



The establishment and spread of *Tamarixia triozae*, a parasitoid of the potato psyllid, in New Zealand

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Abstract The release of *Tamarixia triozae* (Burks) (Hymenoptera: Eulophidae), a parasitoid of the potato psyllid, *Bactericera cockerelli* (Šulc) (Hemiptera: Triozidae), resulted in the successful establishment of the parasitoid in New Zealand. The parasitoid was released at more than 30 sites by the final year of the three-year study throughout New Zealand. Its continued presence over the three-year study was confirmed in two regions (Hawke's Bay and Canterbury). At one site in Canterbury, the parasitoid was released only in the first summer of this study (Nov. 2017–Feb. 2018). It was recovered from potato psyllid infested African boxthorn (*Lycium ferocissimum* Miers) foliage in the second and third summers at this site, demonstrating the parasitoid's ability to survive over successive

winters. We found *T. triozae* parasitized nymphs at 24 sites of the 86 potato psyllid host plant sites surveyed within a 25 km radius of known release sites in Hawke's Bay. The parasitoid was found up to 24 km from the nearest known release site in Hawke's Bay. In Canterbury, the parasitoid was found up to 0.6 km from a known release site. Parasitism rates of 13.7–15.6% were estimated based on two post-release survey methods employed in this study. The parasitoid also feeds on psyllid nymphs so its establishment may lead to helping to reduce or delay potato psyllid populations from reaching damaging levels. Long-term monitoring is needed to determine the consequences of importing *T. triozae* on populations of potato psyllid.

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Introduction

The potato psyllid, *Bactericera cockerelli* (Šulc), is a pest of cultivated Solanaceae crops in North, Central and South America, Western Australia, Norfolk Island and New Zealand (EPPO 2022). It vectors a bacteria, *Candidatus Liberibacter solanacearum*, that can have a severe impact on plants (Hansen et al. 2008; Liefting et al. 2008, 2009). In New Zealand,

the psyllid and bacteria its vectors have cost affected industries hundreds of millions of dollars (Ogden 2011). Consequently, tolerance for the pest in commercial crops is very low, with heavy reliance on insecticides for control (Butler and Trumble 2012a; Munyaneza 2012; Martinez et al. 2015; Wright et al. 2017). In New Zealand, conventionally managed potato crops can receive up to 16 applications of insecticides per growing season (Wright et al. 2017). This has led to the development of insecticide-resistant populations in some regions of the world, although there are no reports of resistance in New Zealand (Dávila et al. 2012; Cerna et al. 2013; Chávez et al. 2015; Szczepaniec et al. 2019). Reliance on insecticides to manage this pest is not sustainable in the long term. A range of strategies that could help reduce populations of potato psyllid, including biological control, are needed to provide longer-term solutions.

The potato psyllid was first reported in New Zealand in 2006 and is now established in most regions across the country (Gill 2006; Teulon et al. 2009). In New Zealand, generalist predators such as *Micromus tasmaniae* Walker (brown lacewing), *Melanostoma fasciatum* (Macquart) (small hover fly), and *Coccinella undecimpunctata* L. (eleven-spotted ladybug) have been reported to predate potato psyllids in Solanaceae crops (MacDonald et al. 2010; Walker et al. 2011). There are no records of any parasitoid attacking this pest in New Zealand and a parasitoid with a narrow host range could provide more targeted control of the pest.

In Mexico and the USA, where the psyllid is a major pest of potato (*Solanum tuberosum* L.), tomato (*S. lycopersicum* L.) and pepper/capsicum (*Capsicum annuum* L.), *Tamarixia triozae* (Burks) is a solitary arrhenotokous ectoparasitoid that attacks *B. cockerelli* (Pletsch 1947; Lomeli-Flores and Partida 2002; Butler and Trumble 2012a; Butler and Trumble 2012b; Yang et al. 2015). It is small (0.7–1.0 mm in length) with a relatively wide host range of psyllid species including those in the Calophyidae, Psyllidae, and Triozidae families (Zuparko et al. 2011). Adult female *T. triozae* also feed directly on host psyllids to acquire protein for egg production (Rojas et al. 2015). In a laboratory study, females that lived an average of 31 days consumed an average of 181 nymphs (Cerón-González et al. 2014). While naturally occurring parasitism

rates reported for conventionally managed crops are relatively low (Butler and Trumble 2012a, 2012b; Yang et al. 2015), *T. triozae* predation and parasitism could contribute to mortality of overwintering psyllids (Johnson 1971; Butler and Trumble 2012a), helping to reduce or delay build-up of potato psyllid numbers the following growing season. This parasitoid has also been mass reared for augmentative release in commercial greenhouses in Canada and Mexico (McGregor 2013; Calvo et al. 2018). The potential benefits of this parasitoid as a component of integrated pest management led the primary industries in New Zealand most affected by the potato psyllid to obtain approval to import and release *T. triozae* (HSNO application APP201955, 2016).

Host testing was carried out as part of the approval to import and release the parasitoid in New Zealand on seven native and one introduced species of psyllid (Gardner-Gee 2012). *Arytainilla spartiophila* (Foerster), the non-native psyllid species tested, was introduced as a biological control agent for Scotch broom (*Cytisus scoparius* L.). The psyllid species tested as potential hosts for *T. triozae* were chosen because their distribution could put them in proximity to potato psyllid and its host plants (Gardner-Gee 2012), thus potentially exposing them to the parasitoid. In laboratory tests, *T. triozae* oviposited on two native psyllid species, albeit at lower rates than on potato psyllids (Gardner-Gee 2012). It did not oviposit on any of the other psyllid species tested, including the introduced *A. spartiophila*. However, adults emerged from one of the native species with reduced fecundity relative to those that emerged from potato psyllid (Gardner-Gee 2012).

The industry application was approved to import and release the parasitoid in 2016 (HSNO application APP201955, 2016). Following importation of the parasitoid from Mexico (BioBest Mexico, Koppert Mexico) into the Invertebrate Containment Facility at The New Zealand Institute for Plant and Food Research Limited (Plant and Food Research) Auckland, New Zealand, the Ministry for Primary Industries approved the release of the first generation. The aims of this study were to determine whether the parasitoid could establish self-sustaining populations in New Zealand and to evaluate how far it may have spread from selected release sites over the course of a three-year study.

Methods and materials

Tamarixia triozae importation

Between June and September 2017, approximately 1,000 *T. triozae* adults from Biobest Mexico and another 1,000 adults from Koppert Mexico were imported into the Plant and Food Research Invertebrate Containment Facility (Mt Albert, Auckland). The imported parasitoids were confirmed to be *T. triozae* by a taxonomist (Darren Ward, Manaaki Whenua Landcare Research, Auckland) and were confirmed free of any external fungal hyphae, following Ministry for Primary Industries importation requirements. Consequently, the first generation of *T. triozae* was approved for release out of containment in August 2017.

Tamarixia triozae gene sequencing

Ten female and ten male *T. triozae* were collected and preserved from the parasitoids released out of containment, for species confirmation through DNA sequencing and analysis. Genomic DNA was extracted in September 2017 using a modified cetyltrimethylammonium bromide (CTAB) method (Beard and Scott 2012). Samples were incubated overnight at 50 °C. After chloroform:IAA addition and agitation, samples were centrifuged for 13 min at 8,000 rpm. Pellets were resuspended in 20 µl EB (10 mM Tris–HCl pH 8.5). Appropriate extraction controls were included.

Two primer sets were selected for polymerase chain reaction (PCR) and subsequent sequencing: MtD6/MtD9 (Simon et al. 1994), target region within cytochrome c oxidase subunit 1 gene (COI) and ERCOII1F/EFCOII1R (Ardeh 2005), and target region within cytochrome c oxidase subunit 2 gene (COII). PCR reactions contained 12.5 µl KAPA 3G Plant PCR kit master mix (Roche), 1 µl of each primer (10 µM concentration), 8.3 µl water, 0.2 µl KAPA 3G Plant PCR kit taq (Roche) and 2 µl gDNA, for a total reaction volume of 25 µl. PCR amplification parameters were: 1×cycle 95 °C for 3 min, 2×cycles 96 °C for 30 s, 65 °C for 30 s and 72 °C for 30 s, 2×cycles 96 °C for 30 s, 60 °C for 30 s and 72 °C for 30 s, 2×cycles 96 °C for 30 s, 55 °C for 30 s and 72 °C for 30 s, 2×cycles 96 °C for 30 s, 50 °C for 30 s and 72 °C for 30 s, 2×cycles 96 °C

for 30 s, 45 °C for 30 s and 72 °C for 30 s, 30×cycles 96 °C for 30 s, 40 °C for 30 s and 72 °C for 30 s, 1×cycle 72 °C for 7 min. Appropriate extraction and no template controls were included. All PCR runs were carried out on a Gradient Palm-Cycler (Corbett Life Science, CG1-96). PCR products were visualised on 1% SB electrophoresis gels, including appropriate DNA reference ladders.

PCR products were purified using QIAquick PCR purification kit (Qiagen) following the manufacturer's instructions. The quantity (ng µl⁻¹) and quality (260/280 and 260/230 ratios) of purified PCR products were quantified using a NanoDrop spectrophotometer (ND-1000). Purified PCR products were subsequently sent to Macrogen (South Korea) for sequencing. All sequences obtained were aligned and analysed using Geneious bioinformatics software (Dotmatics) and compared with those reported by De León and Sétamou (2010) to ascertain haplotype.

Tamarixia triozae releases

Tamarixia triozae were reared on potato psyllid-infested capsicum (*C. annuum*) or tomato (*S. lycopersicum*) plants contained in insect rearing cages (0.5×0.5×0.5 m or 0.75×0.75×1.15 m, BugDorm, Taiwan) in temperature-controlled rooms at Plant and Food Research, Lincoln (22±1 °C, L:D 16:8 photoperiod), from August 2017 to June 2018. Adults from this colony were used for the first summer releases between November 2017 and February 2018. Staff at Lincoln University reared *T. triozae* on potato psyllid-infested plants of capsicum in small greenhouses and supplied adult parasitoids for the second summer releases from November 2018 to March 2019. A company (Bioforce, Auckland) that received *T. triozae* from the colony at Plant and Food Research (Mt Albert, Auckland) was supplying the parasitoid to commercial growers and home gardeners by the third summer (November 2019–March 2020). For the releases across all three years (November 2017–February 2020), adult parasitoids were collected from colonies into small 30 ml unvented vials using an aspirator (maximum of 250 adults per vial). A small smear of honey or droplet of 10% sugar solution was added to the lid of the container as food for the adults during transportation to release sites. Adults were released within 48 h following collection from colonies. Vials were attached to a potato psyllid host plant

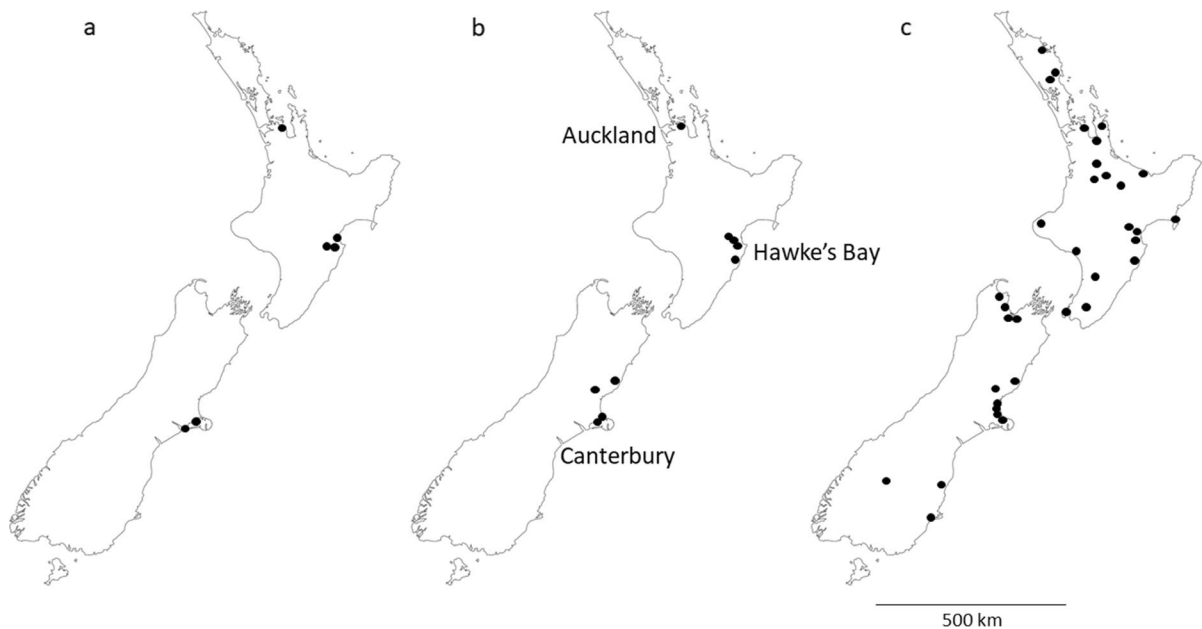


Fig. 1 Map of release of *Tamarixia triozae* in New Zealand. The black dots indicate the approximate locations where *T. triozae* were released: (a) in the first summer (November 2017–March 2018) parasitoids were released in Auckland (800 adults), Hawke's Bay (580 adults), and Canterbury (1,060 adults); (b) in the second summer (November 2018–March

2019) adult parasitoids were released by growers or home gardeners in Auckland (2,900), Hawke's Bay (3400) and Canterbury (1400); and (c) in the third summer (November 2019–March 2020) 20,600 adult parasitoids were released by growers or home gardeners throughout New Zealand

branch and lids were removed, allowing the adults to fly out. Alternatively, the lid was removed, and the base of the opened vial was gently tapped to encourage the parasitoids to fly out of the vial.

Auckland, Hawke's Bay and Canterbury regions were chosen for the first year of releases because they are important potato and/or field tomato cropping regions and growers were willing to have parasitoids released on their properties. The Auckland region in the north of the North Island has a cropping area of approximately 2,800 km², and subtropical climate (NIWA 2022). The Hawke's Bay region on the east coast of the North Island has approximately 8,700 km² of cropping land and a temperate climate (NIWA 2022). Canterbury, on the east coast of the South Island, has a cooler climate than Hawke's Bay (NIWA 2022) and cropping area is approximately 27,400 km².

In the first summer (November 2017–February 2018), releases of *T. triozae* in the Auckland region were carried out by a grower on vegetation surrounding their commercial tomato greenhouses. Plant and

Food Research personnel released the parasitoid in three sites in Hawke's Bay and two sites in Canterbury (Fig. 1a) at perennial potato psyllid host plants of African boxthorn. African boxthorn was introduced to New Zealand from South Africa as a hedge plant but is now considered a serious weed in some regions (Roy et al. 1998).

At all other new release sites in subsequent summers (November 2018–March 2019, November 2019–mid-March 2020) the parasitoid was released by growers, primary industry personnel or home gardeners (Fig. 1b, c). The growers released *T. triozae* in organically grown Solanaceae crops (potatoes, tomatoes, or tamarillos (*S. betaceum* Cav.)) or in surrounding non-crop vegetation.

Post-release surveys to determine survival at release sites

Surveys to determine whether the parasitoid survived at the release sites in Hawke's Bay and Canterbury were carried out across two summers (December

2018 and December 2019 or early January 2020) following their release the previous summer. Potato psyllid-infested host plant material was collected within 20 m from the first summer release sites before additional releases were made. Host plant material at these sites was predominantly African boxthorn, although some sites had potato or tomato foliage. A total of 20–24 African boxthorn, potato or tomato branches 15–30 cm long were collected from each release site. Branches from each release site were kept separately and identified by collecting date and locality. All collected foliage was sent to the Plant and Food Research entomology laboratory in Lincoln, Canterbury. In the laboratory, the cut ends of the branches were placed in 1.8 l containers with approximately 1 l water and placed in insect rearing cages (0.75×0.75×1.15 m, BugDorm, Taiwan) using a single cage for each site. Cages were maintained in a controlled temperature room at 22±1 °C, with a L:D 16:8 photoperiod and checked every 2–3 days, recording numbers of adult *T. triozae* and potato psyllid until no further adults were found.

At the Auckland property, potato psyllid host plants (tomato, eggplant (*Solanum melongena* L.), capsicum, black nightshade (*S. nigrum* L.)) in the vicinity of release sites were surveyed for parasitized nymphs for 60 min. Additionally, a tomato crop in the one of the greenhouses where the grower had reported seeing potato psyllid was surveyed and any potato psyllid nymphs found were collected. The nymphs were transferred to the Plant and Food Research entomology laboratory at Mt Albert (Auckland) and reared in small 4.2 l ventilated plastic containers (174 mm height×139 mm width×139 mm depth) with fresh leaf material added to support development of potato psyllid.

Post-release surveys to determine spread from known release sites

To evaluate the spread of the parasitoid from release sites, potato psyllid host plant sites were located within a 30 km radius of the first summer release sites in Hawke's Bay and Canterbury. Host plant material was examined, and the numbers of parasitized and non-parasitized potato psyllid found within 60 min were recorded. Parasitized potato psyllid nymphs become distinct from non-parasitized conspecifics once *T. triozae* pupae start to develop causing

the potato psyllid nymph to desiccate. Surveys to evaluate the spread of *T. triozae* were carried out in Canterbury and Hawke's Bay from January to April 2019 (summer to autumn), and in Hawke's Bay from December 2019 to mid-March 2020 (summer).

We calculated estimates of the relative abundance of the parasitoid in relation to the relative abundance of potato psyllid for all sites where potato psyllid were recorded. We used data from the two post-release survey methods that we employed (collected potato psyllid host plant material or direct field observations). We calculated percent parasitism rates using:

$$\left[\frac{T. triozae \text{ adults}}{T. triozae \text{ adults} + \text{potato psyllid adults}} \right] \times 100$$

Results

Tamarixia triozae gene sequencing and releases

The samples of *T. triozae* used for the gene sequencing analysis were shown to contain two haplotypes out of six haplotypes described by De Leon and Setamou (2010). Approximately 33,000 parasitoids were released across New Zealand over the three-year study (Fig. 1). In the first summer (November 2017–February 2018), a total of 800 adult parasitoids were released in Auckland, 580 in Hawke's Bay, and 1,060 in Canterbury. In the second summer, a total of 2,900 parasitoids were released in Auckland, 3400 in Hawke's Bay and 1400 in Canterbury (Fig. 1b). In addition to several new release sites in Hawke's Bay and Canterbury, the parasitoid was re-released at the same sites from the first summer except for the southern-most site in Canterbury. At this Canterbury site the parasitoid was only released in the first summer of the study (Fig. 1a). In the third summer (November 2019–mid-March 2020), at least 20,600 adult parasitoids were released by growers, primary industry personnel or home gardeners throughout New Zealand, who purchased parasitoids from Bioforce (Fig. 1c).

Post-release surveys to determine survival at release sites

The parasitoid was recovered from potato psyllid-infested African boxthorn foliage in the second

Table 1 The number of *Tamarixia triozae* adults (No. *T. triozae*) or potato psyllid adults (No. potato psyllid) to emerge from the release sites. Potato psyllid-infested African boxthorn

(Lycium ferocissimum) branches were collected in December 2018 or December 2019, the summers following release of the parasitoid in Hawke's Bay and Canterbury

Region (number of sites)	December 2018		December 2019	
	No. potato psyllid	No. <i>T. triozae</i>	No. potato psyllid	No. <i>T. triozae</i>
Auckland (1)	50	0	— ^a	—
Hawke's Bay (3)	1,616	273	537	30
Canterbury (2)	526	167	2264	162

At the Auckland release site, potato psyllid nymphs were collected from tomato foliage grown in a commercial greenhouse within 20 m of the location where the parasitoid had been released between November 2017 and February 2018

^aNo potato psyllid nymphs were found at the site

(January 2019) and third summer (January 2020) at the Canterbury release site where the parasitoid had only been released in the first summer (November 2017–February 2018), suggesting the parasitoid population had survived over two consecutive winters. *Tamarixia triozae* were recovered in the second (December 2018) and third (December 2019) summer from the three Hawke's Bay sites and second Canterbury site where it had been released in the first summer (Table 1). However, because the parasitoid was released numerous times in subsequent years at or near these first summer sites, we were not able to verify that the parasitized nymphs observed were from *T. triozae* that had survived the winter. In contrast to Hawke's Bay and Canterbury, no parasitoids were seen or recovered from potato psyllid collected from the Auckland site over the course of the study.

Post-release surveys to determine spread from known release sites

We surveyed 99 non-release sites for the presence of *T. triozae*. Most non-release sites were in Hawke's Bay (86 sites). In Hawke's Bay, parasitized potato psyllid nymphs were recorded on apple of Peru (*Nicandra physaloides* L.), thornapple (*Datura stramonium* L.), African boxthorn and conventionally managed potato and field tomato crops (Table 2). Of the 86 non-release sites surveyed in Hawke's Bay, potato psyllids were found at 38 sites, with the parasitoid recorded at 24 of these sites. In Hawke's Bay, *T. triozae* was found up to 24 km from the nearest known release site.

In Canterbury, parasitized potato psyllids were found at two of the 13 non-release sites surveyed.

Sites were 0.05 to 6.5 km from the nearest release site, except one site located 21 km from the nearest known release site. Parasitized potato psyllid nymphs were found a maximum of 0.6 km from the nearest release site in Canterbury (Table 2).

We estimated an average parasitism rate of 13.7% (0–33% min.–max. range) from insects reared from field-collected host plant material. From the field scouting method, we estimated an average percent parasitism of 15.6% (0–100% min.–max.) (Table 2).

Discussion

The release of *T. triozae* in Hawke's Bay and Canterbury over three years resulted in the successful establishment of the parasitoid in these two regions in New Zealand. However, the long-term survival of the parasitoid and its contribution to suppressing potato psyllid populations requires further study. The establishment of biological control agents depends on the agent dispersing from the release locations, locating overwintering sites and dispersing from such sites to locate and exploit host/prey (Corbett and Rosenheim 1996). Where possible, Plant and Food Research personnel chose release sites where they knew populations of potato psyllid were present year-round and exposure to insecticides would be minimal. Similarly, Parra et al. (2010) proposed release sites for the citrus psyllid *Diaphorina citri* Kuwayama parasitoid, *Tamarixia radiata* (Waterston), where exposure to insecticides could be minimised or avoided (e.g., abandoned citrus groves and areas with *Murraya paniculata* L. host plants). The availability of the parasitoid for purchase by the public may increase the likelihood of it

Table 2 The number of non-parasitized potato psyllid (No. potato psyllid), or parasitized nymphs or nymph remains with exit holes (No. *T. triozae*) observed in a 60 min period on potato psyllid host plants at non-release sites

Region	Distance to nearest known release site (km)	Host plants surveyed	No. potato psyllid	No. <i>T. triozae</i>
Hawke's Bay	0.3	<i>Solanum lycopersicum</i> ^a	162	10
	0.5	<i>S. tuberosum</i> ^a	< 20	1
	1.0	<i>S. lycopersicum</i> ^a	6	4
	1.2	<i>Lycium ferocissimum</i>	304	2
	1.7	<i>S. tuberosum</i> (organic) ^b	> 2,000	300
	2.8	<i>L. ferocissimum</i>	2	2
	2.8	<i>L. ferocissimum</i>	128	16
	3.0	<i>Datura stramonium</i>	6	1
	3.1	<i>Nicandra physalodes</i>	34	4
	3.5	<i>L. ferocissimum</i>	10	2
	3.7	<i>D. stramonium</i>	35	3
	4.0	<i>L. ferocissimum</i>	114	35
	4.8	<i>D. stramonium</i>	17	1
	5.0	<i>S. lycopersicum</i> ^a	39	37
	5.0	<i>S. lycopersicum</i> ^a	88	78
	5.0	<i>D. stramonium</i>	19	1
	5.0	<i>D. stramonium</i>	64	11
	5.5	<i>L. ferocissimum</i>	44	1
	6.2	<i>N. physalodes</i>	23	8
	6.7	<i>L. ferocissimum</i>	100	9
	6.8	<i>D. stramonium</i>	154	15
	16.7	<i>D. stramonium</i>	5	1
	17.0	<i>S. lycopersicum</i> ^a	2	2
	24.4	<i>D. stramonium</i>	32	1
Canterbury	0.06	<i>L. ferocissimum</i>	20	8
	0.6	<i>L. ferocissimum</i>	40	1

The tomato or potato crops surveyed included those managed using standard industry best practice (conventional) or certified organic. Other host plants surveyed included African boxthorn (*Lycium ferocissimum*), thornapple (*Datura stramonium*) and apple of Peru (*Nicandra physalodes*). Sites surveyed are listed according to increasing distance to the nearest known release site

^aConventionally managed crops

^b*Tamarixia triozae* adults emerged from potato psyllid nymphs reared in the laboratory on host plant foliage collected from the field

becoming established in other regions in New Zealand if it has not done so already. The commercial availability of the parasitoid also enables growers of Solanaceae crops to use *T. triozae* in augmentative releases to help manage potato psyllid populations (Veronesi et al. 2021).

The parasitoid was found throughout the Hawke's Bay area surveyed, and in several cases, many kilometres from the nearest known release site (Table 2). In contrast, parasitized potato psyllid nymphs were only found on African boxthorn within 0.6 km of the two release sites surveyed in Canterbury. Potential

potato psyllid crop and non-crop host plants were much more prevalent in Hawke's Bay compared with Canterbury. This limited our ability to find potential potato psyllid host plant sites for the post-release surveys in Canterbury. The presence of potato psyllid and their host plants between release and non-release sites could aid *T. triozae* in its movement between the sites. However, we cannot discount the potential for human-assisted movement. We did not know the locations where parasitoids were released by home gardeners, or whether parasitized potato psyllid in potato psyllid-infested host plant material was

transferred to other sites (e.g., green waste from home gardens). Either activity may help explain the observation of the parasitoid many kilometres away from known release sites in Hawke's Bay.

The estimated average parasitism rates of 13.7–15.6% from the two methods reported in this study were similar to those reported in field studies from the USA and Mexico (Pletsch 1947; Lomeli-Flores and Partida 2002; Butler and Trumble 2012b). The field survey method used in the present study resulting in an estimated 15.6% parasitism only recorded potato nymphs that were obviously parasitized and missed nymphs with eggs or very young immature stages of *T. triozae*. However, the method of rearing insects on field-collected material through to adults in the laboratory did account for these parasitoid life stages. Parasitism rates of up to 85% have been reported from Mexico, although the implications for the psyllid population were not described (Lomeli-Flores and Partida 2002; Bravo and López 2007). A thorough investigation of psyllid and parasitoid populations dynamics in the future is needed to better understand the impact this parasitoid could have on potato psyllid populations. At the very least, its introduction to New Zealand may help contribute to reducing or delaying potato psyllid populations from reaching damaging levels each summer, which may in turn reduce the number of insecticides required for control on commercial crops.

The presence of *T. triozae* in conventionally managed potato and tomato crops (Table 2) suggests the parasitoid was able to parasitize hosts potentially exposed to insecticides. Conventionally managed potato crops in New Zealand can receive up to 16 applications of insecticides per annum (Wright et al. 2017). The survival of the parasitoid when insecticides are used may have been due to the psyllid host escaping exposure (e.g., poor spray coverage, location on host plant limited exposure) or could depend on the insecticide. Some insecticides are less toxic to the parasitoid (Liu et al. 2012). Further studies are needed to determine the compatibility of insecticides with *T. triozae*.

The sequencing results indicated the parasitoids tested contained two of the six haplotypes reported for *T. triozae*. The consequences of this are unclear since the individual *T. triozae* analysed by De León and Sétamou (2010) were collected from a single population at Weslaco, Texas, USA. Geographical

genetic differentiation between populations in natal and novel environments has been reported for *Aphidius ervi* (Haliday) imported into Chile to help control a range of aphid pest species (Zepeda-Paulo et al. 2016). In the case of *A. ervi*, only 271 individuals were imported into quarantine for multiplication from 1976 to 1981 (Zepeda-Paulo et al. 2016). Despite the evidence of limited genetic diversity in the *A. ervi* populations in Chile, the parasitoid has proven to contribute to the control of important target pest species (Gerding and Figueroa 1989; Starý et al. 1993; Sepúlveda et al. 2021). In the present study, we imported 2,000 individuals. However, these individuals were obtained from commercial rearing facilities, which may in turn experience limited genetic diversity. Any potential consequences of low genetic diversity in populations of *T. triozae* in New Zealand can only be determined with long-term monitoring of both genetic diversity within the populations and parasitism rates.

The estimated rate of parasitism from post-release surveys that involved collecting foliage from the field could be affected by where the foliage was collected, and how much was collected. Foliage was collected only where potato psyllid nymphs were present and where it was within reach of the collectors, and the quantity of foliage collected was limited. The estimate from the field scouting method relied on rapidly distinguishing parasitized potato psyllid nymphs when they become more distinctive in colour relative to non-parasitized conspecifics. This occurred when *T. triozae* pupae started to develop causing the potato psyllid nymph to desiccate. Potato psyllid nymphs parasitized with eggs or developing larvae of *T. triozae* are more difficult to distinguish from non-parasitized nymphs and can require dissection to determine the presence of the parasitoid egg or larvae. While the mean estimates observed in this study are within the previously reported parasitism rates of less than 20%, it is likely that rates have been underestimated (Pletsch 1947; Butler and Trumble 2012b).

The long-term consequences of importing *T. triozae* on populations of potato psyllid and populations of non-target psyllid species warrant further study. If *T. triozae* can be shown to contribute to the suppression of potato psyllid populations, especially where growers can use the commercial product for augmentative releases, this may add another tool for management practices that lessen our reliance on insecticides.

If the parasitoid is shown to have little or no impact on non-target psyllid species, this would further validate the regulatory approach taken in New Zealand to ensure such risks are mitigated. Further knowledge of the genetic composition of the introduced parasitoid populations could also help to elucidate the consequences on the fitness of the parasitoid in New Zealand as well as inform future programmes that import biological control agents.

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Data availability The datasets from the current study are available from the corresponding author on request.

Declarations

Conflict of interest All authors declare that they have no competing interests either directly or indirectly related to the work submitted for publication.

Research involving human participants and/or animals No humans and/or animals were used in this study that required informed consent or submission to animal welfare committee for evaluation.

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