

## Establishment of *Arytainilla spartiophila* Förster (Hemiptera: Psyllidae), a new biological control agent for broom, *Cytisus scoparius*, in New Zealand

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### Abstract

Results of tests conducted with the psyllid *Arytainilla spartiophila* before it was introduced into New Zealand for biological control of broom, *Cytisus scoparius* (L.) Link, indicate that it is highly host specific, and unlikely to feed and develop on alternative host plants. Psyllids were reared under containment conditions and in the laboratory between 1991 and 1994 and first released in the field in New Zealand in 1993. From all releases made under a user-funded technology transfer programme, psyllids have established at 28% of sites, have been recovered from a further 7%, have failed at 3% of sites, and their fate at the remaining 62% of sites is unknown. From these releases psyllids are known to have established at 33 sites between Tapanui in Southland and Rotorua in the central North Island. The recorded rate of spread has generally been slow, but from one 1999 release psyllids spread 300–500 m from the original release point in 2 years.

**Keywords:** weed biocontrol, host specificity, insect rearing

### Introduction

The programme for biological control of broom (*Cytisus scoparius* (L.) Link) began in 1981. This weedy legume has potential to invade a substantially wider range than it occupied in New Zealand at that time (Williams 1981). A survey revealed that the fauna of broom in New Zealand was meagre in comparison with that of Europe where it is native, but that the specialist twig mining moth *Leucoptera spartifoliella* Hübner (Lyonetiidae) was common and widespread (Syrett 1993). Other specialist insects feeding on broom

in Europe are well known from studies by Waloff (1968) and through specifically targeted surveys for potential biological control agents (Syrett *et al.* 1999). The first insect released intentionally for biological control was the broom seed beetle, *Bruchidius villosus* F., in 1987 (Harman *et al.* 1996). *Arytainilla spartiophila* Förster (Hemiptera: Psyllidae) was accorded high priority as a potential biological control agent because large populations build up on broom in the UK causing substantial damage (Watmough 1968; D.J. O'Donnell, CABI Bioscience, UK, pers. comm., 1985) and the psyllid was reportedly highly host specific (Hodkinson & Hollis 1987).

Two species of psyllid are commonly found on broom in Europe, *A. spartiophila* and *Arytaina genistae* (Latreille) (Hodkinson & Hollis 1987). Eggs of *A. spartiophila* are inserted into the stems of broom from June to August in the UK. Females make trial incisions with their ovipositors beside those into which eggs are inserted, creating a characteristic scarred surface on the stem (Watmough 1968). The eggs are usually fully inserted into the slit, but sometimes are partly exposed. At the end of the summer the slit is covered by a waxy layer, and contains a minute, fully formed first-instar nymph with no sign of the egg. After a prolonged diapause of the first-instar larvae, young nymphs emerge the following March or April, developing through to adults during spring. Nymphs pass through five instars, feeding in buds and on the tender, actively growing parts of the plant. They produce a characteristic exudate of honeydew and fluffy white wax. Adults feed on the younger, green stems. There is only one generation per year. Because the second broom psyllid, *Arytaina genistae*, is multivoltine, completing 2–3 generations per year in the UK,

it was thought initially that it would be the better biological control agent. However, observations that *A. spartiophila* attained high populations more frequently than *Arytaina genistae* (Watmough 1968; D.J. O'Donnell, CABI Bioscience, UK, pers. comm., 1985), and records of *Arytaina genistae* from *Genista tinctoria* and *Chamaecytisus* spp. as well as from *Cytisus scoparius* (Hodkinson & Hollis 1987) indicated that *A. spartiophila* would be a better choice. This was confirmed by preliminary screening at CABI Bioscience, UK, in which *Arytaina genistae* was reared from plants other than broom (Fowler 1990).

Host plant records from the literature indicate that *A. spartiophila* is confined to *C. scoparius*. Hodkinson & Hollis (1987, p 42) report it on this plant in seven European countries. Two other *Arytainilla* species (*A. spartiicola* (Sülc) and *A. gredi* (Ramirez Gomez)) in the same group (*spartiophila*) are also reportedly confined to *C. scoparius*, but they have more limited distributions in France and Switzerland, and Spain, respectively. Known host records for other species groups within *Arytainilla* are all from the family Fabaceae (Hodkinson & Hollis 1987).

Experimental releases of broom psyllids were made in 1994 at 55 sites in Otago in order to measure the relative establishment success of releases of different numbers of individuals (Memmott *et al.* 2005). Results showed, as expected, that larger releases established better than small ones, but also that releases established from as few as two individual psyllids. An experiment to determine whether repeated releases increased the likelihood of establishment was abandoned when recoveries were made from all releases of both 100 and 300 individuals in the first year (unpublished data).

In this paper we report on tests conducted to measure the host range of the broom psyllid, and on the rearing, releases, and field recoveries made under a user-funded technology transfer programme.

## Materials and Methods

### *Host specificity tests*

Host-range tests were conducted in the UK with 59 plant species from 28 families. Test plants were selected according to criteria established by Zwölfer & Harris (1971) and Wapshere (1974).

Thirty test plant species (Table 1) were from the family Fabaceae in which *C. scoparius*, the host plant, is placed, including 10 native New Zealand plant species, plants of economic importance, and representatives of five clades (or nine tribes in an alternative traditional classification (Polhill & Raven 1981)). Categories of plants tested included species taxonomically closely related to the recorded host plant, New Zealand native and economic species not previously exposed to the psyllid, and plants known to be attacked by psyllids closely related to *A. spartiophila* (e.g., *Chamaecytisus palmensis*, which is recorded as a host of *Arytainilla dividens* Loginova (Hodkinson & Hollis 1987)).

All tests carried out with psyllid nymphs were 'no-choice' because adult females perform host selection during the oviposition phase. Three methods of foliage presentation were undertaken using plants with actively growing shoots that had newly bursting buds with young leaves, the parts of the plant favoured by psyllid nymphs. In the first method clip cages were placed on whole plants in pots. These cages were transparent beakers (80 × 100 mm) with fine mesh vents. The plant stem was inserted into a hole in the close-fitting plastic lid, which was sealed with a polyethylene foam bung and cotton wool. The clip cages were supported on the plants with canes and PVC tape. In the second and third methods either small plants in pots or cut shoots (placed upright with stems in small, sealed vials of water) were placed on white trays and individually covered by 150-mm diameter, clear plastic, pot propagator tops, partially vented to maintain humidity while preventing escape by psyllid nymphs. The method selected for each test plant species depended on the form in which plant material was available, with cut shoots used from very large plants growing outdoors. Newly emerged psyllid nymphs were transferred onto each caged part of the test plant with a fine camel-hair brush. Initially three nymphs were used per test plant but this was changed to five to allow for attrition, especially with first instar nymphs. Each set of experimental plants was set up with a matching set of controls. Plants were checked after a few minutes to ensure that nymphs had settled, and had not been damaged during the transfer. Nymphal survival, developmental stage, and extent of feeding were scored after 3–7 and

8–12 days. Extent of feeding was measured by visual estimation of honeydew output on a scale of 0–4, where amount is expressed as 1 = trace, 2 = small, 3 = moderate, 4 = large. Mean proportion of psyllids alive after 3–7 days, mean proportion alive after 8–12 days, and mean extent of feeding were estimated. For the *C. scoparius* controls, extent of feeding was compared among the three different methods of foliage presentation (whole plant, clip cage and cut shoots) using Kruskal-Wallis one-way analysis of variance. Similarly, survival of psyllids was compared by regression using generalised linear models. Analyses were performed using GenStat 8.1.

Because some feeding was recorded on *Robinia pseudoacacia* in no-choice tests with nymphs, an oviposition test was conducted with this plant. Twenty-five adult psyllids field-collected from *C. scoparius* were introduced into each of 12 fine mesh sleeves, 1 × 1.5 m in size. Four of the sleeves enclosed two 1–2-year-old *C. scoparius* plants in 4-litre plastic pots. Four of the sleeves enclosed two sections of *R. pseudoacacia* foliage, from plants growing out-of-doors, equivalent in size to the *C. scoparius* plants. Each of the remaining four sleeves enclosed both a *C. scoparius* plant and a section of *R. pseudoacacia*. The pots containing *C. scoparius* plants were raised up to prevent excessive bending of the *R. pseudoacacia* branches. All plants and branches had young, actively growing foliage, with opening buds, and were cleaned of other herbivores and psyllid predators before the test. The numbers of live and dead psyllids were recorded, and the number of eggs. Approximate numbers of eggs on *C. scoparius* plants were counted on live plants, using a microscope. Foliage of *R. pseudoacacia* was destructively sampled and sections were microscopically examined for the presence of eggs.

### **Release programme Importation**

Psyllid nymphs were first imported into a containment facility at Lincoln, New Zealand, in 1991. Although individuals from this shipment were successfully re-phased from Northern to Southern Hemisphere conditions and a second generation was reared and released from containment, none survived to be released in the field. A further seven shipments of psyllid nymphs arrived from CABI

Bioscience, Silwood Park, UK, over the next 3 years, the last one arriving in May 1994. All collections were made in the vicinity of Silwood Park, but only one shipment had the collection site identified, as Chobham Common.

### **Re-phasing to New Zealand environmental conditions**

The two factors critical to the successful re-phasing of psyllids to Southern Hemisphere conditions were to ensure sufficient time both at summer temperatures and day lengths to allow the first-instar larvae to develop to the diapausing stage, and under a 'winter' regime to allow diapause to be terminated. A regime was developed that compressed the psyllid's life cycle into approximately 6 months. Re-phasing was undertaken either in controlled laboratory conditions in the UK prior to shipment, or in containment in New Zealand after arrival. Similar methods were used in both cases.

Psyllid nymphs (a mixture of instars) were shipped to New Zealand on cut shoots of broom. On arrival they were transferred to fresh cut shoots in a flask of water. The shoots were placed in polyester mesh cages (0.75 × 0.75 × 0.5 m) supported by metal frames. Nymphs were transferred onto fresh shoots 2–3 times as they developed. Adult psyllids were transferred to a similar cage containing potted broom plants. Under a regime of 16:8 h Light: Dark and 20:15°C most nymphs reached the adult stage in 2–3 weeks. Potted plants with young, fresh shoots were selected to provide suitable oviposition sites for female psyllids. Plants were maintained at 16:8 h L:D and 20°C for 3 months, then temperature and day length were gradually reduced to 8:16 h L:D and 5°C over 2 weeks. After a 3-month period conditions were restored to 16:8 h L:D and 20:15°C over a 2-week period.

### **Rearing methods**

On successful completion of re-phasing, adult psyllids were removed from controlled conditions in containment or in the laboratory and given access to healthy, young broom plants. These plants were maintained in outdoor shade houses under natural light and temperature conditions. In order that plants were at a suitable stage for oviposition by female psyllids it was necessary to ensure that they had fresh, young growth, because females insert their eggs inside the soft, green

**Table 1. Extent of feeding and survival of broom psyllid nymphs, *Arytainilla spartiophila*, in no-choice tests on a range of non-target plant species and on the target plant broom, *Cytisus scoparius*.** (<sup>a</sup> clade groupings below Faboideae (= Papilionoideae) subfamily in Fabaceae (= Leguminosae) follow the format in Kajita *et al.* (2001), Wojciechowski *et al.* (2004), and Lavin *et al.* (2005) and are strongly supported. An alternative traditional classification at tribal level can be found in Polhill & Raven (1981); <sup>b</sup> p: tests using whole plants, cc: tests using clip cages, cs: tests using cut shoots; \*native New Zealand plants.)

Plant family/subfamily	Clade <sup>a</sup>	Plant species	No. of replicates (test type) <sup>b</sup>	Proportion alive after 3–7 days (± 1 SE)	Proportion alive after 8–12 days (± 1 SE)	Mean extent of feeding (± 1 SE)
<b>Plants in Fabaceae:</b>						
<b>Subfamily</b>	Genistoid s.l.	<i>Cytisus scoparius</i> (L.) Link	40 (p)	0.92 (± 0.02)	0.89 (± 0.02)	3.40 (± 0.11)
			<b>Faboideae</b>	17 (cc)	0.97 (± 0.02)	0.91 (± 0.03)
			19 (cs)	0.99 (± 0.01)	0.99 (± 0.01)	3.74 (± 0.10)
	Genistoid s.l.	<i>Chamaecytisus palmensis</i> (Christ) Bisby & K.Nicholls	5 (cc)	0	0	0.2 (0.2)
	Genistoid s.l.	<i>Genista hispanica</i> L.	5 (cc)	0	0	0
	Genistoid s.l.	<i>Laburnum anagyroides</i> Medikus	5 (cs)	0	0	0
	Genistoid s.l.	<i>Lupinus arboreus</i> Sims	5 (p)	0	0	0.2 (0.2)
	Genistoid s.l.	<i>Sophora microphylla</i> Aiton*	5 (p)	0	0	0
	Genistoid s.l.	<i>Spartium junceum</i> L.	5 (p)	0	0	0
	Genistoid s.l.	<i>Ulex europaeus</i> L.	5 (p)	0	0	0
	Milletioid	<i>Glycine max</i> (L.) Merr.	5 (p)	0	0	0
	Milletioid	<i>Phaseolus vulgaris</i> L.	3 (p)	0	0	0
	Hologalegina (Robinioid)	<i>Coronilla varia</i> L.	5 (cs)	0	0	0
	Hologalegina (Robinioid)	<i>Lotus corniculatus</i> L.	5 (p)	0	0	0.2 (0.2)
	Hologalegina (Robinioid)	<i>Robinia pseudoacacia</i> L.	10 (cs)	0	0	1.4 (0.31)
	Hologalegina (IRLC)	<i>Carmichaelia australis</i> R.Br. in J. Lindl.*	10 (p)	0	0	0
	Hologalegina (IRLC)	<i>C. compacta</i> Petrie*	4 (cc)	0	0	0
	Hologalegina (IRLC)	<i>C. crassicaulis</i> Hook. f.*	3 (p)	0	0	0
	Hologalegina (IRLC)	<i>C. glabrescens</i> (Petrie) Heenan*	2 (cc)	0	0	0
	Hologalegina (IRLC)	<i>C. monroi</i> Hook f.*	2 (cc)	0	0	0.5 (0.5)
	Hologalegina (IRLC)	<i>C. petriei</i> Kirk*	5 (cc)	0	0	0
	Hologalegina (IRLC)	<i>C. stevensonii</i> (Cheeseman) Heenan*	5 (p)	0	0	0
	Hologalegina (IRLC)	<i>C. torulosa</i> (Kirk) Heenan*	3 (p) 2 (cc)	0	0	0
	Hologalegina (IRLC)	<i>Clianthus puniceus</i> (G.Don) Sol. ex Lindl.*	5 (cc)	0	0	0

Hologalegina (IRLC)	<i>Colutea arborescens</i> L. Coste	5 (p)	0	0	0
Hologalegina (IRLC)	<i>Lathyrus pratensis</i> L.	5 (cc)	0	0	0
Hologalegina (IRLC)	<i>Lens esculentum</i>	5 (p)	0	0	0
Hologalegina (IRLC)	<i>Medicago sativa</i> L.	5 (p)	0	0	0
Hologalegina (IRLC)	<i>Pisum sativum</i> L.	5 (p)	0	0	0
Hologalegina (IRLC)	<i>Trifolium</i> sp.	5 (cc)	0	0	0.2 (0.2)
Hologalegina (IRLC)	<i>Vicia faba</i> L.	5 (p)	0	0	0
Hologalegina (IRLC)	<i>Wisteria sinensis</i> (Sims) Sweet	5 (cs)	0	0	0
<b>Subfamily</b>	<i>Acacia koa</i> Gray	5 (p)	0	0	0
<b>Mimosoideae</b>					
<b>Plants in other families:</b>					
<b>Alliaceae</b>	<i>Allium fistulosum</i> L.	5 (p)	0	0	0.2 (0.2)
<b>Apiaceae</b>	<i>Petroselinum crispum</i> (Mill.) A.W.Hill	4 (p)	0	0	0
<b>Asteraceae</b>	<i>Lactuca sativa</i> L.	5 (p)	0	0	0
<b>Betulaceae</b>	<i>Betula pubescens</i> Ehrh.	5 (cs)	0	0	0
<b>Boraginaceae</b>	<i>Echium vulgare</i> L.	5 (p)	0	0	0
<b>Brassicaceae</b>	<i>Brassica oleracea</i> L.	5 (p)	0	0	0
<b>Caprifoliaceae</b>	<i>Sambucus nigra</i> L.	5 (cs)	0	0	0
<b>Caryophyllaceae</b>	<i>Dianthus</i> sp.	5 (p)	0	0	0
<b>Chenopodiaceae</b>	<i>Spinacia</i> sp.	5 (p)	0	0	0
<b>Ericaceae</b>	<i>Calluna vulgaris</i> L.	3 (cc)	0	0	0
<b>Fagaceae</b>	<i>Quercus robur</i> L.	4 (cc)	0	0	0
<b>Grossulariaceae</b>	<i>Ribes rubrum</i> L.	5 (cs)	0	0	0
<b>Myrtaceae</b>	<i>Eucalyptus gunnii</i> Hook. f.	5 (cs)	0	0	0
<b>Onagraceae</b>	<i>Fuchsia</i> sp.	5 (cs)	0	0	0
<b>Passifloraceae</b>	<i>Passiflora edulis</i> Sims	5 (cs)	0	0	0
<b>Pinaceae</b>	<i>Pinus</i> sp.	5 (cs)	0	0	0
<b>Pittosporaceae</b>	<i>Pittosporum tenuifolium</i> Sol. ex Gaertn.*	5 (cs)	0	0	0
<b>Plantaginaceae</b>	<i>Hebe</i> sp. 1*	5 (cs)	0	0	0
	<i>Hebe</i> sp. 2*	5 (cs)	0	0	0
<b>Poaceae</b>	<i>Triticum aestivum</i> L.	5 (p)	0	0	0
<b>Polygonaceae</b>	<i>Rumex acetosa</i> L.	5 (p)	0	0	0
<b>Ranunculaceae</b>	<i>Clematis vitalba</i> L.	2 (p)	0	0	0
<b>Rosaceae</b>	<i>Malus pumila</i> Mill.	3 (cc)	0	0	0
	<i>Rosa</i> sp.	3 (cc)	0	0	0
<b>Rutaceae</b>	<i>Citrus</i> sp.	5 (cs)	0	0	0
<b>Salicaceae</b>	<i>Salix</i> sp.	5 (cs)	0	0	0
<b>Solanaceae</b>	<i>Lycopersicon esculentum</i> Mill.	5 (p)	0	0	0
<b>Theaceae</b>	<i>Camellia japonica</i> L.	5 (cs)	0	0	0
<b>Urticaceae</b>	<i>Urtica dioica</i> L.	5 (cs)	0	0	0

shoots. Fresh growth was also induced on older plants by maintaining them for several weeks in controlled conditions at 16:8 h L:D and 20°C. Care was taken to ensure that plants were free of other phytophagous insects and of psyllid predators, especially spiders, because it was important to maintain plants in healthy condition and free of predators for a number of months until nymphs emerged from the eggs and developed into adults.

In addition to cage rearing, psyllids were released onto broom bushes planted in a field plot at Lincoln so that a natural population would establish. Psyllids were then collected from this population for release in other parts of the country. They were transported on bouquets of broom twigs in clear plastic boxes lined with filter paper on the bottom. Bouquets were kept fresh by wrapping the ends of the stems in cotton wool secured with Parafilm®.

### Releases

From 1994 until 2001 adult psyllids were released at sites throughout New Zealand (Fig. 1) under a user-funded technology transfer programme. Most releases comprised approximately 1000 unsexed adult psyllids, although release size varied from 200 to 2000 individuals. Preliminary counts indicated an approximately equal sex ratio of males to females. In later releases the number was reduced to 500 individual psyllids per release.

Sites were revisited at least 1 year after release to determine whether the population had survived, and a second or further generation had been produced. To measure survival at a site two main methods of searching were used: examining plants visually and beating insects from foliage onto a beating tray. The number of individuals observed was recorded, whether they were adults or nymphs, and the time spent searching. As well as searching the area around the release point observers also examined bushes at increasing distances from the release point to record the furthest distance that a psyllid was found from the release point. This information was collected on a standard 'recovery sheet'. Most observers received some training from Landcare Research staff, were reminded of the appropriate time of year to check sites, and returned their data sheets to the organisation for compilation into a nationwide database.

## Results

### Host specificity tests

Results of the no-choice larval feeding tests are presented in Table 1. Psyllid nymphs did not survive for more than 3–7 days, or beyond the second instar, on any plants other than *C. scoparius*. Very little feeding occurred on any plants other than *C. scoparius*, although there was slight feeding on *R. pseudoacacia*. For the *C. scoparius* controls, there were no significant differences in the extent of feeding among the three different methods of foliage presentation ( $\chi^2_2 = 3.235$ ,  $p = 0.198$ ). There was significantly higher survival on cut shoots than on whole plants after 3–7 days ( $z = 2.20$ ,  $p = 0.028$ ) and after 8–12 days ( $z = 2.43$ ,  $p = 0.015$ ). Psyllids that were maintained beyond the end of the test period on *C. scoparius* controls successfully completed development to the adult stage.

Results from the test with *R. pseudoacacia* are presented in Table 2. Psyllids survived for no more than 8 days on *R. pseudoacacia* alone, but over 50% survived to the end of the test (15–21 days) in the presence of *C. scoparius*. Generally the number of dead psyllids recovered was low where *C. scoparius* was present, but in one replicate of the treatment with *C. scoparius* alone, 11 dead psyllids were found in a spider's web. Large numbers of psyllid eggs were counted on all *C. scoparius* plants regardless of whether *R. pseudoacacia* was present or not, but no eggs were found on *R. pseudoacacia* foliage and there was no sign of psyllid feeding.

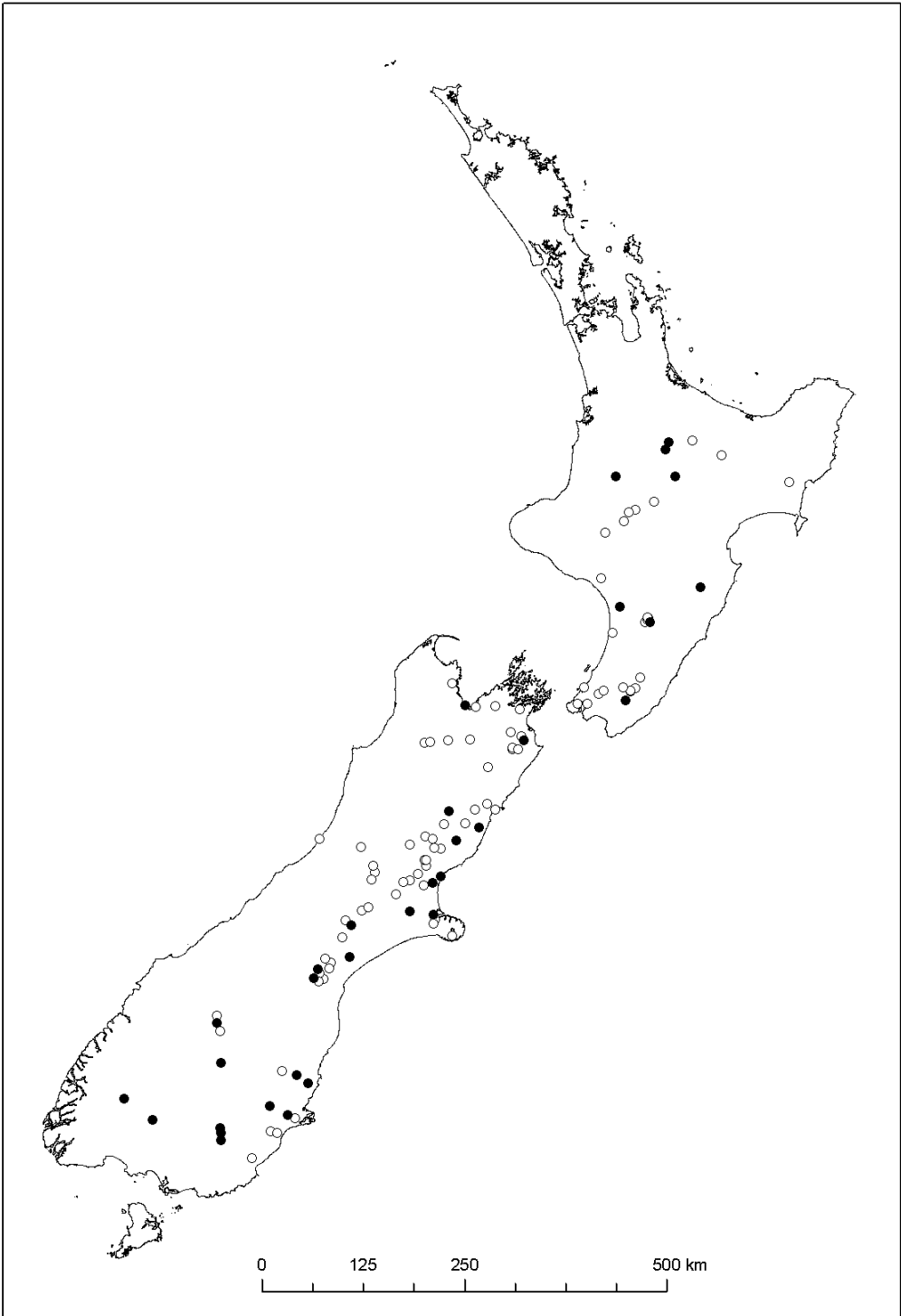
### Release programme

#### Importation

Psyllids from all shipments were released from containment following confirmation of identity and disease clearance. No attempt was made to keep progeny of different shipments separated, and mixed colonies were used for all rearing and subsequent releases.

### Re-phasing to New Zealand environmental conditions

Emerged nymphs were observed feeding and secreting droplets of honeydew within 5 weeks of the beginning of the changeover from 'winter' to 'summer' conditions under the re-phasing regime. Most nymphs emerged over a 2-week period and adults were first observed 1 week after nymphs were first observed on the plants.



**Figure 1.** Map showing locations of release sites for broom (*Cytisus scoparius*) psyllids (*Arytainilla spartiophila*) in New Zealand from 1994 to 2001. ● = sites where psyllids are known to have established, ○ = other release sites.

**Table 2. Survival and oviposition of broom psyllids, *Arytainilla spartiophila*, in no-choice and single-choice field tests with *Robinia pseudoacacia*.**

Test plant	No. surviving adults*	No. dead adults*	No. eggs*	
			<i>C. scoparius</i>	<i>R. pseudoacacia</i>
<i>Cytisus scoparius</i>	13.5 (4.9)	3.0 (5.3)	225 (150)	-
<i>Robinia pseudoacacia</i>	0	15.0 (6.7)	-	0
Both <i>C. scoparius</i> and <i>R. pseudoacacia</i>	15.0 (7.2)	0.5 (1.0)	200 (80)	0

\* mean (SE)

### Rearing methods

Between 1994 and 1999, about 60 000 psyllids were reared.

### Releases

Psyllids were released into the field at 117 sites from Southland to the central North Island (Fig. 1). For successful establishment it was necessary that broom bushes at the release site had adequate fresh, new growth. This was more important than chronological synchrony between stages of the insect and plant development. Some difficulties were experienced initially with transportation methods. Psyllids easily adhered to droplets of water that condensed on the sides of the container so care was taken with later releases that plenty of tissue paper was placed around the bouquets of broom to absorb condensation.

Recovery information from these sites is summarised in Table 3. Psyllids established at 28% of sites, they were recovered from an additional 7%, and aside from the 3% of sites where failure has been recorded the fate of the remaining 62% of releases is unknown. Psyllids have been recorded more than 100 m from the release point at only 4 sites of the 26 where they are known to have established. At 13 sites they were recorded within 20 m of the release point. Only one comment relating to insect damage was recorded, but this information is not specifically requested on the recovery sheet. At Mararoa, Mossburn, in Southland, very high numbers of psyllids were thought responsible for highly stressed and defoliated seedling plants (up to 0.5 m tall). Mature plants at this site exhibited reduced flowering (P. Ayson pers comm.).

### Discussion

Literature records and results of testing described here indicate that the risk of damage to non-target plants by *A. spartiophila* is low. However, increasing concern for potential negative impacts of intentionally introduced biological control agents (Howarth 2000, Louda *et al.* 2003) and a developing interest in improving host-range testing (Withers *et al.* 1999) provide an increased incentive to search for non-target impacts and determine whether they were successfully predicted by host-range tests. Systematic assessment of non-target impacts is increasingly becoming a required part of assessing the impact of biological control agents. However, *A. spartiophila* populations are only just reaching sufficiently high levels at a few sites for it to be worth looking for attack on non-host plants.

Rearing and widespread release of large numbers of biological control agents for a number of weed species has been achieved very effectively through user-funded technology transfer programmes (Hayes 2000). However, detection of surviving insect populations is more successful for those species that are conspicuous and easy to identify, such as cinnabar moth (*Tyria jacobaeae* (L.)) on ragwort (*Senecio jacobaeae* L.). Psyllids can be confused with leafhoppers by inexperienced observers, and even after training, observers from user organisations find this species difficult to identify with certainty. So far, recorded levels of damage have been low, so observers have been reliant on accurately identifying small insects that they have seen few times previously. This probably explains the discrepancy between recovery rates by experienced observers, which, for experimental releases in Otago and North

**Table 3. Status of field releases (2004) of *Arytainilla spartiophila* made from 1994 to 2001.**

Year of release	Psyllids established	Psyllids recovered	Status unknown	Release failed	Total number of releases
1994	2	1	1	1	5
1995	11	2	0	1	14
1996	4	3	3	1	11
1997	7	1	4	0	12
1998	1	0	1	0	2
1999	8	1	29	1	39
2000	0	0	33	0	33
2001	0	0	1	0	1
Totals	33	8	72	4	117

Canterbury after a single year, were 35% (average size of release approximately 50 individuals) and 100% (average size of release 200 individuals), respectively (unpublished data), and a recovery rate of only 29% from releases made under the technology transfer programme over several years (average size of release more than 500 individuals). Other factors that contribute to the failure to recover insects from sites are that sites are visited at inappropriate times, or not at all, through conflicting work commitments of staff from user organisations.

Psyllids are establishing readily in New Zealand (Memmott *et al.* 2005, unpublished data). Besides the main release programme described here, regional council staff are collecting from established field sites and transferring psyllids to new locations. A number of additional releases were made under a farmer-funded project. Psyllids are appearing in large numbers at many sites, but apparently still spreading only slowly. Although the degree of damage psyllids are inflicting on their host plants has been slow to reach that observed in Europe (Watmough 1968; D.J. O'Donnell, CABI Bioscience, UK, pers. comm., 1985) severely damaged plants have been reported from one site in 2002 from a psyllid population released in 1995. One reason for the slow appearance of damaged plants may be the ready availability of host plants in the vicinity of release sites. Another possibility is predation by the native mirid *Sejanus albisignatus* (Knight), which has been recorded previously on broom (Syrett 1993). It can be very common on

psyllid-infested broom at Lincoln and was seen to kill psyllids in a simple confinement trial (S.V. Fowler, unpublished data). Psyllids do not appear to have spread far, so numbers may not yet have built up sufficiently for them to reach the limit of their food resource and compete with each other to a level where they severely damage their food supply. As with a number of other weed biological control agents, particularly those that reproduce only once per year and attack perennial plants, it may take some years for the full impact of the psyllids to be realised.

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## References

- Fowler SV. 1990.** Report on weed work for New Zealand. International Institute of Biological Control, Silwood Park, Ascot, UK.
- Harman HM, Syrett P, Hill RL, Jessep CT. 1996.** Arthropod introductions for biological control of weeds in New Zealand, 1929–1995. *New Zealand Entomologist* 19: 71-80.
- Hayes LM. 2000.** Technology transfer programs for biological control of weeds – the New Zealand experience. In: *Proceedings of the X International Symposium on Biological Control of Weeds*, 4-14 July 1999, Montana State University, Bozeman, Montana, USA. (ed. NR Spencer) pp. 719-727. Montana State University, Bozeman, Montana.
- Hodkinson ID, Hollis D. 1987.** The legume-feeding psyllids (Homoptera) of the west Palaearctic Region. *Bulletin of the British Museum of Natural History (Entomology)* 56: 1-86.
- Howarth FG. 2000.** Non-target effects of biological control agents. In: *Biological Control: Measures of Success* (eds G Gurr, S Wratten) pp. 369-403. Kluwer Academic Publishers, Dordrecht, The Netherlands.
- Kajita T, Ohashi H, Tateishi Y, Bailey DC, Doyle JJ. 2001.** *rbcL* and legume phylogeny, with particular reference to Phaseoleae, Millettieae, and allies. *Systematic Botany* 26: 515-536.
- Lavin M, Herendeen PS, Wojciechowski MF. 2005.** Evolutionary rates analysis of Leguminosae implicates a rapid diversification of lineages during the Tertiary. *Systematic Biology* 54: 575-594.
- Louda, SM, Arnett AE, Rand TA, Russell FL. 2003.** Invasiveness of some biological control insects and adequacy of their ecological risk assessment and regulation. *Conservation Biology* 17: 73-82.
- Memmott J, Craze PG, Harman HM, Syrett P, Fowler SV. 2005.** The effect of propagule size on the invasion of an alien insect. *Journal of Animal Ecology* 74: 50-62.
- Polhill RM, Raven PH. (eds) 1981.** Advances in legume systematics, part 1. Royal Botanic Gardens, Kew, UK.
- Syrett P. 1993.** The insect fauna of broom (*Cytisus scoparius*) in New Zealand. *New Zealand Entomologist* 16: 75-83.
- Syrett P, Fowler SV, Coombs EM, Hosking JR, Markin GP, Paynter QE, Sheppard AW. 1999.** The potential for biological control of Scotch broom (*Cytisus scoparius*) (Fabaceae) and related weedy species. *Biocontrol News and Information* 20: 17N-34N.
- Waloff N. 1968.** Studies on the insect fauna on Scotch broom *Sarothamnus scoparius* (L.) Wimmer. *Advances in Ecological Research* 5: 87-208.
- Wapshere AJ. 1974.** A strategy for evaluating the safety of organisms for biological weed control. *Annals of Applied Biology* 77: 201-211.
- Watmough RH. 1968.** Population studies on two species of Psyllidae (Homoptera, Sternorhyncha) on broom (*Sarothamnus scoparius* (L.) Wimmer). *Journal of Animal Ecology* 37: 283-314.
- Williams PA. 1981.** Aspects of the ecology of broom (*Cytisus scoparius*) in Canterbury, New Zealand. *New Zealand Journal of Botany* 19: 31-43.
- Withers TM, Barton Browne L, Stanley J. (eds) 1999.** *Host Specificity Testing in Australasia: Towards Improved Assays for Biological Control*. Scientific Publishing, Department of Natural Resources, Brisbane, Australia.
- Wojciechowski MF, Lavin M, Sanderson MJ. 2004.** A phylogeny of legumes (Leguminosae) based on analysis of the plastid *matK* gene resolves many well-supported subclades within the family. *American Journal of Botany* 91: 1846-1862.
- Zwölfer H, Harris P. 1971.** Host specificity determination of insects for biological control of weeds. *Annual Review of Entomology* 16: 159-178.