosecurity officer.

Monitoring establishment and dispersal

Look for signs of disease, evidenced by pale orange leaves, from spring to autumn. Symptoms most often appear 7 to 10 days after the first, good-rainfall event in spring. Disease symptoms always show up in new growth (not old tillers). Once the disease has killed the tiller, it will have a shrivelled, hayed-off appearance and importantly, can be easily removed from the plant by hand because the crown tissue has died. Monitor transplant sites initially after six months and reintroduce diseased plants if no disease is evident after 12 months. If crown rot is present, begin monitoring agent dispersal at incremental distances away from each nursery site as per your guidelines (Appendix 3). Continue to monitor annually.

Further information

Appendix 1

Biocontrol agent field collection and redistribution techniques

Redistribution of biocontrol agents can occur when a well-established agent has increased its population to a level high enough to be field collected. Several techniques are available to collect and redistribute biocontrol agents. These techniques may be used individually or in combination, depending upon the species of biocontrol agent and the collection site.

Aspirators

Aspirators (also called pooters) are used when you require a targeted approach to collecting the desired agent and/or to prevent the unintended transport of contaminants. They are often used in combination with other techniques, such as insect nets or beating trays, for agents such as small to medium sized insects. There are two types of aspirators, manual and mechanical:

- **A manual aspirator** has two tubes that enter the collecting container. One tube, the sucking tube, is equipped with fine mesh over the end closest to the container to prevent agents being sucked into the mouth. To collect agents, the collector creates a partial vacuum on the sucking tube, while placing the far end of the second tube near the agent to draw it into the collecting container. You can vary this pressure to minimise damage to the agent, so this device is useful for collecting smaller insect species.
- **A mechanical aspirator** is simply a motorised version of a manual aspirator, using a small fan to create the partial vacuum. Fine gauze needs to be placed between the fan and the container to prevent agents being drawn into the fan. A mechanical aspirator reduces the operator's exposure to allergens, e.g. chitin and insect frass (waste products).

A. McConnachie

Manual aspirator (left) and mechanical aspirator (right).

A. McConnachie

Using a pooter to collect insects on mother-of-millions in Madagascar.

Beating

This method consists of placing a tray or sheet beneath the weed of interest. The collector taps or beats the foliage to dislodge the agents. The agents can then be collected from the tray or sheet. This method is useful for cryptic insects and mites that are not readily visible on the host plant. Sometimes a sieve or aspirator are helpful to assist with this process.

A. McConnachie

Beating agents from gorse (Ulex europaeus) to confirm establishment of the gorse soft shoot moth (Agonopterix umbellana) in Sunny Corner, New South Wales.

Recommendation

Upon collection, insect agents can be placed into a sealed container with small air holes for ventilation prior to redistribution. To prevent desiccation and/or slow active insects down, place them in an Esky or refrigerator prior to redistribution.

Sweep netting

Sweep netting is a commonly used method for collecting jumping, flying and very active agents that reside on the weeds surface. The sweep net is moved from side to side several times, either just above the weed or within the main vegetative growth, to collect the insects. To prevent very active agents from escaping, the net should be twisted through 90 degrees to trap the contents.

P. Sullivan

Collecting Paterson's curse flea beetles with a sweep net.

A. McConnachie

Turning the opening of the sweep net through 90 degrees to prevent escape of insects.

Hand collecting

While laborious and time consuming, hand collecting is sometimes necessary when agents, often sedentary, are not easily dislodged from the plant material. Use suitable implements (e.g. tweezers, tongs and vials) and appropriate clothing for working with agents and/or target plant species.

S. Ivory, SARDI

Hand collecting cochineal (Dactylopius opuntiae) for redistribution on wheel cactus (Opuntia robusta) in South Australia.

Field cage technique

The use of field cages can be useful for biocontrol agents that develop within the stem or root of the plant and that have asynchronous eclosion (staggered or mistimed emergence of adult males and females) or particular requirements for mating. The redistribution of the horehound clearwing moth and the dock moth are examples of species which could be redistributed in this way. The following steps are recommended:

- Identify stems which contain internally feeding larvae. To do this look out for stems that are hollowed out and appear green (horehound) or stunted plants that have died off prematurely with a mattock (docks) – these are most likely to contain larvae.
- Trim down stem in 2 cm increments using secateurs, from the top of the stem working your way down. If there are signs of tunnelling and frass (waste products), retain the rootstock.
- For both the horehound and dock moths, at least 100 females will be required to establish a viable nursery site, so assuming a 1:1 sex ratio, try to collect around 200 root stocks.
- Infested rootstocks will need to be caged in a moth-proof field cage (e.g. made of 70% woven shade cloth) with dimensions of about 2 × 2 × 2 m in the field in early September (docks) or October (horehound).
- Plant root stocks containing larvae into trays filled with a well-draining material such as loam soil or vermiculite. Plant the rootstock in an upright position at approximately the same depth as when they were collected.
- Once adults emerge, male moths should be left in the cage so that they are able to find and mate with newly emerged female moths. A daily supply of fresh flowering horehound or docks will be needed to provide nectar to sustain the caged males.
- The female moths should be collected each evening for release. Daily attendance of the cage will be required from late-September until late-January, after which all the moths should have emerged.

Cuttings

Agent-infested weed cuttings are useful for redistributing some pathogens, mites, small insects, some larval stages and agents that have life stages that develop within the weed. Ideally, secure your cuttings (using plant wire or zip ties) to the release plant to limit desiccation and/or loss of the agent. As cuttings desiccate, agents and their offspring will naturally disperse onto new host plants.

Use extra diligence to minimise the inadvertent transportation of predators and parasitoids when transporting plant material containing agents. Inspect your cuttings for predators and parasitoids before transporting any material.

P. Sullivan

Scotch broom cuttings containing galls of the biocontrol agent secured to the host plant with plant wire.

Redistribution of pathogens – the spore water method

This technique is used to redistribute some pathogen agents (e.g. rusts). A spore water solution is made by washing infected leaves in rainwater to dislodge the spores. A handful of heavily infected leaves will make up several litres of spore solution. Remove leaves and debris by pouring the solution through a sieve before decanting into a spray bottle/unit. Spray the solution onto the target plant as soon as possible after mixing, as spores generally degrade over time in solution.

Spray the spore water solution on the underside of leaves on mild days or in the cool of the evening (if day temperatures are >25°C). Rust fungi attack the plant by entering the leaves through open stomata, most of which are located on the underside of leaves. Stomata are open wider in moderate temperatures and when the humidity is high. Keep in mind that for some spores, leaves need to remain moist for a while for infection to occur. Artificially increasing the humidity by covering the inoculated weeds with a plastic bag and by inoculating with spore water over rainy periods can help with pathogen establishment.

Recommendation

Only use rainwater to create the spray solution, as chlorinated town water and minerals from bore water may negatively affect the spores. Ensure spray units are cleaned (flushed) before use.

Follow-up after spraying spore water. If no sign of infection is seen within two months (species dependent), another dose may be required. The spore water technique does not work in all situations or for all pathogens. Repeated failure may indicate that conditions are unsuitable for establishment and/or a different application technique may be required. Monitor and record your activities (see Appendix 3).

S. Ivory, SARDI

Spore water solution preparation for distributing the bridal creeper rust fungus.

P. Sullivan

Early signs of rust damage on bridal creeper after inoculation using the spore water release technique.

There are a variety of other methods for redistribution of rusts. For example, see redistribution guidelines by CSIRO: https://research.csiro.au/crofton-weed/more-information-on-rust-fungus/

Smoker tents

Agents (insects) that are difficult to access can be flushed out using smoker tents. For example, those like the gorse soft shoot moth living within a spikey habitat. Smoke can be pumped upwards from the plant base using a bee smoker to flush insects into a small tent made of insect gauze (same material as the sweep nets). Insects fly upwards to evade the perceived 'fire' and are trapped inside the tent. The tent is then brought down to ground level and the insects are collected from inside the tent using suitable methods, as outlined above.

A. McConnachie

Use of a smoker tent to collect the gorse soft shoot moth.

A. McConnachie

Collecting the gorse soft shoot moth after collection using the smoker tent method.

Emergence traps

Emergence traps are an effective passive technique for collecting a large number of agents (usually insects) from vegetative material. They can be any shape and size but must be suitable for the target weed (e.g. plastic box for aquatic weeds).

Material is collected in the field and transferred into the emergence trap which is then placed close to a light source but out of direct sunlight. The end of the trap, closest to the light source, requires a collecting chamber. A jar is often used as a collecting chamber as a lid can be easily attached to the emergent trap removed for agent collection. The emergent trap can also be wrapped to darken it and expedite insect movement to the collecting chamber.

A. McConnachie

Emergence trap used to capture large numbers insects off a bulk weed sample.

Submergence method

A submergence method or technique is useful for collecting agents (especially weevils) from a variety of aquatic weeds. The aquatic weed is submerged to a depth of approximately 30 cm using a metal grid weighted down by bricks or rocks. After a period, agents float to the surface and can be collected. While some float immediately to the surface, others can take several days. For example, the salvinia weevil can prolong its survival underwater due to finding air bubbles in the mass of submerged plant material; as such, they can take up to two days to reach the surface.

P. Sullivan

Grid and bricks used to submerse water hyacinth for weevil collection. Weevils generally float to the surface within 30 minutes.

Appendix 2

Example only: Release form for Weed Biological Control Agents

Please fill in this form each time you release an agent and forward it to your weed or biosecurity officer.

This is the kind of information you would fill in to release your agents. You may need to modify this form to suit the context of your release. You can submit this information directly to the Australian Biocontrol Hub (https://biocollect.ala.org.au/biocontrolhub).

Name the weed and agent including the number required

Weed name	
Agent name	
How many units? (eggs, larvae, adults, infected cladodes, cuttings, etc.)	

Releaser

Name		Organisation	
Email		Phone	

Release site (mark the release site with either flagging tape or a marker peg for future monitoring and a fixed-point photograph associated with marker peg)

Site location	Latitude: Longitude:		
No. & Street / Road			
Area / Village / Town		Post code	
Creek / River System		Region	

Landowner (if different from releaser)

Name		Phone	

Weed infestation

Weed density [tick box]	Light: scattered patches and clumps (1–10%) ☐	Moderate: some spaces between plants (11–50%) ☐	Dense: completely covering in thick layer (>50%) ☐
Area of weed infestation (approximate size m^2 or ha^2):			

Release details

Date of release:		Time:		Temp (°C):	
Weather (please circle):	sunny	overcast	windy	light rain	heavy rain
Draw/insert map here			General comments		

Appendix 3

Example only: Biocontrol Agent Monitoring form

Weed name		Agent name	
Site name		Date	
Site location	Latitude: Longitude:	Time of day	
Observer(s)			
Organisation			

Weather Conditions when monitoring

1. Sunny / Partly cloudy / Overcast / Rain
2. Strong wind / Light wind / Calm
3. Temperature (°C): _____

Insect or Disease information

4. Number and life stage of insect observed: _____ and _____
   or symptom/s of disease observed: _____
5. Overall agent effect: None / Occasional / Patchy / Heavy / Severe
6. Time spent searching (mins): _____
7. Area searched (Number m wide × Number m deep): _____ × _____
8. Furthest distance agent/agent damage found from release point (m):

Weed information

9. Name of subspecies/flower colour (if applicable: _____
10. Infestation (approximate size m^2 or ha^2): _____
11. Percentage cover at densest accessible point: _____
12. Photos taken: Yes / No ***Ensure GPS is turned on for digital photos***
13. Photo file name: _____ Photo location: _____

Comments

14. Have any of the following happened to the site recently?
    Agent removal / Mowing / Spraying / Grazing / Flood / Drought / Fire / Other?
15. Please use the back of this sheet to record any further observations or comments about the site.

Additional comments

Instructions for filling out Biocontrol Agent Monitoring form

- **Weed and Agent names** – use common names as per conventions in WeedWise (https://weeds.dpi.nsw.gov.au).
- **Site name** – please use the same name for a site each time it is monitored.
- **Site location** – please follow the format requested.
- **Observer(s)** – the names of people who helped.
- **Organisation** – the name of your organisation.

Current Weather Conditions

1–3. Choose the words that best describe the weather conditions.

Insect or Disease information

4. Counts and life stage of agents found, e.g. 30 galls, 20 adults, five pupal cases, etc. Refer to the best practice guidelines for each species in the manual.
5. Overall agent effect: Record the impact or density of agent on the weed. None (no impact or presence); occasional (impact/agent present but not common), patchy (impact/ presence is variable throughout the site); heavy (the majority of plants are impacted showing signs of stress/agent present commonly across site); and severe (severe damage is obvious and widespread/ agent present on almost all plants).
6. Record how long you spent actively searching. Ideally spend five minutes checking the release point intensively and then look more widely around the site for another 10 minutes.
7. Estimate the size of the area that you conducted your search (e.g. 20 m wide × 10 m deep).
8. If you have time to look further away, we would like to know how far away from the release point there is evidence of the agent being present.

Weed information

9. Record the subspecies of the weed, e.g. white flowered lantana growing at the release point.
10. Estimate and record the approximate size of the infestation in m^2 or ha.
11. Estimate the percentage cover of the weed at the densest accessible point over an area of 5 × 5 m, or if the site doesn't lend itself to a square use an equivalent sized shape.
12. Please indicate if you have taken photos.
13. If you have taken photos, please record the file name for the photo and attach hard copies to this form if you can.

Comments

14. Please indicate if any of these important events have happened to the site.
15. Tell us any other important information we should know about the site (e.g. whether you have been harvesting for release at new sites). Use the top of this page if you need extra space.

Acknowledgement

This form is an adaptation of a single species monitoring form used by New Zealand Landcare Research. Available from The Biological Control of Weeds Book at https://www.landcareresearch.co.nz/discover-our-research/biosecurity/weed-management/using-biocontrol.

Appendix 4 Summary of biocontrol programs, agent availability, recommendations and weed control

Program status

- **Limited control** (undetermined or negligible)
- **Minor control** (context or situation dependent)
- **Effective control** (context or situation dependent)
- ★ **Available from mass rearing centres** (e.g. NSW DPI, Grafton Primary Industries Institute)

Australian agencies responsible for the development of biocontrol agents to the point of release

- ◻ Agriculture Victoria
- ◇ Commonwealth Prickly Pear Board and the Queensland Prickly Pear Travelling Commission
- ● CSIRO Australia
- ○ Department of Primary Industries, Parks, Water and Environment Tasmania
- ❖ NSW Department of Primary Industries
- ◆ Queensland Department of Agriculture and Fisheries
- ⌘ Western Australian Department of Agriculture
- △ Unknown

Current agency names are given, and symbols above are indicated in the table below in alphabetical order.

Note: Where biocontrol is not suitable for your site, refer to your State or Territory contacts for other management options (e.g. NSW WeedWise). See 'Further Information – Other electronic resources' on page 201. This table is not extensive and only includes common agents listed for the biocontrol programs covered within this manual. Further, agent availability and recommendations are dynamic.

Weed	Biological control agent(s)	Recommendation	Weed control
Alligator weed ● (*Alternanthera philoxeroides*)	Alligator weed flea beetle (*Agasicles hygrophila*)	Widespread distribution. **Suitable** for redistribution (aquatic form only).	Aquatic Terrestrial
	Alligator weed moth (*Arcola malloi*)	Not recommended for redistribution. Not available for release.	
Bitou bush ● (*Chrysanthemoides monilifera* subsp. *rotundata*)	Bitou tip moth (*Comostolopsis germana*)	Widespread distribution. Redistribution unnecessary but **suitable** to accelerate dispersal.	
	Bitou leaf-roller moth (*Tortrix* sp.)	Widespread distribution. **Suitable** for redistribution.	
	Bitou seed fly (*Mesoclanis polana*)	Widespread distribution, no need for redistribution.	
	Bitou tortoise beetle (*Cassida* sp. 3)	Isolated establishment, not available for release.	
Boneseed ◻ ● (*Chrysanthemoides monilifera* subsp. *monilifera*)	Boneseed leaf-buckle mite (*Aceria* sp.)	Isolated establishment. **Suitable** for redistribution.	
Blackberry ◻ ● (*Rubus fruticosus* aggregate)	Rust fungus (*Phragmidium violaceum*)	Widespread distribution, no need for redistribution.	
Blue heliotrope ● (*Heliotropium amplexicaule*)	Blue heliotrope leaf-beetle (*Deuterocampta quadrijuga*)	Isolated establishment. **Suitable** for redistribution at specific sites only.	

Weed	Biological control agent(s)	Recommendation	Weed control
Bridal creeper (common form) ● (*Asparagus asparagoides*)	Bridal creeper rust fungus (*Puccinia myrsiphylli*)	Widespread distribution. Redistribution generally unnecessary and only recommended at specific sites.	
	Bridal creeper leafhopper (*Erythroneurini* tribe – undescribed species)	Widespread distribution. Redistribution generally unnecessary and only recommended at specific sites.	
	Bridal creeper leaf beetle (*Crioceris* sp.)	Isolated establishment, not available for release.	
Cacti			
Boxing glove cactus ◆ (*Cylindropuntia fulgida* var. *mamillata*)	*Dactylopius tomentosus* 'cholla' lineage	**Suitable** for redistribution.	
Hudson pear: brown-spined ◆ (*Cylindropuntia tunicata*)	★*D. tomentosus* 'acanthacarpa var. echinocarpa' lineage	**Available** for release.	
Hudson pear: white-spined ◆ (*Cylindropuntia pallida*)	★*D. tomentosus* 'californica var. parkeri' lineage	**Available** for release.	
Jumping cholla ◆ (*Cylindropuntia prolifera*)	★*D. tomentosus* 'californica var. parkeri' lineage	**Available** for release.	
Klein's cholla ◆ (*Cylindropuntia kleiniae*)	*D. tomentosus*	**Suitable** for redistribution.	
Pencil cactus ◆ (*Cylindropuntia leptocaulis*)	*D. tomentosus*	**Suitable** for redistribution.	
Rope pear ◆ (*Cylindropuntia imbricata*)	*D. tomentosus* 'cylindropuntia' lineage	**Suitable** for redistribution.	
Snake cactus ◆ (*Cylindropuntia spinosior*)	*D. tomentosus* 'bigelovii' lineage	**Suitable** for redistribution.	
Harrisia cactus ◆ (*Harrisia martini, H. pomanensis, H. tortuosa*)	Cactus mealybug (*Hypogeococcus festerianus*)	**Suitable** for redistribution.	
Common prickly pear ◊ (*Opuntia stricta*)	*Dactylopius opuntiae* 'stricta' lineage	**Suitable** for redistribution.	
	Cactoblastis moth (*Cactoblastis cactorum*)	Widespread distribution, no need for redistribution.	

Cacti continued overleaf/…

Weed	Biological control agent(s)	Recommendation	Weed control
Riverina pear ◊ (*Opuntia elata*)	*Dactylopius opuntiae* ('stricta' and 'ficus' lineages)	**Suitable** for redistribution.	
	Dactylopius ceylonicus	**Suitable** for redistribution.	
	Cactoblastis moth (*Cactoblastis cactorum*)	Widespread distribution, no need for redistribution.	
Smooth tree pear ◊ (*Opuntia monacantha*)	★*Dactylopius ceylonicus*	**Available** for release.	
Tiger pear ◊ (*Opuntia aurantiaca*)	*Dactylopius austrinus*	**Suitable** for redistribution.	
	Cactoblastis moth (*Cactoblastis cactorum*)	Widespread distribution, no need for redistribution.	
Velvety tree pear ◊ (*Opuntia tomentosa*)	*Dactylopius opuntiae* 'stricta' lineage	**Suitable** for redistribution.	
Wheel cactus ◊ (*Opuntia robusta*)	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Opuntia elatior ◊	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Opuntia engelmannii ◊	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Indian fig ◊ (*Opuntia ficus-indica*)	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Opuntia humifusa ◊	Cactoblastis moth (*Cactoblastis cactorum*)	Widespread distribution, no need for redistribution.	
Opuntia puberula ◊	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Opuntia schickendantzii ◊	*Dactylopius ceylonicus*	**Suitable** for redistribution.	
Opuntia streptacantha ◊	*Dactylopius opuntiae* 'ficus' lineage	**Suitable** for redistribution.	
Cape broom ● (*Genista monspessulana*)	Cape broom psyllid (*Arytinnis hakani*)	Widespread distribution. **Suitable** for redistribution.	
Cat's claw creeper ◆ (*Dolichandra unguis-cati*)	Cat's claw creeper leaf-feeding tingid (*Carvalhotingis visenda*)	Widespread distribution. **Suitable** for redistribution.	
	★Cat's claw creeper jewel beetle (*Hylaeogena jurecki*)	Widespread distribution. **Available** for release.	
	Cat's claw creeper leaf tying moth (*Hypocosmia pyrochroma*)	Isolated establishment. Not available for release.	

Weed	Biological control agent(s)	Recommendation	Weed control
Crofton weed ● ◆ (*Ageratina adenophora*)	Crofton weed rust fungus (*Baeodromus eupatorii*)	Widespread distribution. **Suitable** for redistribution.	
	Crofton weed gall fly (*Procecidochares utilis*)	Widespread distribution, no need for redistribution.	
	Crofton weed leaf spot fungus (*Passalora ageratinae*) ✦	Widespread distribution, no need for redistribution.	
Docks ⌘ (*Rumex* spp.)	Dock moth (*Pyropteron doryliformis*)	Widespread distribution. Redistribution unnecessary but **suitable** to accelerate dispersal.	
Gorse □ ● ○ (*Ulex europaeus*)	Gorse seed weevil (*Exapion ulicis*)	Widespread distribution. **Suitable** for redistribution.	
	Gorse spider mite (*Tetranychus lintearius*)	Widespread distribution. **Suitable** for redistribution.	
	Gorse thrips (*Sericothrips staphylinus*)	Widespread distribution but populations low. **Suitable** for redistribution.	
	Gorse soft shoot moth (*Agonopterix umbellana*)	**Suitable** for redistribution.	
Horehound □ ● (*Marrubium vulgare*)	Horehound plume moth (*Wheeleria spilodactylus*)	Widespread distribution. Redistribution unnecessary but **suitable** to accelerate dispersal.	
	Horehound clearwing moth (*Chamaesphecia mysiniformis*)	**Suitable** for redistribution but laborious.	
Lantana ● ◆ (*Lantana camara*)	Lace bug (*Teleonemia scrupulosa*)	Widespread distribution, no need for redistribution.	
	Leaf-mining hispine beetle (*Uroplata girardi*)	Widespread distribution, no need for redistribution.	
	Leaf-mining beetle (*Octotoma scabripennis*)	Widespread distribution, no need for redistribution.	
	Stem-sucking bug (*Aconophora compressa*)	Widespread distribution, no need for redistribution, but **suitable** to aid with establishment at sites where not already present.	
	Bud mite (*Aceria lantanae*)	Not available for redistribution.	

Lantana continued overleaf/…

Weed	Biological control agent(s)	Recommendation	Weed control
Lantana ● ◆ (*Lantana camara*) …/ continued from previous page	Flower-feeding moth ✦ (*Lantanophaga pusillidactyla*)	Widespread distribution, no need for redistribution.	
	Flower and bud-feeding moth (*Crocidosema lantana*)	Widespread distribution, no need for redistribution.	
	Leaf-feeding moth (*Hypena laceratalis*)	Widespread distribution, no need for redistribution.	
	Leaf-feeding moth (*Neogalea sunia*)	Widespread distribution, no need for redistribution.	
	Leaf-feeding moth (*Salbia haemorrhoidalis*)	Widespread distribution, no need for redistribution.	
	Leaf-mining hispine beetle (*Octotoma championi*)	Widespread distribution but populations low. No need for redistribution.	
	Leaf-mining fly (*Calycomyza lantanae*)	Widespread distribution, no need for redistribution.	
	Rust (*Prospodium tuberculatum*)	Widespread distribution, no need for redistribution.	
	Seed-feeding fly (*Ophiomyia lantanae*)	Widespread distribution, no need for redistribution.	
Madeira vine ◆ (*Anredera cordifolia*)	★Madeira vine leaf-feeding beetle (*Plectonycha correntina*)	**Available** for release and redistribution.	
Mistflower ● ◆ (*Ageratina riparia*)	Mistflower gall fly (*Procecidochares alani*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	
	White smut fungus (*Entyloma ageratinae*) ✦	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	
Noogoora burr △ (*Xanthium chinense; Xanthium orientale*)	Stem-galling moth (*Epiblema strenuana*)	Widespread distribution, no need for redistribution. impact level yellow	
	Noogoora burr rust fungus (*Puccinia xanthii*) ✦	Widespread distribution, no need for redistribution.	

Weed	Biological control agent(s)	Recommendation	Weed control
Paterson's curse ● (*Echium plantagineum)*	Paterson's curse leaf-mining moth (*Dialectica scalariella*)	Widespread distribution, no need for redistribution.	
	Paterson's curse crown weevil (*Mogulones larvatus*)	Widespread distribution. Redistribution generally unnecessary and only recommended at specific sites.	
	Paterson's curse root weevil (*Mogulones geographicus*)	Widespread distribution. Redistribution generally unnecessary and only recommended at specific sites.	
	Paterson's curse flea beetle (*Longitarsus echii*)	Widespread distribution. Redistribution generally unnecessary and only recommended at specific sites.	
	Paterson's curse stem beetle (*Phytoecia coerulescens*)	Widespread but uncommon. Not available for redistribution.	
	Paterson's curse pollen beetle (*Meligethes planisculus*)	Established. Not available for redistribution.	
Ragwort ● (*Jacobaea vulgaris*)	Ragwort flea beetles (*Longitarsus flavicornis*; *Longitarsus jacobaeae*)	Widespread distribution, no need for redistribution in Tasmania. **Suitable** for redistribution in Victoria to enhance dispersal.	
	Ragwort stem and crown-boring moth (*Cochylis atricapitana*)	Widespread distribution, no need for redistribution.	
	Ragwort plume moth (*Platyptilia isodactyla*)	Widespread distribution, no need for redistribution.	
Salvinia ● (*Salvinia molesta*)	★Salvinia weevil (*Cyrtobagous salviniae*)	Widespread distribution. **Available** for release.	

Weed	Biological control agent(s)	Recommendation	Weed control
Scotch broom ● ❖ (*Cytisus scoparius* var. *scoparius*)	Scotch broom gall mite (*Aceria genistae*)	Widespread distribution. **Suitable** for redistribution.	
	Scotch broom twig mining moth (*Leucoptera spartifoliella*)	Isolated establishment. Not available for release.	
	Scotch broom psyllid (*Artainilla spartiophilia*)	Isolated establishment. Not available for release.	
	Scotch broom seed bruchid (*Bruchidius villosus*)	Not available for redistribution.	
St John's wort ● (*Hypericum perforatum*)	St John's wort chrysomelid beetles (*Chrysolina quadrigemina* and *Chrysolina hyperici*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	
	St John's wort mite (*Aculus hyperici*)	Widespread distribution. **Suitable** for redistribution (narrow-leaf variety only).	
	St John's wort root beetle (*Agrilus hypericin*)	Isolated establishment. Not available for release.	
	St John's wort aphid (*Aphis chloris*)	Widespread distribution, no need for redistribution.	
	St John's wort gall fly (*Zeuxidiplosis giardi*)	Widespread distribution, no need for redistribution.	
Thistles			
Nodding thistle ● (*Carduus nutans*)	Receptacle weevil (*Rhinocyllus conicus*)	Widespread distribution, no need for redistribution.	
	Seed fly (*Urophora solstitialis*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	
	Rosette weevil (*Trichosirocalus horridus*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	
Slender thistles ● (*Carduus pycnocephalus; Carduus tenuiflorus* -winged)	Rust fungus (*Puccinia cardui-pycnocephali*)	Widespread distribution, no need for redistribution.	

Weed	Biological control agent(s)	Recommendation	Weed control
Spear thistle ◻ ● (*Cirsium vulgare*)	Receptacle weevil (*Rhinocyllus conicus*)	**Suitable** for redistribution (source from Victoria).	
	Gall fly (*Urophora stylata*)	**Suitable** for redistribution.	
	Rosette weevil (*Trichosirocalus horridus*)	**Suitable** for redistribution.	
Onopordum thistles ● ▪ Scotch thistle (*Onopordum acanthium*) ▪ Illyrian thistle (*Onopordum illyricum*) ▪ Stemless thistle (*Onopordum acaulon*)	Seed-head weevil (*Larinus latus*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	Unknown impact on stemless thistle.
	Stem-boring weevil (*Lixus cardui*)	Widespread distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	Does not survive on stemless thistle.
	Crown weevil (*Trichosirocalus briesei*)	Restricted distribution. Redistribution unnecessary but **suitable** for redistribution to accelerate dispersal.	Unknown impact on stemless thistle.
	Crown moth (*Eublemma amoena*)	Restricted distribution. **Suitable** for redistribution.	Unknown impact on stemless thistle.
Water hyacinth ● (*Pontederia crassipes*)	★Water hyacinth weevil (*Neochetina bruchi; Neochetina eichhorniae*)	Widespread distribution. **Available** for release.	
	Water hyacinth moth (*Niphograpta albiguttalis*)	Widespread distribution, no need for redistribution.	
Water lettuce ● (*Pistia stratiotes*)	★Water lettuce weevil (*Neohydronomus affinis*)	**Available** for release.	
Weedy *Sporobolus* grasses ❖ ▪ Giant Parramatta grass (*Sporobolus fertilis*) ▪ Parramatta grass (*Sporobolus africanus*)	*Nigrospora* crown rot fungus (*Nigrospora oryzae*) ✪	**Suitable** for localised redistribution.	

Footnotes: ✦ This agent was detected by researchers in the field and did not go through the formal approval process for release. ✪ This species is native to Australia and has not gone through any formal process to establish its safety or efficacy as a management tool for its target weed.

Glossary

aestivate	A period of dormancy where an organism is metabolically inactive due to high temperatures and dry conditions, usually during summer.
agent or biocontrol agent	An insect, mite, pathogen or other organism that is used to control another organism through feeding damage or infection.
arthropods	Invertebrate animals having an exoskeleton (external skeleton), a segmented body, and paired jointed appendages. They include insects, mites, spiders, centipedes, crabs, prawns, etc.
aspirator	A device that produces vacuum and is used to suck insects into a collecting container.
axil	The angle between a leaf stalk or branch and a stem or trunk.
basidiospore	A type of rust fungus spore that often infects the host, especially in spring.
biomass	The total quantity or weight of organisms in a given unit area or volume.
biotype	A group of organisms that have an identical genetic composition.
bracts	Modified leaves that are usually at the base of a flower.
capitulum (capitula plural)	A compact head of a flower structure, in particular a dense flat cluster of small flowers or florets, as in plants of the daisy family.
chlorosis	The loss of the normal green colour of leaves (and sometimes stems). It can be caused by feeding of insects (especially bugs), mites, disease, iron deficiency in lime-rich soils, or lack of light.
cladode	A flattened leaf-like stem that contains chlorophyll, and functions as a leaf.
crown	A plant structure at the root/stem interface from where leaves, stems and roots emerge.
diapause	A period of suspended development, especially during unfavourable environmental conditions.
ecotype	A distinct form or race of a plant or animal species occupying a particular habitat.
elytron (elytra plural)	Each of the two wing cases of a beetle.
epidermis	The outer layer of cells of a plant similar to a skin.

erineum (erinea plural)	A buckle or gall associated with dense matted white/brown hairs that distort leaf formation. They are often associated with the feeding of arthropods, especially mites.
floret	One of the small flowers making up a composite flower head, e.g. in a daisy or grass.
gall	A hard, spherical plant structure that often forms in response to the feeding by galling insects and mites or an infection by fungi.
gregarious	Living together in loosely organised communities.
inoculation	To implant a disease agent, e.g. spore, on a plant.
in vivo	Performed or taking place in a living organism or in the natural environment.
instar	A juvenile insect growth stage separated by moulting.
larva (larvae plural)	A juvenile stage of insects that have metamorphosis occurring between the egg and pupal stages. Larvae may be commonly called caterpillars, grubs or maggots.
meristem	The tissue (in most plants) that contains undifferentiated cells (meristematic cells), where growth can take place. Meristematic cells give rise to various organs of the plant and keep the plant growing.
mesophyll	The inner central tissue of a leaf.
nymph	A juvenile stage of insects that does not undergo metamorphosis. There may be several nymphal stages of development, each of which takes the general form of the adult (but lacks wings).
nursery site	A release site where agents have multiplied sufficiently to enable harvesting to occur without damage to the population.
ovule	The part of the ovary of seed plants that contains the female germ cell and after fertilisation becomes the seed.
parasitoid	An organism that lives in close association with its host at the host's expense.
pathogen	A disease producing organism. In biocontrol this is often a fungal disease, e.g. a rust.
pedicel	A small stalk bearing an egg or flower.
petiole	The stalk that joins a leaf to a stem.
polyphagous	Feeding on a wide range of plants.

pupa (pupae plural)	A phase between larval and adult stages where larval anatomical features disintegrate and adult features are constructed (metamorphosis).
pustule	A small raised spot or swelling.
receptacle	The thickened end part of a stem from which the flower organs grow.
rhizome	A modified underground main stem that sends out roots and shoots from its nodes.
root cortex	The outermost layer of the root of a plant.
rosette	A growth stage for some plants where leaves grow from one central point, usually at ground level. One or more flowering stems will often develop from the crown.
spermogonia	A cup shaped reproductive structure.
spores	A fungal reproductive cell.
sp. / spp.	An abbreviation of species, singular and plural.
stolon	A horizontal, above-ground 'plant stem' that forms off the main stem and takes root at points along its length to form new plants.
stomata	Tiny openings in leaf or stem tissue allowing gas exchange.
strain	A group of organisms whose characteristics are different in some way from others in that group, e.g. they may have been collected from different countries.
subsp.	Abbreviation of subspecies.
syn.	Abbreviation of synonym. Used to indicate a taxonomic name which has been superseded and is no longer valid.
telia	Structures used by rust fungus for the release of teliospores.
teliospore	A type of rust fungus spore that is often the surviving/overwintering stage and later on germinates to produce basidiospores.
thorax	The middle section of an insect between the head and abdomen.
uredinispore	A type of rust fungus spore that often re-infects the host. Often profuse and red/orange in colour.

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Other electronic resources

Resource	Web or Email address
Agriculture Victoria – Weeds	https://agriculture.vic.gov.au/biosecurity/weeds
Atlas of Living Australia (search for species names to see distribution maps, photos and information on weeds)	https://www.ala.org.au/
Australian Biocontrol Hub (biocontrol information and resources, agent release and spread data)	https://biocollect.ala.org.au/biocontrolhub
Australian Biological Resources Study – Glossaries	https://www.environment.gov.au/science/abrs/online-resources/glossaries
Australasian Virtual Herbarium	https://avh.chah.org.au/
Australian Plant Name Index	https://www.anbg.gov.au/apni/
CSIRO Weed Biocontrol Research	https://research.csiro.au/weed-biocontrol/
Flora of Australia	https://www.environment.gov.au/science/abrs/online-resources/flora-of-australia-online
Herbarium NSW	https://plantnet.rbgsyd.nsw.gov.au/
Herbarium Queensland	https://www.qld.gov.au/environment/plants-animals/plants/plants-weeds
Herbarium South Australia	https://www.environment.sa.gov.au/topics/Science/science-research/State_Herbarium
Herbarium Tasmania	https://www.tmag.tas.gov.au/collections_and_research/tasmanian_herbarium
Herbarium Victoria	https://www.rbg.vic.gov.au/science/herbarium-and-resources/
Herbarium Western Australia	https://florabase.dpaw.wa.gov.au/
iBiocontrol (search biocontrol agents globally and their target weed)	https://www.ibiocontrol.org/catalog/

other electronic resources

Resource	Web or Email address
Impact Evaluation of Weed Biological Control Agents	https://archive.dpi.nsw.gov.au/__data/assets/pdf_file/0008/348056/biocontrol-impact-evaluation-best-practice-guide.pdf
NSW Biocontrol Agent request form	Request the form from weed.biocontrol@dpi.nsw.gov.au
NSW DPI Grafton – Weed Biocontrol Mass-rearing Facility	https://www.dpi.nsw.gov.au/about-us/science-and-research/centres/grafton
NSW DPI – Orange Agricultural Research Institute – Weed Biocontrol Quarantine Facility	https://www.dpi.nsw.gov.au/about-us/science-and-research/centres/orange
NSW DPI – Weed Research Unit	https://www.dpi.nsw.gov.au/biosecurity/weeds/weed-control/weeds-research
NSW DPIE – Weeds	https://www.environment.nsw.gov.au/topics/animals-and-plants/pest-animals-and-weeds/weeds
NSW Weed Biocontrol Taskforce	https://www.dpi.nsw.gov.au/biosecurity/weeds/weed-control/biological-control/nsw-weed-biocontrol-taskforce
NSW WeedWise	https://weeds.dpi.nsw.gov.au/ https://www.dpi.nsw.gov.au/biosecurity/weeds/nsw-weedwise-app
Queensland Department of Agriculture and Fisheries – Weed Biocontrol Research	https://www.daf.qld.gov.au/business-priorities/biosecurity/invasive-plants-animals/research/current/landscape-protection-and-restoration
South Australia – Department of Primary Industries and Regions – Biological Control of Weeds	https://pir.sa.gov.au/biosecurity/weeds_and_pest_animals/weeds_in_sa/biological_control_of_weeds#:~:text=Biological control involves the release,roots
Weeds Australia (Weeds of National Significance manuals and weeds information)	https://profiles.ala.org.au/opus/weeds-australia

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