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THE WĒTĀ

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The Wētā is the bulletin of the Entomological Society of New Zealand and, like the society's journal *New Zealand Entomologist*, promotes the study of the biology, ecology, taxonomy and control of insects and arachnids in an Australasian setting. The bulletin provides a medium for both amateur and professional entomologists to publish records, observations, news, views, and the results of research projects.

Details for the submission of articles are given on the inside back cover.

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Cover illustration

Forest Ringlet *Dodonidia helmsii.*
(Photo: Aman Hunt https://www.inaturalist.nz/observations/378328834).

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J. K. Fox Award, Amateur and Citizens Scientists

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Each year, the Entomological Society of New Zealand (Ent Soc NZ) awards a number of grants to society members and non-members, including students, independent researchers, community groups or individuals. One of the awards given by the society is the K.J. Fox Award. The purpose of this award is to assist amateur and non-funded members of the Ent Soc NZ to attend the society's annual conference.

The K.J. Fox memorial fund was established by The Entomological Society of NZ following K.J. Fox's death in 1985 and the first awards were made in 1988. Applications for the award are called for on an annual basis. The Society's committee evaluates applications for the award based on the applicant's financial situation and the potential benefits of attendance for both the individual and the society. The amount awarded can vary, with distance travelled to the conference as well as conference fees taken into account. Applicants must be current members of the society. Applications for the award usually close a few months before the annual conference, with successful applicants notified in advance, so recipients can plan their travel in a timely manner (https://ento.org.nz/awards-and-grants/k-j-fox-awards/).

In 2025, I was one of the recipients of the K.J. Fox Award, which enabled me to attend the 5th Combined Australian and New Zealand Entomological Societies Conference held in Canberra. This conference marked the 60th anniversary of the establishment of the Australian Entomological Society, the 56th Annual Conference of the Australian Entomological Society and the 73rd Ent Soc NZ Conference. I am very

grateful to have been able to attend this conference, which allowed me at the same time to visit the Australian National Insect Collection (ANIC). Together with fellow dipterist and lead invertebrate curator Dr Julia Kasper, from the Museum of New Zealand Te Papa Tongarewa (Wellington) we were shown the new ANIC facilities (opened 14 August 2025) and the insect collection (Fig. 1) by Dr. Keith Bayless. True to my passion I was also able to view some Tachinidae specimens, which were exiting to see (Figs. 2 & 3).

Figure 1: Insect units at the Australian National Insect Collection, Canberra

Figure 2: Australian Tachinidae

Figure 3: *Trichopoda* specimen

The combined entomological conference was held from 1– 4 Dec. 2025 in the Marie Reay Teaching Centre at the Australian National University in Canberra. The conference had a high attendance (290 Registrations) (Fig. 4) with well over 100 presentations spread over 4 concurrent sessions and 25 posters.

Figure 4: Conference photo of the 60th anniversary of the establishment of the Australian Entomological Society, the 56th Annual Conference of the Australian Entomological Society and the 73rd Ent Soc NZ Conference

Presentations covered a wide range of topics, *e.g.* Invasive Species and Biosecurity, Ecosystem Services, Ecosystem Services, Insect-Plant Interactions and their effects on Biodiversity, Biosecurity and Bio-Agroecosystems, Insect-Plant Interactions and their effects on Biodiversity, Biosecurity and Bio-Agroecosystems, Insect-Plant Interactions and their effects on Biodiversity, Biosecurity and Bio-Agroecosystems, Pests and Beneficials, Dung, Death, and Decay – Carrion

Related Insects and Forensic Entomology, Ecology and Evolution, and Social Insects.

Of particular interest to me were of course presentations covering Diptera. Such as: "A Revision of the cryptic Australasian cluster flies (Diptera: Polleniidae)" by Liam Foley (PhD Student University of Wollongong); "A Revision of New Zealand's Small Headed Flies (Acroceridae)" by Shaun Thompson (PhD student Lincoln University and Invertebrate Collections Technician Museum of New Zealand Te Papa Tongarewa; "Population Dynamics of Glow-Worms in NZ and Tasmania" by Dr Julia Kasper (Invertebrate Curator Museum of New Zealand Te Papa Tongarewa) and "The taxonomy and phylogeny of the New Zealand Endemic Wasp Genus *Degithina* Cameron, 1901 (Hymenpotera: Ichneumonidae)" by Simon Crampton (Postgraduate student Lincoln University), just to name a few. The presentation by Dr Juanita Rodriguez (Senior Research Scientist ANIC-CSIRO) stuck out for me, because it is observations like her's that make me in awe of the world we live in and make entomology so interesting. Her presentation covered "The first known troglomorphic, eyeless spider-wasp (Hymenoptera: Pompilidae) from the Nullarbor Caves, Western Australia". This pompilid wasp is eyeless and brachypterous (without wings) unlike any member of the family known globally. I presented "A Revision of the New Zealand tachinid subfamily Phasiinae and rediscovery of some lost New Zealand Diptera types". The complete conference programme can be viewed at https://consec.eventsair.com/australian-entomological-society-56th-annual-conference/conference-program

As mentioned previously, my attendance at the conference was made possible thanks to contributions to my travel and conference cost by the K.J. Fox award of the Ent Soc NZ. ~~Little did I know~~I knew little about the collector K.J. Fox when I described the species *Zorion tarakiniensis* Schnitzler, 2005 (Schnitzler & Wang 2005, see also Schnitzler 2001) (Fig. 5) on hand of cerambycid (long horn beetle) specimens which K.J. Fox had collected from the western slopes of Mt Taranaki, New Plymouth, NZ.

Nor did I know anything about K.J. Fox when I applied for the award. I assume that I am not alone here and many Ent Soc NZ members do not know who Kenneth John Fox (also known as Ken) was.

Figure 5: *Zorion guttigerum*. (image: commoncopper 2023, https://www.inaturalist.nz/photos/333918835, accessed 30 April 2026)

Ken was an amateur NZ entomologist who migrated from London, U.K. He became an active member of the Ent Soc NZ and an active, enthusiastic insect collector, primarily from around new Plymouth and Mt Taranaki. He also observed and recorded transoceanic insect migrations, often with a focus on moth migrations to New Zealand. Ken published his findings in several New Zealand journals (see attached bibliography). But what made Ken stand out from the majority of NZ entomologist was that he was self-taught, not trained as a professional entomologist. However, he studied

medicine and after marrying New Zealander Margret Bull, they moved to NZ and he worked as a general partitioner in Manaia, South Taranaki. He always maintained his passion in entomology and became NZ's Ent Soc first amateur vice-president. Had it not been for his untimely death in January 1986; he might have become the society's president (Dugdale 1987, Hornabrook 1986). I believe Ken was not only the first, but also the only amateur holding the position of vice-president of the NZ Ent Soc.

Also, Ken was a great promoter of amateur entomology. Ken bemoaned the lack of amateur entomologists in NZ compared to other countries and called on the Ent Soc NZ for better support and encouragement of junior and amateur entomologists (Dugdale 1987, Fox 1971). Ken also attributed the lack of amateur entomologists to the fact that there was so little material available to identify species, which he thought would discourage pursuing an interest in entomology (Fox 1971).

Since Ken's contribution was written, means of communication have vastly improved. I recall one plenary speaker presentation at the Canberra Conference by Associate Professor Tanya Latty covering the seemingly decline of Australian Christmas Beetles (https://consec.eventsair.com/australian-entomological-society-56th-annual-conference/conference-program). In short, to address shortcomings of scientific knowledge about Christmas Beetle population trends, ecology and habitat needed the national citizen science project "Christmas Beetle Count", was launched, which invited the public to submit sightings of Christmas beetles to *iNaturalist* (see https://www.inaturalist.org/projects/christmas-beetle-count and https://invertebratesaustralia.org/christmas-beetles). At the time I am writing this article 10,986 observers submitted 25,684 records of 40 species of Christmas beetles since the beginning in 2020 (https://www.inaturalist.org/projects/christmas-beetle-count accessed 07 May 2026). Amongst other records, new sightings of species that had not been observed in decades were reported.

Perhaps citizens science is in part a new form of amateur science and the platform *iNaturalist* a new tool to assist science research. Translating this into the NZ entomology context, being an amateur entomologist doesn't need to entail physical collecting of insects. Contributions to recording species, for example, on *iNaturalist* by citizen scientists, can contribute to increase knowledge of seasonal cycles, species locations, population dynamics, etc. To date when exploring recordings under the term Arthropods (Phylum Arthropoda) within NZ at *iNaturalist*, there are 925,683 observations covering 6,890 species by 24,843 observes and 8,772 identifiers (a lot more than professional entomologists) (https://www.inaturalist.nz/observations?place_id=6803&taxon_id=47120 accessed 07 May 2026).

Acknowledgements

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First host record of the parasitoid wasp *Meteorus pulchricornis* (Braconidae) from larvae of the Forest Ringlet butterfly *Dodonidia helmsii* (Nymphalidae)

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Observation

The Forest Ringlet butterfly (*Dodonidia helmsii* Butler, 1884) is an "Threatened - Nationally Vulnerable" endemic New Zealand nymphalid associated with native forest habitats (Hoare *et al*. 2026). At Mahakirau Forest Sanctuary, larvae develop on cutting sedges, namely *Gahnia pauciflora* and *G. setifolia*. Elsewhere, *G. procera* and other monocots, especially *Chionochloa conspicua* have also been recorded as host plants (Barker & Smerdon 2024). Larvae are slow-growing and relatively sedentary, typically remaining on a single host plant for the duration of their development.

During routine monitoring of Forest Ringlet larvae at Mahakirau Forest Sanctuary, Coromandel Peninsula, a parasitoid cocoon was observed attached to a third-instar *D. helmsii* larva (Figs. 1-2). The cocoon was suspended beneath the larva on the underside of a *G. pauciflora* leaf by a silk thread characteristic of parasitoids in the genus *Meteorus* (Braconidae).

An exit wound was clearly visible on the lower abdominal flank of the host larva (Fig. 2). The larva and cocoon (Fig. 2) were collected on 17 February 2026. Fifteen days later, on 4 March 2026, an adult female parasitoid wasp (Fig. 4) emerged from the cocoon. A second, unsuccessful cocoon subsequently formed within the collection bag, suggesting that a single Forest Ringlet larva may host two parasitoid larvae.

Morphological features of both the adult wasp (Fig. 3) and cocoon (Figs. 1-4) are consistent with *Meteorus pulchricornis* (Wesmael 1835), a solitary koinobiont endoparasitoid known to attack a wide range of Lepidoptera larvae (see Walker & Martin 2018). Identification was supported by the distinctive suspended "basket-type" cocoon typical of the genus *Meteorus*.

Figure 1: *Meteorus pulchricornis* cocoon attached to *Dodonidia helmsii* larva (lateral view).

Figure 2: *Meteorus pulchricornis* cocoon suspended beneath *D. helmsii* larva with clearly visible exit wound on the larva (ventral view).

Figure 3: Emerged *Meteorus pulchricornis* female with cocoon.

Figure 4: *Meteorus pulchricornis* cocoon with lid cut open.

A second cocoon of identical structure was subsequently found at a separate location within Mahakirau Forest Sanctuary associated with a dead *D. helmsii* larva. An adult female emerged from the cocoon 14 days after collection (25 February-10 March 2026) by cutting open a

characteristic "lid" at the distal end (Fig. 4). In both cases the host larvae were determined to be third instar, indicating that *M. pulchricornis* is capable of utilising mid-stage Forest Ringlet larvae as hosts.

Discussion

Meteorus pulchricornis is a cosmopolitan solitary koinobiont endoparasitoid of Lepidoptera larvae and is recognised by its distinctive suspended cocoon (Shaw & Huddleston 1991). It is a highly polyphagous species and is considered adventive in New Zealand, having established following accidental introduction (Berry & Walker 2004; Berry 2007; Walker & Martin 2018). It was first detected in 1996 (Berry 1997) and is now widely distributed, with records ranging from Northland to Central Otago (Berry & Walker 2004).

A wide range of moth families have been recorded as hosts (Berry & Walker 2004; Schnitzler *et al.* 2004; Walker & Martin 2018), but the present observation appears to represent the first documented case of a New Zealand butterfly (Rhopalocera: Nymphalidae) being used as a host.

An experimental study by Chhagan *et al.* (2008) looking at the developmental biology of *M. pulchricornis* on larvae of the Painted Apple Moth (*Teia anartoides*) found that developmental times were greatly influenced by ambient temperatures. Development time from egg to adult ranged from as little as 16 days at 25ºC, to as long as 64.5 days at 13ºC. The duration of egg-to-pupa stages was roughly equal to, but a few days longer than pupal development. Given that the pupal stage of the present study was at least 15 days, complete development from egg to adult was probably c.35 days or greater. This suggests that oviposition probably took place in second instar *D. helmsii* larvae, with the parasite exiting from the larva during third instar. Chhagan *et al*. (2008) showed that of the four instars of *Teia anartoides*, there was a strong preference for second and third instar host larvae being used by *P. pulchricornis*.

Although parasitoids are recognised as potential pressures on *Dodonidia helmsii* (e.g. Gibbs 1980; Barker & Smerdon 2024) confirmed host associations remain limited. The observations reported here provide direct evidence of *Meteorus pulchricornis* parasitising *Dodonidia helmsii*. The discovery of multiple specimens within the sanctuary suggests this interaction may occur widely within the local population, and as *M. pulchricornis* is widely distributed in New Zealand, other *D. helmsii* populations may be similarly affected.

This record contributes a new parasitoid association for *D. helmsii* and adds to current knowledge of ecological pressures affecting this rare endemic butterfly. Further monitoring will help determine how frequently this basket-cocoon parasitoid utilises Forest Ringlet larvae as hosts and whether it represents a significant mortality factor within populations.

Acknowledgements

We thank the volunteers supporting ongoing biodiversity monitoring at Mahakirau Forest Sanctuary. Field observations contributing to this note were collected during broader research into the ecology and conservation of the Forest Ringlet butterfly. All images: Sara Smerdon © 2026.

https://linktr.ee/Mahakirau

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Substrate-dependent colour variation along the Kāpiti–Taranaki coastline and biological notes on *Actizeta albata* (Coleoptera: Tenebrionidae)

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This study investigated whether colouration of the beetle *Actizeta albata* Pascoe, 1875 matches the sand substrate along the Kāpiti–Taranaki coastline of the North Island. In *A. albata*, colouration may vary from mostly white to mostly black. Specimens were collected from the black sand beaches of the Taranaki–Whanganui region and grey sand beaches of the Kāpiti region. The extent of dark patterning on each specimen was traced and then quantified in ImageJ. With this data, specimens from black sand beaches were found to be significantly darker than those from grey sand beaches using a Mann-Whitney-Wilcoxon test ($p < 0.01$). Although this pattern is consistent with cryptic colouration, another possibility is that the darker colouration is the result of differences in sand grain abrasiveness causing scales to be eroded more quickly, making the beetles appear darker in colour. Additionally, general biological observations of the genus *Actizeta* from fieldwork and keeping the beetles in captivity are also recorded.

Introduction

Within species, colouration can serve various functions and often varies substantially between populations. Some species may exhibit cryptic colouration where the colouration of the body may closely match the surrounding environment. This has been observed in species that live in sandy environments such as the New Zealand seaweed beetle *Chaerodes trachyscelides* White 1846, which can vary substantially in colour and tends to match the sand substrate they reside on (Harris 1988). This has also been observed in several dune-dwelling species of the tiger beetle *Zecicindela* (Larochelle & Larivière 2013). Similar substrate-dependent

variation may occur in other sand-dwelling beetles but is poorly documented in New Zealand.

The genus *Actizeta* is composed of two species: *Actizeta albata* Pascoe, 1875 and *Actizeta fusca* Watt, 1992. Both species live along the coastline on sandy beaches and in sand dunes, with *A. albata* being recorded from the North Island and northern half of the South Island whereas *A. fusca* is only known from the North Island (Watt 1992b). Although they are relatively abundant, due to their small size and burrowing habits they are seldom observed. When the sand is warm enough, the beetles climb to the surface of the sand and can be seen roaming in search of food. As adults, they act as scavengers and will feed on various food sources such as dead invertebrates and fish (Harris 1970a). The body of the beetle is covered in distinctive white and black patterning, particularly on the elytra and pronotum, that can vary substantially within and between populations (Fig 1).

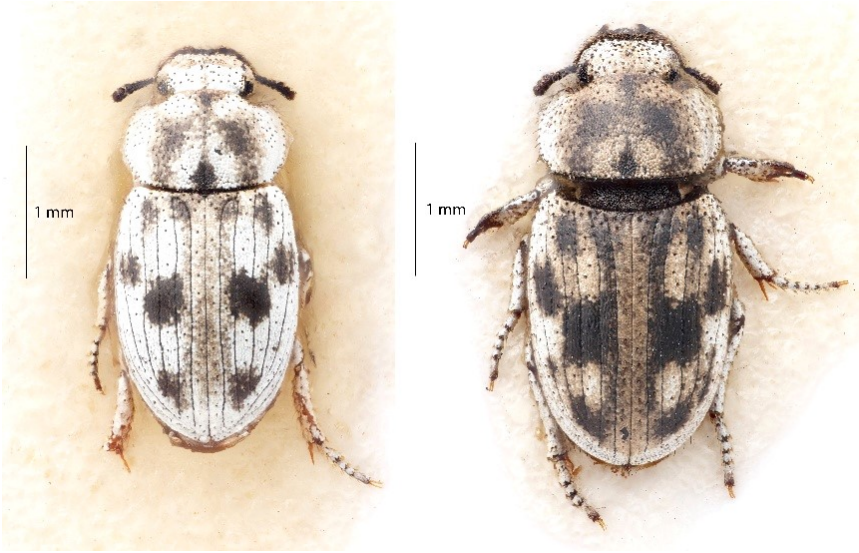

Figure 1. *Actizeta albata* collected from Waikawa beach (left) and from Patea beach (right). Photos by Bryce McQuillan. All rights reserved.

The sand along the coastline of the Taranaki and Whanganui regions is derived from volcanic andesite rocks which are prevalent in the region. These rocks are rich in titanomagnetite and other darkly coloured minerals which give the sandy beaches in the region a distinctive black colour (Brathwaite et al. 2017). To the south of these regions the source material for the sand changes to sedimentary greywacke, which causes the sand to gradually shift to a light grey colour in the Kāpiti region (Kasper-Zubillaga et al. 2006). While travelling in these regions, the author searched for *A. albata*, particularly in the Taranaki region where few specimens have been observed. During this time, it was noted that populations of *A. albata* in the black sand regions appeared to be coloured darker than those from grey sand regions (Fig 1), but the differences were subtle enough that more extensive sampling would be needed to confirm this.

To investigate this observation, this study aims to determine if there is variation in the colouration of *A. albata* along the Kāpiti–Taranaki coastline. Based on previous field observations, it was hypothesised that *A. albata* from the black sands of the Whanganui–Taranaki region would have darker colouration than those from the grey sands of the Kāpiti region. Observations on the general biology of the genus *Actizeta* from fieldwork and keeping specimens in captivity are also recorded.

Method

Collection

The beetles were collected from several beaches along the Kāpiti–Taranaki coastline throughout January and early February 2026 (Fig 2). This stretch of coastline was selected as the overall sand colour is light grey in the Kāpiti region before transitioning to black coloured in the Whanganui–Taranaki region. In the Kāpiti grey sand region, Waikanae, Waikawa and Foxton beaches were selected whilst in the Whanganui–Taranaki region the Patea and Ohawe beaches were chosen. Kai Iwi and Kaupokonui beaches were also considered, but the beetles were either absent or too few.

From each beach, a minimum of twenty live *A. albata* specimens were collected. The beetles were primarily collected by sieving sand with a stainless-steel sieve that had a mesh size of 3 x 3 mm. Loose dry sand from

the base of dunes was passed through the sieve and the remaining debris were placed into a tray and searched through for the beetles. The sand was then returned to the dune to prevent erosion. Alternatively, the beetles were sometimes found walking on the surface of sand or feeding on dead carrion. After collection, the beetles were card mounted.

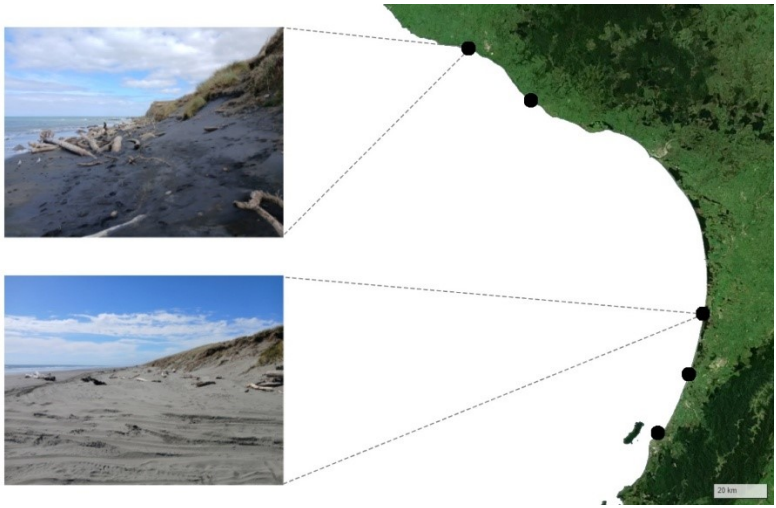

Figure 2. Map of collection sites (black dots) on the Kāpiti–Taranaki coastline with images of Ohawe beach (top) and Foxton beach (bottom) to demonstrate differences in sand colour between regions.

Body colour

The elytra and pronotum are covered in scales that range from white to light brown in colour while the base colour of both features are black. Some scales may be wholly or partly transparent, which exposes the black colour of these segments and thus creates black patterning which makes the beetle appear overall darker in colour. Alternatively, scales may also be missing, which also causes black colouration. To determine the extent of black patterning, the beetles were examined under microscope, and the extent of black areas was digitally traced in Photopea (2026) onto an

outline template of the elytra and pronotum and coloured black. This image was then imported into ImageJ (Schneider et al. 2012). The histogram function was used to count the number of black pixels. The proportion of black pixels relative to other pixels was then used to determine the percentage of the elytra and pronotum that were coloured black.

Table 1. Location of collection sites with their sand colour type and specimens collected.

Beach	Sand colour	Number of specimens
Waikanae	Grey	22
Waikawa	Grey	22
Foxton	Grey	31
Patea	Black	21
Ohawe	Black	29

Statistical tests

All statistical tests were performed in RStudio (Posit team 2026) and the dataset was broken down into the two beach sand colour categories: grey and black. The normality of the data was tested using the Shapiro-Wilk test and was found to be non-normally distributed. The two sand colour categories were tested for significant differences between groups using a Mann-Whitney-Wilcoxon test. The data from all five beaches were used to create box and whisker plots to visualise the dataset.

Results

A total of 125 specimens were collected: 75 from grey sand beaches and 50 from black sand beaches (Table 1). The box and whisker plots (Fig 3)

show that the beetles collected from the black sand beaches were generally darker in overall colour than those from the grey sand beaches. This is further confirmed by the Mann-Whitney-Wilcoxon test, which found there was a significant difference between the two groups ($p < 0.01$).

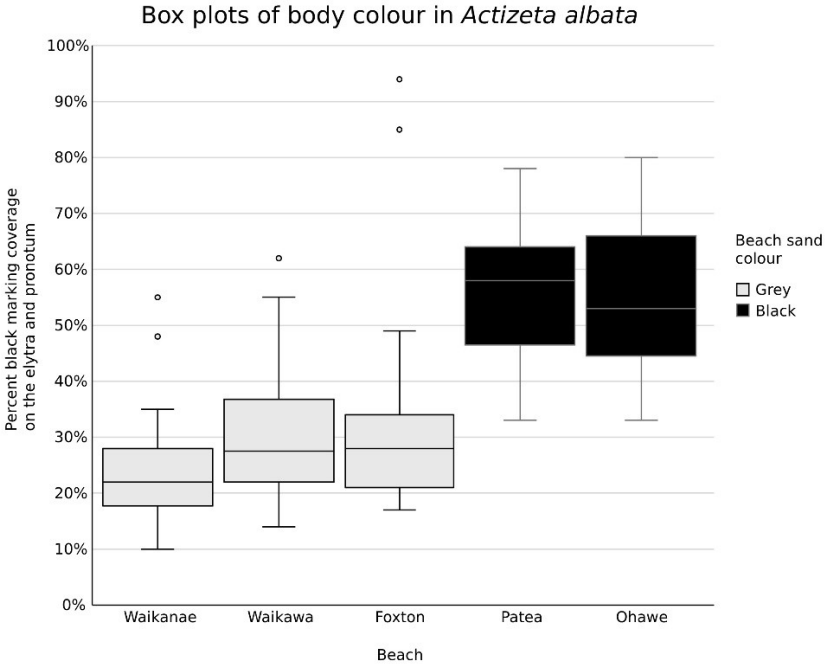

Figure 3. Box-and-whisker plots showing the percentage of black markings covering the elytra and pronotum. Plots are presented for each collection site, with beach sand colour indicated.

Discussion

Specimens from the black sand beaches in the Taranaki–Whanganui region were distinctly darker than those from the grey sand beaches of the Kāpiti region, confirming the hypothesis. However, this finding is limited to the

Kāpiti–Taranaki coastline and further investigation is needed to confirm that this pattern is consistent throughout New Zealand's beaches.

The darker colour of *A. albata* on the black sand beaches could be interpreted as an example of cryptic colouration, where a species is coloured to blend in with its surrounding habitat. Individual *A. albata* with colouration that more closely matches their substrate may blend in better and be more able to avoid being predated on. This pattern is consistent with some other coastal beetles, such as *C. trachyscelides* and *Zecicindela*, where colouration matches the sand substrate (Harris 1988; Larochelle & Larivière 2013).

Although cryptic colouration is a tempting explanation, another possibility is that the different sand types wear away the body scales at different rates. Under microscope, sand grains from the two black sand sites were larger and more angular than those from the grey sand sites. It is possible that because of this, the black sand causes the scales to erode more quickly, thus making the dark markings more extensive. As such, the darker colouration of *A. albata* on the black sand beaches may instead be coincidental. To test this, further studies could compare colouration of *A. albata* between beaches with similar colours, but differing grain sizes and shapes.

In conclusion, along the Kāpiti–Taranaki coastline *A. albata* from black sand beaches were consistently darker than those collected from grey sand beaches. Although on a surface level this may appear to be cryptic colouration, it is also possible that sand abrasion is the actual cause of this pattern. Further research is needed to examine these two explanations for the substrate-dependent colour variation in *A. albata* along the Kāpiti–Taranaki coastline.

Biological notes

Diet

During fieldwork, *A. albata* was often observed feeding upon dead invertebrates, particularly beetles and sandhoppers, with larger invertebrates having numerous *A. albata* swarming their dead bodies. In captivity, they were successfully sustained for several weeks on freshly killed terrestrial amphipods and would also accept dead flies (both adults and larvae). They were also frequently found feeding on dead seabirds,

sometimes occurring in large numbers. These observations conform with Harris (1970a), who also observed that they frequently fed upon a wide range of dead invertebrates and fish.

It was reported by Watt (1992a) that the gut content of *Actizeta* appeared to be mostly dead plant material. Feeding on dead plant debris was never observed during this study and attempts to feed them carrot in captivity appeared to be rejected, so more study is required to determine if plants constitute part of the adult diet.

Activity

Actizeta has previously been reported to be diurnal by Harris (1970b). However, field observations found numerous instances of *Actizeta* on the surface feeding on carrion at all hours of night. They were also frequently spotted resting above the surface and sometimes seen walking around at night. This was also observed in captive *A. albata* when a camera was set up to image their enclosure every fifteen minutes at night. This appears to be contrary to another experiment by Harris (1970b), where the beetles would burrow whenever the sky became cloudy and the temperature dropped. This behaviour was observed to be true during daytime, but the beetles' nocturnal activities appear to indicate that this behaviour has another layer of complexity to it.

Distribution

Watt (1992b) reported that *A. albata* is widely distributed throughout the North Island and northern South Island as far down as the mid-Canterbury region. Several records on the website iNaturalist have extended this range to Dunedin. Due to the cryptic nature of the species, it remains possible that its distribution extends even further south.

***Actizeta fusca* habitat**

On some beaches, particularly Waikawa and Foxton beach, *A. fusca* was frequently found co-occurring with *A. albata*, albeit in smaller numbers. Watt (1992a) reported that this species tends to occur further inland than *A. albata*. Field observations during this study noted that while both species can be found throughout dune systems, *A. fusca* did not appear to be more abundant further inland. This should be further investigated with a population distribution survey.

Acknowledgements

The author wishes to thank Bryce McQuillan for providing excellent photos of *Actizeta albata* and for initial instruction on how to find the beetle.

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Moths from Mahakirau Forest Estate, Manaia, Coromandel, New Zealand, 2015 – 2026: an annotated list

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Abstract

We provide a list of the Lepidoptera recorded during the first author's residence at the Mahakirau Forest Estate from 2015 to 2026 with annotations. A total of 423 species is recorded, representing the longest list of species so far recorded for a single restricted site in northern New Zealand. The most significant finds are briefly discussed and the list compared to those for forested areas of the Auckland district.

Introduction

The Mahakirau Forest Estate comprises approximately 600 hectares of mature broadleaf/podocarp QEII convenanted forest located on the crest of the upper Coromandel Range. Many of the ancient trees, such as kauri (*Agathis australis*), would have been cleared during earlier logging operations; nevertheless much of the remaining forest can be considered relatively pristine. There are significant areas of kānuka (*Kunzea ericoides*), fivefinger (*Pseudopanax arboreus*), rewarewa (*Knightia excelsa*), māhoe (*Melicytus ramiflorus*), putaputawētā (*Carpodetus serratus*), nikau palm *(Rhopalostylis sapida),* māpou (*Myrsine australis*) interposed with taraire (*Beilschmiedia taraire*) and northern rātā (*Metrosideros robusta*), and mature podocarps such as Hall's tōtara (*Podocarpus hallii*) and rimu (*Dacrydium cupressinum*).

The entrance, with co-ordinates -36.846°, 175.548° is accessed through a password-protected electric gate located off the 309 Road which links Coromandel and Whitianga townships. The estate itself, with altitudes ranging from 250 to 550 m, is sub-divided into 25 lots, from 15 to 25

hectares each, including an allocated build site accessed off a sealed easement road traversing the estate. AJS's first property on the estate, with co-ordinates -36.866°, 175.547° was purchased in March 2015, with initial light trapping commencing in November 2015. Catches were recorded on iNaturalist and a reference collection established with voucher specimens.

In February 2018, due to changing personal circumstances, a decision was made to sell up and return to the UK permanently. AJS donated the valuable collection to the New Zealand Arthropod Collection (NZAC) at Manaaki Whenua Landcare Research (now Bioeconomy Science Institute) in Auckland. RJBH subequently transferred the specimens which added significantly to knowledge of Coromandel's fauna, formerly rather poorly represented in any collection. It should be noted that whilst the Lepidoptera fauna of the Auckland district (AK, as defined by Crosby et al. 1998) has been relatively well sampled (see, e.g., Emmerson & Hoare 2019) that of the Coromandel district, CL, is far less well studied, and the current list represents the first major survey to be published. Many collection records from the CL district remain unpublished. A short list of 61 species recorded from Hauturu (Little Barrier Island) was included in the book by Wade and Veitch (2019), but one name (*Azelina venustula*) listed is a synonym of another (*Ischalis gallaria*) so the actual list is 60 species. This undoubtedly represents only a fraction of the Lepidoptera diversity of this very important island.

At the end of 2021, after three and a half years living in the UK, AJS made the decision to return to New Zealand and to Mahakirau Forest Estate, but this time to a different plot. The new location's immediate habitat differs noticeably to the previous in being far more open, hence exposed, and at a lower altitude (circa 310m) with large areas of exotic weeds such as gorse and blackberry which are slowly being restored to native shrubland. The rest of the surounding property, circa 12 hectares, comprises QEII covenanted bush.

Shortly after moving back to New Zealand AJS decided to re-commence a survey and monitoring programme and put together a new collection however, this time with a focus on the micro-moths. Whilst the majority of the light trapping nights were carried out at AJS's property, on occasion

other sites on the estate were sampled both at night and during the day time.

Methods

Moths were either trapped with a 125-Watt mercury vapour light fitted to a Robinson-pattern moth trap run off mains electricity or a home-made set-up utilising a m.v. lamp only, positioned on a large white sheet on the property's deck. Many moths, whilst attracted by the light, chose to settle on the adjacent building's walls and windows enabling their inspection and recording. The technique of regularly 'patrolling' the immediate vicinity increased the chances of spotting fleeting visitors before their inevitable departure.

When sampling at more remote locations a portable generator was used to provide power. Infrequently a portable Heath Trap with a 40W actinic (blue) tube run off a 12v DC battery was used. The normal procedure was to run the traps for no more then 2 to 3 hours after dark, taking into consideration whether conditions were considered suitable, i.e relatively mild, perferably overcast with little to no wind.

Surveys were performed throughout the year, more frequently either side of winter, and species uploaded to iNaturalist allowing for subsequent confirmation of identification when necessary. Specimens of doubtful identity, mainly micro-moths, were compared with material in the New Zealand Arthropod Collection (NZAC) and identified by RJBH. Moths were also recorded when seen during the day and at the house windows at night. All the species listed are represented by voucher specimens, located either in AJS's (2nd) personal collection in Mahakirau or in NZAC (1st collection).

The general procedure has been to compile a species list from each mothing event and then to upload to iNaturalist observations of note, i.e. unusual or unique records. The number of specimens of each species, i.e. abundance data, was only recorded when considered noteworthy by the first author. Hence, assessments of species status given in lists are usually subjective and reflect overall impressions of frequency and abundance; however, an attempt is made here to give a degree of rigour to these assessments based on the following guidelines derived from Emmerson and Hoare (2019). Species seen in at least 80% of the counts during their

flight period are judged to be ‘very common’; those recorded in between 50% and 80% of counts are ‘common’ and those between 20% and 50% ‘fairly common’. Those recorded in less than 20% of samples during their flight-time are ‘uncommon’. For those species for which there are few records, the date for each record is given. More subjectively, very common species that are often observed in numbers (i.e. 10 or more per sampling event) are described as ‘abundant’.

The classification largely follows Dugdale (1988) as updated by Hoare (2010), but the order of families is taken from van Nieukerken *et al*. (2011), and the family classification of Gelechioidea follows Heikkilä *et al.* (2014) as updated by Yapar *et al.* (2025), and that of Tineoidea follows Regier *et al.* (2015). Nomenclature of Scopariinae follows the online guide of Hoare and Rhode (2015), and there are some updates to Noctuidae arising from the revisions of Hoare (2017; 2019). Identification/interpretation of problematic names in unrevised groups (e.g. Oecophoridae) follows that in NZAC, and some changes are likely following revisionary work. Usage of the abbreviations ‘sp. near’ (or ‘sp. nr’) and ‘cf.’ follows Hoare (2001), except that *Pasiphila* sp. cf. *magnimaculata* (Geometridae) is certainly an unnamed species distinct from *P. magnimaculata*; the name is used here because of its appearance in the latest threat assessment of New Zealand Lepidoptera (Hoare *et al.* 2026). Where current subfamily or genus placement of a species is doubtful or incorrect, the affected name is followed by ‘s.l.’ (in parentheses after the genus), indicating ‘in the broad sense’ (*sensu lato*). Where more than one species may currently be confused under a name in an unrevised group, the species name is followed by ‘s.l.’.

Results and Discussion

Lepidoptera species list comparison

The total of 423 species recorded across the Mahakirau Forest Estate during the period 2015 – 2026 represents by far the most significant survey of the Lepidoptera of the Coromandel peninsula.

There have been numerous short- to medium-term Lepidoptera surveys published from single localities in New Zealand since 1960; see Emmerson and Hoare (2019). These lists differ quite substantially in area covered, local habitat, period of survey, and collecting methodology and

intensity. With the caveats outlined above, the Mahakirau list is the richest in total species followed by the Hawkes Bay (Davies 1973) list at 376 species and then the Redvale, AK (Emmerson & Hoare 2019) list at 357 species.

Further context for this survey is provided by two unpublished lists currently being compiled by RJBH. These are (1) the cumulative list (see Introduction) of Lepidoptera species recorded in the Auckland district (AK) and (2) a cumulative list of those recorded in the Waitakere Ranges of west Auckland. At the time of writing (2026), the Auckland list includes 812 species (of which 19 are considered 'accidentals', i.e. non-established adventives that have occurred once only, or which established briefly and were exterminated). The Waitakere Ranges list includes 663 species. Both lists, like the present one, include unnamed species recognised as such in NZAC.

Notable records

For Biostatus abbreviations (E, N, A, V) after each species name, see below under 'Species List'.

***Schiffermuelleria (s.l.) orthophanes* E.** First recorded in November 2022 attracted to a 40W Heath Trap adjacent to the bush. On January 2025 a second example was discovered inside AJS's bathroom during the daytime.
***Trachypepla cyphonias* E (Fig. 1).** Recorded once at 11 Mahakirau Road in December 2023 then at 25 Mahakirau Road in February 2024. Threat status 'Naturally Uncommon'. Associated with trapped leaf-litter of kānuka (reared specimens in NZAC).
***Ochetarcha* sp. A. E (Fig. 2).** Unnamed species recorded once in April 2025. Larvae feed in rachides of silver fern (*Alsophila dealbata*).
***Amblyptilia deprivatalis* E.** Two at m.v. light in Jan 2017 (both in NZAC). This probably represents the first published record of this mysterious species since its description by Walker in 1864. However, there is a specimen in NZAC that also appears to belong to *A. deprivatalis*, collected by Alfred Philpott at Nelson on 3 July 1927; this specimen is missing its abdomen. It can be distinguished from the common *A. repletalis* by the almost unpatterned yellowish forewing that lacks transverse strigulae, and by the pallid base to the hind-tibia (brownish in *A. repletalis*). Note that what has been referred to as '*Platyptilia deprivatalis*' in the literature, e.g. by Hudson (1928), is a very strongly marked moth that does not resemble Walker's type of *deprivatalis* at all, and appears to be an unnamed

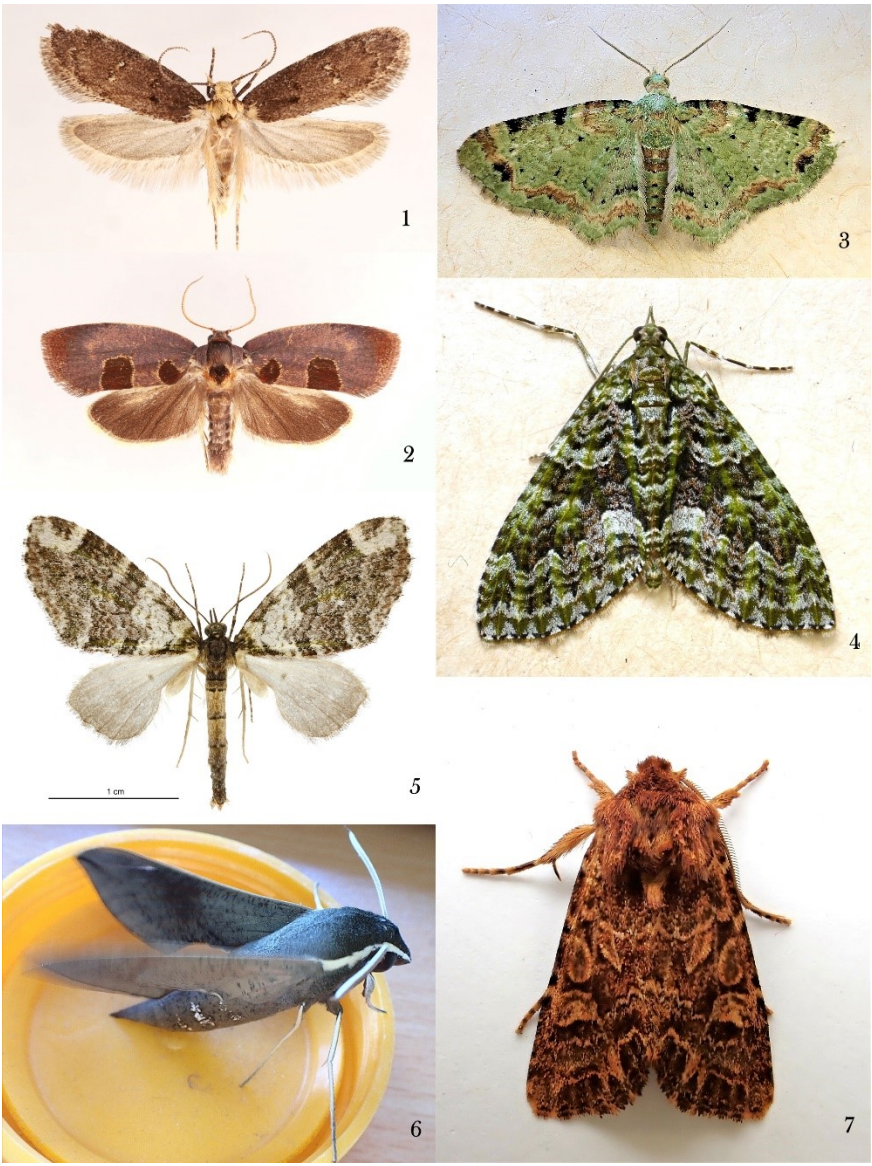
species related to *P. campsiptera*.
***Pasiphila* sp. cf. *magnimaculata* E (Fig. 3).** Unnamed 'Jade Green' pug whose larvae feeds on *Brachyglottis kirkii* flowers. First recorded in August 2016 attracted to UV light then occasionaly from August to October every year. An early spring season species.
***Tatosoma agrionata* E (Fig. 4).** First recorded in October 2022 and then subsequently on four more occasions through to January. By far the most northerly records. Larvae are mistletoe feeders.
***Tatosoma apicipallida* E (Fig. 5)**. First recorded on 11 Oct 2016, followed by regular observations through spring. At the very north of its range. Host-plant unknown.
***Hippotion scrofa* V (Fig. 6).** First record for New Zealand was at Mahakirau in February 2025. A common moth in South Eastern Australia. A second example was observed in Whangarei on 26th February 2025 and recorded on iNaturalist.
***Ichneutica brunneosa* E (Fig. 7).** At the north of its range, first recorded in October 2016 and then on 6 further occasions throughout summer months. A regular visitor to U.V. lamps. Host-plant unknown.

Species List
The following abbreviations after the species name indicate the presumed biostatus of each species in New Zealand: E= Endemic, A= Adventive, N=Native, V=Vagrant.

Micropterigidae (3 species)
***Zealandopterix zonodoxa* E.** Can be commonly found resting on outside walls during the daytime in November and December. Occasional visitors to light.
***Sabatinca doroxena* E.** More than half a dozen recorded at light in November 2017.
***Sabatinca lucilia* E.** Recorded once at light, in February 2017.

Hepialidae (2 species)
***Aenetus virescens* E.** Males extremely common at the light trap all year round but abundant from late summer. Females only occasionally encountered and never at a light trap.
***Dumbletonius unimaculatus* E.** Very common in late summer, e.g. January and February.

Heliozelidae (1 species)
***Heliozela catoptrias* A.** Recorded once in November 2024 but minute and inconspicuous so likely to be much more common. The identity of this adventive species needs checking; Australian species of *Heliozela* are unrevised and the identification is based on superficial appearance only. It is a leaf-miner on kānuka.



Significant records from Mahakirau. Live specimens are from Mahakirau, photographed by AJS; set specimens are non-CL specimens in NZAC. Fig. 1, *Trachypepla cyphonias* (Oecophoridae); 2, *Ochetarcha* sp. A (Tortricidae); 3, *Pasiphila* sp. cf. *magnimaculata* (Geometridae); 4, *Tatosoma agrionata* (Geometridae); 5, *T. apicipallida* (Geometridae); 6, *Hippotion scrofa* (Sphingidae); 7, *Ichneutica brunneosa* (Noctuidae).

Cecidosidae (1 species)
Xanadoses nielseni Hoare & Dugdale E. Recorded for the first time in December 2025.

Psychidae (6 species)
Cebysa leucotelus **A.** First recorded (male) in March 2025 during the daytime.
Liothula omnivora **E.** Larval cases common; they are presumed to belong to this species, but the case of the unnamed *Liothula* mentioned below is unknown so some may possibly be referable to that species.
Liothula **sp. A. E.** Fairly common at light, an unnamed species which is closely related to *omnivora* but emerges in March to early April and has more transparent hindwings. Larval case unknown.
Lepidoscia heliochares **A.** Uncommon with only a single record in September 2023; attracted to light.
Lepidoscia protorna **A.** Extremely common, occuring throughout the summer (December through to April).
Lepidoscia lainodes **A.** Uncommon with only a single record in July 2025 during daytime.

Tineidae (34 species)
Habrophila compseuta **E.** Recorded at two separate locations in November and February.
Sagephora phortegella **E.** Common from November to January.
Amphixystis hapsimacha **E.** Recorded from November to February every year.
Archyala paraglypta **E.** Fairly common, recorded from December to February.
Crypsitricha agriopa **E.** Recorded once, in January 2022.
Crypsitricha generosa **E.** Common, recorded from September to June.
Crypsitricha mesotypa **E.** Recorded from September to December.
Crypsitricha pharotoma **E.** Fairly common, recorded late spring/early summer.
Crypsitricha roseata **E.** Fairly common, recorded in December, January and February.
Endophthora omogramma **E.** First recorded in February 2023, then on two further occasions in November and December.
Endophthora pallacopis **E.** Only recorded once, in December 2022.
Endophthora rubiginella **E.** Scarce, only recorded once in January 2025.
Erechthias chasmatias **E.** Recorded regularly in November and December.
Erechthias chionodira **E.** Recorded on several occasions in January and February.
Erechthias exospila **E.** Recorded once, in December 2024.
Erechthias hemiclistra **E.** Recorded for the first time in December and then March 2025.
Erechthias stilbella **E.** Recorded twice in January 2025.
Lysiphragma epixyla **E.** First recorded in December 2016 then January 2025. More than one species is currently confused under this name (RJBH, pers. obs.) but the Mahakirau specimens are the true *L. epixyla*.

***Monopis crocicapitella* A.** Attracted to moth traps in low numbers throughout the year.
***Monopis dimorphella* E.** First recorded in April 2025.
***Monopis ethelella* A.** Attracted to moth traps in low numbers throughout the year.
***Monopis* sp. near *icterogastra* A.** Fairly common, first recorded in March 2023.
***Monopis ornithias* E.** First recorded in January 2023, then the following December, both during daytime indoors.
***Opogona omoscopa* A.** Very common and can be encountered throughout the year.
***Prothinodes grammocosma* E.** Uncommon, recorded in late spring. There is currently uncertainty in differentiating between the two *Prothinodes*, see *P. lutata* below.
***Prothinodes lutata* E.** Larger and paler specimens without a distinct longitudinal streak are tentatively referred to *P. lutata* here. Variation is such that dissection will be required to confirm; there appear to be differences in male genitalia between the two species (RJBH, pers. obs.).
***Tephrosara cimmeria* E.** Only recorded twice, in March and April, both indoors during daytime.
***Tinea* (s.l.) *argodelta* E.** Scarce, recorded once in March 2024 indoors during daytime.
***Tinea* (s.l.) *belonota* E.** Recorded twice, in February 2023 and March 2025.
***Tinea* (s.l.) *conferta* E.** Recorded in October, November and December.
***Tinea* (s.l.) *conspecta* E.** Recorded at a number of MFE sites. In November 2023, two individuals flying over rotting logs in native bush at sunset.
***Tinea* (s.l.) *fagicola* E.** Recorded for the first time on 21 December 2025.
***Tinea* (s.l.) *margaritis* E.** Recorded for the first time on 24 November 2025.
***Trithamnora certella* E.** Recorded once on an outside window during daytime in November 2022.

Roeslerstammiidae (2 species)
***Dolichernis chloroleuca* E.** Only recorded once, in April 2023.
***Vanicela disjunctella* E.** Only recorded once, in February 2025.

Gracillariidae (8 species)
***Acrocercops* (s.l.) *leucocyma* E.** Recorded twice, once in November and then December.
***Caloptilia chalcodelta* E.** Recorded in June and December.
***Caloptilia chrysitis* E.** Common from November to February.
***Caloptilia linearis* E.** Commonly recorded from October to April.
***Conopomorpha cyanospila* E.** Recorded once when attracted to light in February 2025. Surprising as no mature titoki is present on the estate.
***Eumetriochroa* (s.l.) *panacitorsens* E.** Recorded once, in January 2023.
***Eumetriochroa* (s.l.) *panacivagans* E.** Recorded once, in December 2021.
***Macarostola miniella* E.** Quite common, first recorded in January 2023.

Yponomeutidae (1 species)
***Zelleria porphyraula* E.** Recorded twice, in February 2023 and then November 2025.

Plutellidae (2 species)
***Leuroperna sera* N.** Recorded twice, both in January.
***Plutella xylostella* A.** Very common, recorded through summer and early autumn.

Glyphipterigidae (10 species)
***Chrysorthenches drosochalca* E.** Common from October to March.
***Chrysorthenches glypharcha* E**. Recorded for the first time on 21 December 2025.
***Chrysorthenches polita* E.** Fairly common from October to March.
***Chrysorthenches porphyritis* E.** Common from October to March with a couple of August records.
***Doxophyrtis hydrocosma* E.** Fairly common, recorded from October to February.
***Glyphipterix leptosema* E.** Only recorded once, in November 2017.
***Glyphipterix tungella* E.** Recorded once, in January 2023.
***Glyphipterix zelota* E.** Recorded once, in December 2023 during daytime sweeping of ponga.
***Proditrix gahniae* E.** Recorded twice, once in January and once February.
***Proditrix tetragona* E.** Uncommon, first recorded in March 2016, then in September and December. Usually attracted to UV Light.

Praydidae (1 species)
***Prays nephelomima* A.** Recorded once, in January 2023, attracted to UV light.

Lyonetiidae (1 species)
***Stegommata sulfuratella* A.** Fairly common, recorded from December to January.

Family unknown (1 species)
***Cadmogenes literata* E.** Recorded twice, once in November and then December.

Lecithoceridae (3 species)
***Compsistis bifaciella* E.** Common in early summer, often swept from roadside vegetation during daytime.
***Lecithocera micromela* A.** First recorded in November 2016 and then March 2024.
***Sarisophora leucoscia* A.** Only recorded once, in January 2025.

Xyloryctidae (28 species)
***Gymnobathra dinocosma* E.** Fairly common, recorded in November and December.
***Gymnobathra flavidella* E.** Very common through the summer months. There may be two species under this name, in which case both occur on the estate.
***Gymnobathra hyetodes* E.** Common, occuring throughout the summer, sometimes

during daytime.
***Gymnobathra omphalota* E.** Fairly common, all records in November.
***Gymnobathra sarcoxantha* E.** Only recorded once, in December 2022 during the daytime.
***Gymnobathra tholodella* E.** Very common, recorded from January to April.
***Lathicrossa leucocentra* E.** Recorded twice, once in October and January.
***Hierodoris atychioides* E.** Recorded once at light, in December 2022.
***Hierodoris illita* E.** Recorded once in January 2017, then twice in November 2025 and again in December.
***Hierodoris stella* E.** Recorded once, in December 2017 in daytime.
***Hierodoris tygris* E.** Recorded once, in March 2017 and for a second time in February 2026.
***Izatha attactella* E.** Fairly common, recorded from August to October.
***Izatha austera* E** Fairly common, recorded in November, December and January at light.
***Izatha balanophora* E.** Only recorded once, in December 2016.
***Izatha blepharidota* E.** Uncommon, recorded in November and December.
***Izatha churtoni* E.** Common during the summer.
***Izatha dasydisca* E.** Uncommon, first recorded in November 2017 then January 2023, January 2026 and February 2026.
***Izatha epiphanes* E.** Common from November to February.
***Izatha gibbsi* E.** Recorded for the first time on 17 February 2026.
***Izatha hudsoni* E.** Common from October to December.
***Izatha mesoschista* E.** Recorded twice, in November and December.
***Izatha metadelta* E.** Recorded twice, first in January 2017 then January 2024.
***Izatha peroneanella* E.** Very common throughout the summer.
***Izatha phaeoptila* E.** Fairly common during mid-summer.
***Izatha prasophyta* E.** Recorded once on 27 January 2026.
***Schiffermuelleria* (s.l.) *orthophanes* E.** First recorded in November 2022 attracted to a Heath Trap. On January 2025 a second example was discovered inside the bathroom during the daytime.
***Thamnosara sublitella* E.** Recorded twice, both in November.

Oecophoridae (34 species)
***Atomotricha isogama* E.** Common, recorded from September to January.
***Barea codrella* E.** Recorded once on 13 January 2026.
***Endrosis sarcitrella* A.** Only recorded twice, once in April and once in July, both indoors.
***Eulechria* (s.l.) *zophoessa* E.** Recorded three times, all in January.
***Hofmannophila pseudospretella* A.** Recorded once in October and then December. Both inside buildings.
***Mermeristis spodiaea* E.** Recorded for the first time on 28 September 2025 then 15 January 2026.

***Tachystola acroxantha* A.** Fairly common from October to April.
***Tingena actinias* E.** Fairly common, all records in November.
***Tingena ancogramma* E.** Common from December to February.
***Tingena apanthes* E.** Recorded twice, in February then March.
***Tingena armigerella* E.** Very common, from October to February.
***Tingena chloradelpha* E.** Recorded twice, once in October 2022 and then November 2023.
***Tingena chrysogramma* E.** Common from November to February.
***Tingena clarkei* E.** Very common from December to February.
***Tingena compsogramma* E.** Recorded three times, all in December.
***Tingena contextella* E.** First recorded in November 2023 then November 2024.
***Tingena crotala* E.** Recorded in October 2017 then September 2022.
***Tingena grata* E.** Recorded once, in January 2017.
***Tingena hemimochla* E.** Fairly common, recorded in October, November and December.
***Tingena innotella* E.** Recorded once, in October 2022.
***Tingena nycteris* E.** Recorded once, in December 2021.
***Tingena ombrodella* E.** Recorded three times across two sites, all in November.
***Tingena penthalea* E.** First recorded in January 2018 then September 2022.
***Tingena plagiatella* E.** Fairly common, recorded from October to December.
***Trachypepla amphileuca* E.** Recorded once in December 2022.
***Trachypepla conspicuella* E.** Recorded for the first time on 8 December 2025.
***Trachypepla cyphonias* E.** Recorded once at 11 Mahakirau in December 2023 then at 25 Mahakirau Road in February 2024.
***Trachypepla festiva* E.** Recorded twice, once in December and January.
***Trachypepla galaxias* E.** Very common from November to December.
***Trachypepla hieropis* E**. First recorded in December 2025.
***Trachypepla leucoplanetis* E.** First recorded in November 2016, then December 2024 and November 2025.
***Trachypepla nimbosa* E.** Recorded twice, once in December 2025 and then January 2026.
***Trachypepla protochlora* E.** Only recorded once in December 2017.
***Trachypepla* (s.l.) *indolescens* A.** Recorded from December to February.

Stenomatidae (1 species)
Note. Stenomatidae, formerly a subfamily of Depressariidae, was erected to full family rank by Yapar *et al*. (2025).
***Agriophara colligatella* E.** Recorded in January and March 2017 and then in November 2025 at a different location.

Depressariidae (4 species)
***Agonopterix umbellana* A.** Gorse feeder, only recorded once in August 2022.
***Donacostola notabilis* E.** Recorded three times, November, December and January.

***Nymphostola galactina* E.** Fairly common, recorded at a number of sites from October to January.
***Phaeosaces coarctatella* E.** Common, but all records in October.

Cosmopterigidae (2 species)
***Pyroderces* (s.l.) *apparitella* E.** Fairly common from December to January.
***Pyroderces* (s.l.) *deamatella* E.** Fairly common, first recorded in January 2023. Attracted to light.

Gelechiidae (5 species)
***Anisoplaca cosmia* A.** First of two records in January 2025.
***Bilobata subsecivella* A.** Only recorded once, in January 2025.
***Phthorimaea operculella* A.** Recorded once, in February 2025.
***Platyedra subcinerea* A.** Recorded once, in January 2025.
***Symmetrischema tangolias* A.** Recorded once, in January 2024.

Elachistidae (6 species)
Note. The updated classification of Yapar *et al*. (2025), which includes *Eutorna* (Cacochroinae) in Elachistidae is followed.
***Elachista archaeonoma* (s.l.) E.** Very common but more than one species may be confused under this name. Yet to be resolved.
***Elachista gerasmia* E.** Recorded once in February and then November 2023.
***Circoxena ditrocha* E.** Fairly common in January and February.
***Microcolona limodes* E.** Very common from October to June.
***Eutorna inornata* (s.l.) E.** Very common, recorded from August to September. Since *E. inornata* is usually a coastal species with larvae on *Goodenia radicans*, the identification of this forest species needs checking.
***Eutorna symmorpha* E.** Very common, recorded from August to April.

Coleophoridae (3 species)
***Coleophora alcyonipennella* A.** Recorded once, in January 2018.
***Coleophora deauratella* A.** Recorded for the first time on 26 November 2025.
***Coleophora mayrella* A.** Only recorded once in March 2017 at the author's first property.

Batrachedridae (2 species)
***Batrachedra arenosella* s.l. E.** Recorded twice, once in January 2017 and once in December 2024. More than one species is confused under this name, Walker's type specimens are lost and the taxonomy has yet to be resolved.
***Batrachedra psithyra* E**. Very common amongst mātātā fern (*Histiopteris incisa*) in late spring through early summer. This is a likely larval host.

Stathmopodidae (6 species)
***Stathmopoda aposema* E.** Uncommon, recorded in March 2017.
***Stathmopoda caminora* E.** Recorded once, in October 2024.
***Stathmopoda horticola* E.** Very common, recorded from October to March.

***Stathmopoda skelloni* E.** Recorded twice, once in September and once in April.
***Stathmopoda trimolybdias* E.** Recorded three times, twice in December and once in February.
***Thylacosceles radians* E.** Recorded twice, once in December and once in January.

Pterophoridae (5 species)
***Amblyptilia deprivatalis* E.** Two at m.v. light in Jan 2017: one at AJS's first property and one at the picnic area at lower elevation. See under 'Notable Records' above.
***Amblyptilia falcatalis* E.** Only recorded once in January 2018.
***Amblyptilia repletalis* E.** Very common, recorded from September to February.
***Platyptilia isodactylus* A.** Uncommon, all records in January**.**
***Stenoptilia zophodactylus* N.** Fairly common, recorded from October to April.

Copromorphidae (2 species)
***Isonomeutis amauropa* E.** Recorded regularly in the daytime during November and December.
***Isonomeutis restincta* E.** Recorded on four occasions, in December and January.

Carposinidae (9 species)
***Carposina neurophorella* A.** Recorded twice, once in December and once in January.
***Ctenarchis cramboides* E.** Considered a scarce moth, recorded for the first time in January 2025.
***Glaphyrarcha euthrepta* E.** Another scarcity, recorded twice in October 2016.
***Heterocrossa contactella* E.** Recorded twice, both in November.
***Heterocrossa eriphylla* E.** Common, recorded from October to January.
***Heterocrossa gonosemana* E.** Recorded from January to March.
***Heterocrossa iophaea* E.** Only recorded once; 29 January 2025.
***Heterocrossa philpotti hudsoni* E.** Recorded from June to September.
***Heterocrossa rubophaga* E.** Recorded from October to January and once in June.

Choreutidae (2 species)
***Asterivora combinatana* E.** Recorded once; 12 February 2017.
***Tebenna micalis* N.** Common, recorded from December to March both during the daytime on daisies and at the light trap.

Tortricidae (41 species)
***Argyroploce* (s.l.) *chlorosaris* E.** Common, recorded in October and December.
***Crocidosema plebejana* A.** Recorded for the first time on 8 December 2025.
***Cryptaspasma querula* E.** Very common; can be found throughout the year.
***Cydia succedana* A.** Very common due to predominance of European gorse on the estate.
***Holocola charopa* E.** Only recorded once; 11 July 2022.
***Holocola emplasta* E.** Fairly common, recorded from August to December.

***Holocola parthenia* E.** Common, recorded from July to January.
***Holocola zopherana* E.** Very common, recorded from August to December.
***Strepsicrates ejectana* E.** Common from December to March.
***Apoctena conditana* E.** Recorded twice, both in October.
***Apoctena flavescens* E.** Very common, recorded from August to December.
***Apoctena orthocopa* E.** Very common in December and January.
***Apoctena orthropis* E.** Recorded twice, both in March.
***Apoctena tigris* E.** Fairly common, recorded from December to February.
***Capua intractana* A.** Very common throughout the year.
***Capua* (s.l.) *semiferana* E.** Very common throughout the year.
***Catamacta gavisana* E.** Fairly common from October to May.
***Catamacta alopecana* E.** Recorded twice, once in August and once in February.
***Catamacta lotinana* E.** Uncommon, recorded once in January.
***Clepsis divulsana* A.** Fairly common, recorded from September to March.
***Cnephasia* (s.l.) *incessana* E.** Common, recorded from December to March.
***Cnephasia* (s.l.) *jactatana* E.** Very common throughout the year.
***Ctenopseustis fraterna* E.** Very common, recorded most months of the year.
***Ctenopseustis obliquana* E.** Common from December to May.
***Dipterina imbriferana* E.** Recorded from December to February.
***Ecclitica torogramma* E.** Fairly common, recorded from December to February.
***Epalxiphora axenana* E.** Very common, encountered throughout the year.
***Epiphyas postvittana* A.** Very common throughout the year.
***Lopharcha insolita* E.** Uncommon, recorded in January and March.
***Ochetarcha miraculosa* E.** Scarce, only recorded once in February 2017.
***Ochetarcha* sp. A. E.** Unnamed species recorded once in April 2025.
***Planotortrix notophaea* E.** Common, recorded from December to February.
***Planotortrix excessana* (s.l.) E.** Common most months of the year. (No attempt has been made to distinguish this from *P. octo*, distinct on sex pheromones, and both may occur.)
***Prothelymna niphostrota* E.** Common from August to November.
***Pyrgotis arcuata* E.** Uncommon, recorded singly in November and January.
***Pyrgotis calligypsa* E.** Recorded twice, once in October and once in January.
***Pyrgotis chrysomela* E.** Common from September to April.
***Pyrgotis plagiatana* E.** Very common from September to January.
***Pyrgotis plinthoglypta* E.** Very common from August to February.
***Pyrgotis zygiana* E.** Uncommon, single records in August, November and February.
***Tortrix* (s.l.) *incendiaria* E.** Rare, only recorded once in September 2022.

Pyralidae (8 species)
***Achroia grisella* A.** Recorded on two occasions, in February and April.
***Crocydopora cinigerella* N.** First recorded in April 2025 at moth trap. Most northerly record on iNaturalist and first for the Coromandel. Possibly a migrant

individual.
***Ephestiopsis oenobarella* A.** Recorded in January and February.
***Galleria mellonella* A.** Fairly common from December to April.
***Homoeosoma anaspila* E.** Recorded once, in June 2023.
***Patagoniodes farinaria* N.** Fairly common, all records in January.
***Plodia interpunctella* A.** Two individual adults found in March 2017 in Rat Café.
***Gauna aegusalis* A.** Fairly common, recorded from September to March.

Crambidae (66 species)
***Culladia cuneiferellus* A.** Extremely common, recorded most months of the year.
***Argyria* (s.l.) *strophaea* E.** Fairly common, recorded December to April.
***Gadira acerella* E.** Fairly common, recorded in December and January.
***Glaucocharis auriscriptella* E.** Very common from November to February.
***Glaucocharis chrysochyta* E.** Common from October to February.
***Glaucocharis elaina* E.** Recorded twice, both in November.
***Glaucocharis harmonica* E.** Fairly common, recorded in October and November.
***Glaucocharis holanthes* E.** Fairly common, recorded in November and December.
***Glaucocharis lepidella* E.** Common, recorded from November to February.
***Glaucocharis leucoxantha* E.** Very common, recorded from August to November.
***Glaucocharis metallifera* E.** Only recorded once, on 29 November 2017.
***Glaucocharis parorma* E.** Fairly common, recorded from October to December.
***Glaucocharis pyrsophanes* E.** Fairly common from September to November.
***Glaucocharis selenaea* E.** Common from November to January.
***Orocrambus angustipennis* E.** Uncommon, recorded in December and March.
***Orocrambus apicellus* E.** Common from October to March.
***Orocrambus flexuosellus* E.** Extremely common from spring to autumn.
***Orocrambus ramosellus* E.** Common from October to April.
***Orocrambus vittellus* E.** Very common, encountered most months of the year.
***Trichophysetis* cf. *cretacea* A.** Recorded once on 12 February 2024.
***Hellula hydralis* V.** First recorded in December 2024 followed by another example in February 2025. A rare migrant from Australia, but there were several records in that summer.
***Musotima nitidalis* N.** Common from September to April.
***Hygraula nitens* N.** Common in November and December.
***Clepsicosma iridia* E.** Common from November to May.
***Deana hybreasalis* E.** Common from October to May.
***Diasemia grammalis* E.** Only recorded twice; 1 Feb 2017 and 8 Apr 2023.
***Diplopseustis perieresalis* N.** Very common most months of the year.
***Herpetogramma licarsisalis* A.** Only recorded once, on 19 Mar 2023.
***Proternia philocapna* E.** Common, recorded in December and January.
***Leucinodes cordalis* E.** Only recorded twice; 26 Feb 2017 and 5 Dec 2025.
***Spoladea recurvalis* A.** Recorded for the first time on 18 Apr 2023 and then on two subsequent occasions.

***Udea daiclesalis* E.** Fairly common, recorded from October to February.
***Uresiphita maorialis* E.** Recorded once, on 14 Feb 2024.
***Antiscopa epicomia* E.** Common, recorded from September to April.
***Eudonia aspidota* E.** Recorded commonly in October and November.
***Eudonia asterisca* E.** Fairly common, recorded from October to April.
***Eudonia bisinualis* E.** Very common, recorded in numbers from October to May.
***Eudonia characta* E.** Uncommon, recorded in December and January.
***Eudonia chlamydota* E.** Common from October to April.
***Eudonia choristis* E.** Commonly recorded in November.
***Eudonia colpota* E.** Fairly common from November to January.
***Eudonia dinodes* E.** Common from November to March.
***Eudonia diphtheralis* E.** Very common, recorded from November to April.
***Eudonia dochmia* E.** Uncommon, recorded in October and November.
***Eudonia hemiplaca* E.** Very common, recorded from October to May.
***Eudonia leptalea* E.** Common from November to May.
***Eudonia leucogramma* E.** Only recorded once, on 1 Jan 2018.
***Eudonia minualis* E.** Common from December to April.
***Eudonia octophora* E.** Recorded once, on 14 February 2017.
***Eudonia periphanes* E.** Fairly common, recorded from November to March.
***Eudonia philerga* E.** Commonly recorded from September to December.
***Eudonia pongalis* E.** Common, recorded from November to March.
***Eudonia sabulosella* E.** Common from November to January.
***Eudonia steropaea* E.** Fairly common, recorded from February to April.
***Eudonia submarginalis* E.** Very common from November to March.
***Eudonia trivirgata* E.** Uncommon, recorded twice; 10 Feb 2017 and 24 Sep 2023.
***Eudonia zophochlaena* E.** Fairly common, recorded from November to January.
***Scoparia acharis* E.** Common, recorded from October to February.
***Scoparia* (s.l.) *animosa* E.** Common, recorded from December to February.
***Scoparia chalicodes* E.** Very common, recorded most months of the year excluding mid-winter.
***Scoparia* (s.l.) *illota* E.** Uncommon, recorded twice; February 2023 and February 2024.
***Scoparia* (s.l.) *indistinctalis* E.** Very common, recorded in reasonable number throughout the year.
***Scoparia* (s.l.) *minusculalis* E.** Very common, recorded from October to May.
***Scoparia* (s.l.) *phalerias* E.** Uncommon, recorded twice in February 2017 and twice in December 2024.
***Scoparia ustimacula* E.** Very common, recorded in most months of the year except mid-winter.

Sphingidae (1 species)
***Hippotion scrofa* V.** First recorded in February 2025, when new to New Zealand; see above, under ‘Notable Records’.

Geometridae (67 species)
***Chalastra aristarcha* E.** Common, recorded from September to May.
***Chalastra pellurgata* E.** Common, recorded from September to May.
***Cleora scriptaria* E.** Very common, recorded most months of the year.
***Gellonia dejectaria* E.** Very common, recorded from October to May.
***Gellonia pannularia* E.** Common, records commence in December through to May.
***Declana floccosa* E.** Abundant, can be attracted to light throughout the year.
***Ipana atronivea* E.** Very common and a real stunner. Can be encountered all year round.
***Ipana hermione* E.** Very common, recorded from July to November.
***Ipana junctilinea* E.** Recorded once, on 27 October 2016.
***Ipana leptomera* E.** Very common, recorded most months of the year.
***Ischalis gallaria* E.** Common, recorded from October to May.
***Ischalis nelsonaria* E.** Fairly common, recorded from September to May.
***Ischalis variabilis* E.** Very common, recorded from September to June.
***Pseudocoremia fenerata* E.** Fairly common, recorded from October to June.
***Pseudocoremia leucelaea* E.** Very common from September to June.
***Pseudocoremia monacha* E.** Fairly common in spring and through summer.
***Pseudocoremia productata* E.** Very common in summer and recorded all year round.
***Pseudocoremia rudisata rudisata* E.** Very common from October to June.
***Pseudocoremia suavis* E.** Very common, recorded all year round across the estate.
***Sarisa muriferata* E.** Very common, recorded from September to June.
***Sestra flexata* E.** Common from October to May.
***Sestra humeraria* E.** Uncommon, recorded spasmodically from September to July.
***Anachloris subochraria* N.** Recorded twice, on 1 Feb 2017 and 24 Sep 2017.
***Asaphodes beata* E.** Fairly common, recorded from September to December.
***Asaphodes chlamydota* E.** Common during the summer months.
***Asaphodes limonodes* E.** Two records, both in February 2017.
***Austrocidaria bipartita* E.** Common most months of the year.
***Austrocidaria gobiata* E.** Common from September to December.
***Austrocidaria similata* E.** Very common, recorded most months except mid-winter.
***Chloroclystis* (s.l.) *filata* A.** Common most months apart from mid-winter.
***Elvia glaucata* E.** Fairly common from October to June.
***Epicyme rubropunctaria* N.** Recorded twice, 21 Dec 2023 and 20 Dec 2024.
***Epiphryne verriculata* E.** Uncommon, singular records in November, December and April.
***Epyaxa lucidata* E.** Common, recorded most months.
***Epyaxa rosearia* E.** Common, recorded from September to January.
***Helastia cinerearia* E.** Common from October to May.
***Helastia cymozeucta* E.** Fairly common, recorded from October to December.

***Helastia mutabilis* E.** Fairly common throughout spring and summer months.
***Helastia semisignata* E.** Common, recorded from September to April.
***Homodotis megaspilata* E.** Common most months of the year.
***Hydriomena* (s.l.) *deltoidata* E.** Fairly common, recorded in December and January.
***Hydriomena* (s.l.) *hemizona* s.l. E.** Uncommon, all records in November. Two species may be confused under this name, in which case this is the unnamed one.
***Hydriomena* (s.l.) *rixata* E.** Fairly common, recorded from September to February.
***Microdes epicryptis* E.** Fairly common, recorded from September to January.
***Microdes quadristrigata* E.** Recorded on 5 Feb 2017 and 18 Feb 2025.
***Pasiphila bilineolata* E.** Common, recorded from September to January.
***Pasiphila inductata* E.** Abundant, recorded every month.
***Pasiphila lunata* E.** Recorded once, on 24 Sep 2017.
***Pasiphila melochlora* E.** Uncommon, recorded once on 18 Apr 2023.
***Pasiphila plinthina* E.** Fairly common, recorded in April and July.
***Pasiphila semochlora* E.** Uncommon, recorded in October and April.
***Pasiphila sphragitis* E.** Common from October to July.
***Pasiphila testulata* N.** Very common, recorded from November to March.
***Pasiphila* sp. cf. *magnimaculata* E.** Unnamed pug whose larva feeds on *Brachyglottis kirkii* flowers. First recorded in August 2016 then regularly from August to October.
***Phrissogonus laticostata* A.** Common most months of the year.
***Poecilasthena pulchraria* N.** Abundant all year round.
***Poecilasthena schistaria* E.** Very common all year round.
***Tatosoma agrionata* E.** First recorded in October 2022, by far the most northerly record; subsequently recorded on four more occasions through to January. Larvae are mistletoe feeders.
***Tatosoma apicipallida* E.** First recorded on 11 Oct 2016, followed by occasional observations through spring. At the very north of its range.
***Tatosoma lestevata* E.** Fairly common, recorded on a couple of occasions in October and December.
***Tatosoma tipulata* E.** Very common, found most months except mid-winter.
***Tatosoma topea* E.** Fairly common, recorded from September to December.
***Tatosoma transitaria* E.** Fairly common, recorded from October to June.
***Xyridacma alectoraria* E.** Common, recorded from October to January.
***Xyridacma ustaria* E.** Common, recorded most months of the year.
***Cyclophora obstataria* A.** Recorded once, on 18 Apr 2023.
***Scopula rubraria* N.** Abundant from February until April, mainly during the day but occasionally at light.

Erebidae (10 species)
***Dasypodia cymatodes* A.** Common from late summer through autumn.

***Hypocala deflorata* V.** First recorded in January 2018, subsequently in February and March 2023 and then February 2026.
***Nyctemera amicus* A.** Both true *N. amicus* (Australian) and the hybrid with the endemic *N. annulata* seem to occur at Mahakirau, based on wing pattern elements of adults, as compared with vouchers in NZAC. Further work on these species and their hybridisation is currently being conducted by W.T. Frost (Lincoln University).
Nyctemera annulata x amicus.
***Pantydia sparsa* A.** Very common at the moth trap, particularly late summer/early autumn.
***Rhapsa scotosialis* E.** Very common all year round.
***Schrankia costaestrigalis* N.** Can be found commonly throughout the year.
***Trigonistis anticlina* E.** Common, recorded from October to December.
***Utetheisa lotrix lotrix* V.** Recorded once, in April 2023.
***Utetheisa pulchelloides vaga* V.** First recorded at light in March 2023 then subsequently in January 2024.

Noctuidae (46 species)
***Bityla defigurata* E.** Very common from January to April.
***Helicoverpa armigera conferta* A.** Very common from January to April.
***Helicoverpa punctigera* V.** First recorded in October 2023 then February 2024.
***Austramathes purpurea* E.** Common, recorded from spring through to late summer.
***Cosmodes elegans* A.** Regularly attracted to light from September to March. Usually an autumn immigrant from Australia.
***Proteuxoa tetronycha* E.** Very common at light from October to end of summer.
***Peripyra sanguinipuncta* A.** Very common from January to early autumn.
***Physetica prionistis* E.** Only recorded twice; November and January. A rarity this far north (Hoare 2019).
***Physetica sequens* E.** Recorded once on 27 January 2026.
***Ichneutica arotis* E.** Uncommon; both recorded in October.
***Ichneutica atristriga* E.** Very common throughout the summer.
***Ichneutica brunneosa* E.** First recorded in October 2016 and then on 6 further occasions throughout summer months.
***Ichneutica chlorodonta* E.** Common, can be found throughout the year.
***Ichneutica inscripta* E.** Two recorded; October and November.
***Ichneutica insignis* E.** Abundant in summer, recorded all year round.
***Ichneutica lignana* E.** Very common throughout the year.
***Ichneutica mustulenta* E.** Fairly common from January to March.
***Ichneutica plena* E.** Very common, recorded throughout the year.
***Ichneutica mutans* E.** Abundant in summer, recorded all year round.
***Ichneutica purdii* E.** First recorded at light in February 2016 and then December 2024 but not a common moth.

***Ichneutica semivittata* E.** Common from September to February.
***Ichneutica steropastis* E.** Common from September to March.
***Ichneutica subcyprea* E.** This recently named species was formerly confused with *I. chlorodonta*; the differences are pointed out by Hoare (2019). A further example was recorded in September 2023.
***Ichneutica sulcana* E.** Common across the summer months.
***Ichneutica ustistriga* E.** Very common and can be found all year round.
***Feredayia graminosa* E.** Abundant in summer, recorded all year round.
***Meterana* n. sp. nr *asterope* E.** First recorded on 5 November 2023 then 24 October 2025. Unnamed species near *M. asterope*, sparsely recorded through the North Island.
***Meterana alcyone* E.** Recorded for the first time in December 2025.
***Meterana coeleno* E.** Recorded from August to October.
***Meterana decorata* E.** First record from November 2023, now regularly encountered.
***Meterana diatmeta* E.** Uncommon, recorded from October to December.
***Meterana stipata* E.** Found from September to January but not in large numbers.
***Meterana vitiosa* E.** Common from autumn until spring.
***Persectania aversa* E.** First recorded in April 2017, then twice more in October and February.
***Mythimna separata* N.** Common, recorded all year round.
***Leucania stenographa* A.** Fairly common, recorded in April and May.
***Athetis tenuis* A.** Fairly common, can be found from December to May.
***Spodoptera litura* A.** Abundant from late summer to autumn.
***Agrotis ipsilon* N.** Abundant from late summer to autumn.
***Agrotis munda* A.** First and only record in December 2024.
***Diarsia intermixta* N.** Fairly common throughout the summer months.
***Chrysodeixis argentifera* A.** Occasional migrant first recorded in December 2016.
***Chrysodeixis eriosoma* N.** Common across the summer to early autumn.
***Ctenoplusia albostriata* A.** Fairly common from March to May.
***Ctenoplusia limbirena* A.** Very common from January to June.
***Thysanoplusia orichalcea* A.** Fairly common from January through to May.

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Observations on a larval population of the Mountain Giant dragonfly *Uropetala chiltoni* Tillyard, 1921 (Odonata: Petaluridae) at Cass, inland Canterbury

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Introduction

Two articles in Volume 59 of The Wētā (Merien 2026, Mackenzie Dodds & de Koenigswarter 2026), deal with aspects of the biology of *Uropetala carovei* (White, 1843) (Odonata: Petaluridae), which along with *U. chiltoni* Tillyard, 1921 are New Zealand's largest dragonflies. A third article discusses the biogeography of the genus *Uropetala* in the context of tectonic uplift (Grehan & Marinov 2026). *Uropetala chiltoni*, is the subject of this note, which presents unpublished observations on a larval population made by the authors starting in the 1980s. Given the resurgence of studies on *Uropetala*, our findings may be of interest to readers.

Uropetala chiltoni was described by Tillyard (1921) from material collected in 1920 at Arthurs Pass and Cass in the central South Island. Tillyard stayed at the University of Canterbury's mountain biological station at Cass and named his new species after his host Dr Charles Chilton, Professor and Head of the Department of Biology at Canterbury College. The larvae of *U. chiltoni* (Fig. 1) live in burrows in forest clearings and treeless, upland bogs, unlike those of *U. carovei,* which are usually found in the banks of shaded, forest streams (Rowe, 1987). An intensive study of *U. chiltoni* was made by Wolfe (1953) who examined

aspects of its life history, ecology and breeding biology at several localities in the vicinity of Cass. Our investigations expand on his work.

At one of Wolfe's study sites close to the eastern shore of Lake Sarah (43° 2.80' S, 171° 46.62'E), we examined the distribution of larval burrows, measured their openings and made observations on habitat features and associated fauna. Our original aim was to monitor changes in the numbers and sizes of burrows over time, but this objective was not met as too many burrow markers were lost and one of us (RJR) emigrated to Australia. Nevertheless, we did obtain some interesting data, which add to our knowledge of this iconic dragonfly. They provide the basis of this note.

Methods

In November 1981, two adjacent quadrats, (each 25 m^2) were defined with pegs at the study site close to the shore of lake Sarah at an elevation of about 580 m above sea level. The site was dominated by seepage and a gently sloping bog surrounded predominantly by tussock grassland. The surface comprised a patchwork of tussocks (*Schoenus*), mosses (including *Sphagnum*), liverworts, fine gravel, mud and a few small shrubs (Fig. 2). The roughly circular entrances to burrows of *U. chiltoni* were counted and marked with garden plant labels that were pushed into the ground. Counts were repeated in March 1984 and the diameters of 17 burrow entrances of various sizes were determined. Larvae were extracted from these burrows and their body lengths and head widths (across the eyes) were measured.

In March 1999, a further survey was undertaken within the same area, but burrow entrances were counted in 42, 1m^2 quadrats laid out in rows across the site. The diameters of all entrances were measured and the distance of each opening to the nearest tussock was calculated. This latter measurement was made because tussocks provide the main emergence sites of *U. chiltoni* at the Cass sites and adult females alight on them before moving down towards the ground to oviposit (Wolfe 1953). The nature of the habitat immediately surrounding each burrow was also noted as, either, bryophyte (*Sphagnum*, other mosses, liverworts), tussock, shrub, mud and gravel, or combinations of them.

Figure 1: The final instar larva of *Uropetala chiltoni* drawn by Karen Mason for MJW.

Figure 2: The late Richard Rowe (centre) At the study site in 1983.

Additionally, the ground-dwelling invertebrate fauna at the site, and therefore what potential prey species of *U. chiltoni* larvae were present, was investigated using pitfall traps left out for various periods of time. Traps were glass jars (500 ml, 75 mm diameter) containing ethylene glycol as preservative, which were buried with their openings flush with the ground surface. Lastly, gut content analyses were made on 9 *U. chiltoni* larvae collected from the site in March 1982 and March 1984, including two from pitfall traps. Some gut contents were mounted on slides and examined microscopically to aid identification.

Results and discussion

Distribution and abundance of burrow entrances

The 17 larval burrows measured in 1984 ranged in diameter from 8 – 36 mm. Larvae taken from these burrows were 14 – 45 mm long and had head widths of 4.5 – 12 mm. Body length, head width and burrow entrance diameter were strongly correlated (Table 1). Rowe (1987) stated that burrow diameter was about half the length of the occupant, whereas the length: burrow diameter ratio of our 17 larvae was somewhat greater than

that (mean ratio 0.67 ± SD 1.0). Mean head width, in contrast, was 0.42 of burrow diameter, and therefore very similar to the ratio indicated by the photographs of *U. carovei* in the article by Mackenzie Dodds & de Koenigswarter (2026), which show the heads and forelegs of larvae at burrow entrances.

Table 1: Pearson correlations (r) between body length, head width and burrow entrance diameter for 17 *U. chiltoni* larvae.

	Head width	Burrow diameter
Body length	0.92	0.85
Head width	-	0.85

In 1981, 124 and 62 burrow entrances were found in the two 25 m^2 quadrats, respectively, whereas in 1984 the same quadrats contained 118 and 53 entrances. In 1999 128 burrow entrances were located in the 42, 1m^2 quadrats examined. Mean densities of burrows in the three years were therefore 3.7, 3.4 and 3.05 per m^2. Because some burrows can have two entrances as noted by Wolfe (1953) and Rowe (1987), counts of openings may overestimate the actual numbers of burrows present.

The 1999 data set enabled further analysis of the distribution patterns of burrows within the site. They had a highly clumped distribution (variance to mean ratio 5.63) and nine of the 42 quadrats were empty. In contrast, 16 and 17 entrances were found in two quadrats. The plot of burrow numbers versus burrow entrances diameter (Fig. 3) shows major peaks between 6–7 and 8–9 mm and three smaller peaks, all of which may represent age/year classes of larvae.

The most common immediate surrounds of burrow entrances were bryophytes (*Sphagnum*, other mosses and liverworts), and tussocks, either alone or together (Fig. 4). The surrounds of another 5% of burrows comprised bryophytes and either grass, mud or a shrub, and 6% had a tussock and either grass, mud or a shrub in their immediate vicinity. Mud, a shrub or grass alone, surrounded the remainder of the burrow openings. Most burrow entrances were close to a tussock or less frequently a shrub.

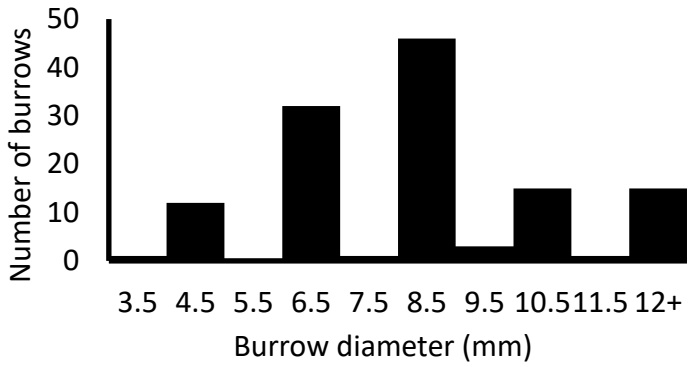

Figure 3: Numbers of burrow entrances in ten 1 mm diameter size classes at the study site in 1999.

Figure 4: The percentages of plants and other materials forming the immediate surrounds of 128 burrow entrances present in the 42 quadrats examined in 1999. Bryo = bryophytes, Tuss = tussock, B + T = bryophyte and tussock.

Thus, 81 of the 128 openings were within 20 mm of a plant and only six were more than 120 mm away from one.

Wolfe (1953) did not estimate burrow density at any of his sites, but his comment that their entrances were most commonly found at the base of *Schoenus* tussocks and amongst bryophytes, is consistent with our findings. He also found them in shingle, at the edge of well-defined bog streams and in clay banks, but these habitat features were not present at our site.

Associated invertebrate fauna

Pitfall trapping indicated that a diverse invertebrate fauna was present at the site (Table 2). Thus, 584 invertebrates belonging to four phyla were taken in total. Ninety percent were arthropods, including insects (70%) and spiders (19%). Small numbers of flatworms, earthworms, small land snails, slugs and the aquatic snail *Potamopyrgus antipodarum* were also trapped. Larval and adult dipterans, and the hemipteran *Microvelia mcgregori*, which lives on the water surface, were the most abundant insects. Four *U. chiltoni* larvae, three of which were in the final instar, were found in the traps.

Gut contents of *Uropetala chiltoni* larvae

Of the nine larvae we dissected, seven were final instars, one a penultimate instar (F-1) and one a smaller individual. Eight of the larvae contained gut contents and one had an empty gut. Fragments of a spider were present in three larvae, while earthworms, identified by their chaetae, were found in two. The only other prey items seen were a mite, a weevil, a chironomid midge larva and unidentified fragments of a dipteran and a beetle. Small plant fragments were present in four larvae, two of which also contained the remains of earthworms. Therefore, it is possible that the pieces of plant came from the guts of the worms. However, the only animal material in the guts of the other two larvae, which contained plant fragments were a spider and a dipteran, neither of which would be expected to ingest plant matter. Winstanley & Rowe (1980) noted that Harding (1977) had also found ‘vegetable material’ in the guts of *U. carovei* larvae and speculated they may have been pieces of liverwort trimmed by the larvae around their burrow entrances.

Table 2: Total numbers of invertebrate taxa (n) taken in pitfall traps at the Lake Sarah study site in October and November 1981, March 1982 and March 1984.

Taxa	n	Taxa (continued)	n
Platyhelminthes		**Coleoptera** (beetles)	
Geoplanidae (flatworms)	7	Curculionidae adults	1
Annelida		*Liodessus deflectus* (Dytiscidae)	10
Lumbricidae (earthworms)	9	*L. deflectus* larvae	1
Mollusca		Scirtidae larvae	26
Potamopyrgus antipodarum (snails)	7	Hydrophilidae larvae	6
Small land snails	10	Staphylinidae	3
Athoracophoridae (slugs)	2	**Hemiptera** (bugs)	
Crustacea		*Microvelia mcgregori* (Veliidae)	130
Ostracoda (seed shrimps)	3	Aphididae	2
Arachnida		Aphrophoridae	6
Araneae (spiders)	110	Other Hemiptera	5
Pseudoscorpionida (pseudoscorpions)	1	**Diptera** (flies)	
Acari (mites)	1	Stratiomyidae larvae	14
Opiliones (harvestmen)	1	Chironomidae larvae	13
Collembola (springtails)	24	Mycetophilidae adults	1
Insecta		Psychodidae larvae	1
Odonata: *Uropetala chiltoni* larvae	4	Tipulidae larvae	43
Plecoptera (stoneflies)		Tipulidae adults	5
Notonemoura latipennis adults	54	*Paradixa* (Dixidae) larvae	1
N. latipennis larvae	1	*Neolimnia sigma* (Sciomyzidae) adults	1
Trichoptera (caddis)		*N. sigma* larvae	2
Pseudoeconesus hudsoni adults	2	Other Diptera	98
Psilochorema mimicum adults	1	**Hymenoptera** (wasps)	1
P. mimicum larvae	1	**Lepidoptera** (moths)	
Oxyethira albiceps adults	1	Noctuidae adults	3
O. albiceps larvae	1	Caterpillars	2
Orthoptera		Other moths	5
Pteronemobius sp. (crickets)	1		
Blattodea (cockroaches)		**Total**	584
Celatoblatta sp.	1		

Wolfe (1953) found that the food of late instars of *U. chiltoni* comprised ground-dwelling insects and arachnids, while Mackenzie Dodds & de Koenigswarter (2026) observed that insects and worms were eaten by larvae of *U. carovei*. In their study, all spiders that were attacked got away, however, like us, Wolfe (1953) found that *U. chiltoni* had successfully ingested spiders. Because larvae were never seen to leave their burrows in the *U. carovei* study, successful attacks occurred only when prey were very near a burrow. We do not know if the same is true for *U. chiltoni*, and a study with continuously-running cameras as done by Mackenzie Dodds & de Koenigswarter (2026) may well be informative. Although the final instar larvae we found in pitfall traps may have been individuals searching for emergence sites, the presence of a smaller, trapped individual suggests that at least some larvae may not be confined to their burrows at all times.

Acknowledgments

We thank the Zoology students from the University of Canterbury who assisted in the field, in particular the fourth year Limnology class of 1999. The authors made these observations together in the 1980s, and although Richard Rowe provided input to the 1999 study, he did not participate in it as he was living in Australia.

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Distinguishing features of male and female Forest Ringlet (*Dodonidia helmsii*) butterflies.

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Abstract

Almost 130 museum specimens and dorsal photographs of the Forest Ringlet butterfly (*Dodonidia helmsii*, Butler 1884) are examined. Measured variations in butterfly size, abdomen dimensions and wing markings are evaluated to assess which, if any, of these attributes may aid identification of male and female of the species. Abdomen dimensions are found to be the most reliable discriminator, with females having a mean abdomen length: width (L:W) ratio of 3.54+/-0.59, whilst males have a mean L:W ratio of 8.10+/-1.39. There was no overlap between the sexes. Measured wingspans ranged from 39.77-57.56mm and forewing lengths from 22-30mm. Whilst most females are larger, being >47mm wingspan and >25mm forewing length, there is considerable overlap between the sexes, so size alone is considered unreliable for gender recognition, at least for samples across a wide geographic area. *Dodonidia helmsii* forewing markings show subtle but distinct differences between the sexes based on the extent and connectivity of yellow-orange bands. Used in conjunction with the abdomen L:W ratio, the pattern of forewing markings is considered a highly reliable way of confirming the sex of individuals. This improved confidence in gender recognition will greatly aid our observation and understanding of gender-related behavioural differences and ecological knowledge of the species.

Introduction

The Forest Ringlet butterfly (*Dodonidia helmsii*, Butler 1884) is endemic to Aotearoa New Zealand and is classified as "Threatened – Nationally Vulnerable" (Hoare *et al*., 2026). A detailed report by Barker & Smerdon (2024) presented new research findings and provided an overview of all previous work on the species. Even so, one aspect that remains poorly documented in the literature is characterisation of differences, if any, in markings and morphology of male and female of the species. Defining such differences, if they exist, could be important for understanding gender specific behavioural traits, reliable definition of male and female flight periods, and population dynamics at individual sites. The present study, based on first-hand observation of museum specimens and photographs aims to determine those features that are most reliable for the correct identification of male and female of the species.

Early descriptions and illustrations

The holotype of *D. helmsii*, a male (Fig.1a), was first described by Fereday (1882) based on a specimen collected by Mr R. Helms in the Paparoa Ranges in 1881. The Fereday (1882) description gives a full account of wing markings, venation and other features, but fails to define the genus, and consequently his naming of the species was invalid. It was Butler (1884a, b), therefore, who by assigning the species to a new genus, *Dodonidia*, is recognised as the person who first named the species. Whilst Butler (1884a) provides an underside view of the wing markings in an illustration depicting the species in typical resting position with wings closed (Fig.1b), the Fereday (1882) paper has no illustration.

The first illustration of the Forest Ringlet showing a dorsal view is the beautiful watercolour painting by A.P. Buller (1893), now held at the New Zealand National Library (Fig.2). Although this wonderful depiction of the Forest Ringlet does not specify the gender of the two individuals illustrated, the short plump abdomen and connectivity of orange areas on both forewing and hindwing upper sides are features typical of females of the species, as the present study will explore further. An illustration (Fig.3) by Marshall (1895), although not of the same quality as Buller (1893) is perhaps the first example to illustrate the distinctive wing

markings and abdomen of the male Forest Ringlet, although not actually stated as such. The first illustrations depicting and identifying male and female *D. helmsii* (Fig.4) are those of Hudson (1898) and Hudson (1928). In terms of description of variations in markings and size of *D. helmsii*, Hudson (1898) and reiterated in Hudson (1928), states that: *This insect appears to vary a little in the extent of the yellowish-orange colouring of the upper side. It also varies in size, specimens from the North Island* [principally from Silverstream] *being slightly larger than those from the South Island* [principally from Dun Mtn.]. Hudson does not, however, attribute any of the variations in size or markings to differences between male and female.

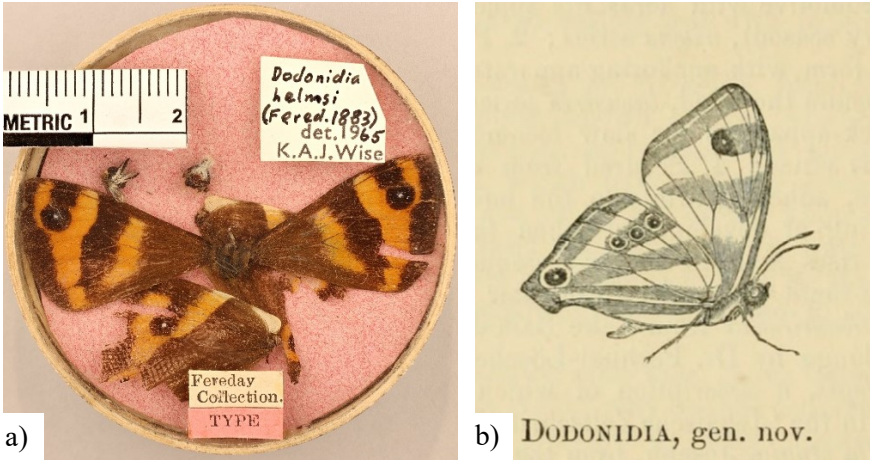

Figure 1: a) Type specimen of *Dodonidia helmsii* (male) held in collections of Canterbury Museum, New Zealand (CMNZ); ref: CMNZ 2007.212.216. Photographed 01.05.2026. (Image courtesy of Johnathon (Johno) Ridden (CMNZ)). **b)** The first published illustration of *Dodonidia helmsii*, by Butler (1884a).

The next published account to give a detailed description of the species is that of Gibbs (1980), who says (pg.78) that: *Local variations in size are noticeable in this species, but its colouration and markings are more constant than our other satyrines*. He goes on to say (pg.79), that: *Both sexes are alike and may be distinguished only by the shape of the abdomen which is long and slender in the male and shorter and stout in the female*.

Smerdon & Barker (2024) also state that abdomen shape is a reliable feature to distinguish male from female and were the first to suggest that forewing upperside markings may also aid gender recognition (see Smerdon & Barker, 2024, Fig.15).

Figure 2: Watercolour painting of *Dodonidia helmsii* adopted from A.P. Buller (1893). The short plump abdomen and connectivity of orange areas on the wings of the example illustrating dorsal view are typical of a female. (Image © New Zealand National Library. Buller, Arthur Percival, 1866-1910. Buller, Arthur Percival, 1866-1910: *Dodonidia helmsii*, 1893. Buller, Arthur Percival 1866-1910: New Zealand Lepidoptera. [1893]. Ref: E-088-q-012. Alexander Turnbull Library, Wellington, New Zealand. /records/23224737).

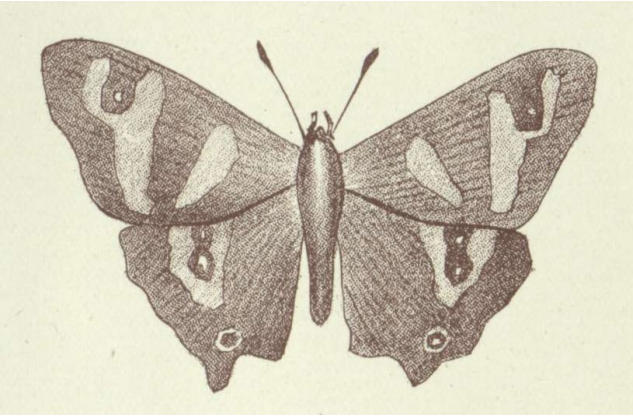

Figure 3: Dorsal view of *Dodonidia helmsii*. The thin elongate abdomen combined with depiction of the upper forewing innermost (orange) lensoid patch separated from large outermost (orange) band is typical of a male. (Illustration adopted from Marshall 1895).

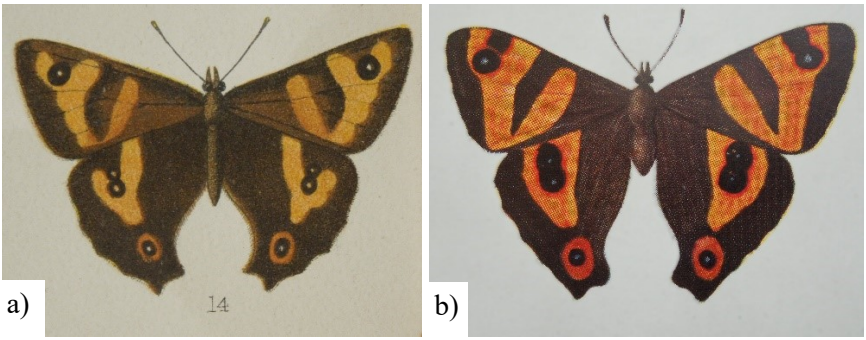

Figure 4: *Dodonidia helmsii* illustrations. **a)** described as a **male** showing a long thin abdomen (Hudson 1898, Pl.XI, Fig.14). **b)** defined as a **female** (showing a short plump abdomen (Hudson 1928, Pl.IV, Fig.16).

Methods

The present study considers variations in butterfly size, abdomen dimensions and upperside wing markings (forewing and hindwing) to evaluate the extent to which these attributes may aid identification of male and female *D. helmsii*. It is based on examination of 97 museum specimens (72 measured), and a further 32 dorsal view photographs.

Results

Abdomen length (L) and width (W)

As stated by Gibbs (1980) for male *D.helmsii*, the abdomen is proportionately longer and thinner, and in females it is shorter and plump (Fig.4). Figure 5 summarises the data from all *D. helmsii* museum specimens measured to date (n=62), based on abdomen length and maximum abdomen width. There are two distinct groupings, the upper (red dots) with generally shorter and wider abdomens are females, and the lower grouping (blue dots) with longer and narrower abdomens are males. It can be noticed that there are many more male than female specimens in museum collections. This was also noted by Gibbs (1980), who gave a male:female ratio of 1.7:1. The present study found the m:f museum specimen ratio to be even higher, at 1.85:1, whilst in photos of wild individuals the m:f ratio is 0.78:1 (ie more females photographed). This probably indicates that females are more easily accessible for dorsal view photographs at low level as they spend more time lower down egg-laying on *Gahnia spp*. and resting on lower-level vegetation, whereas males spend more time higher up. The reason for almost twice as many males than females in museum specimens may be due to collectors obtaining their specimens early in the season to obtain fresh individuals, at a time when proportionately more males would be on the wing.

Based on Fig.5 and the discussion above, there are clear differences in L:W ratios. This is helpful as a ratio is dimensionless so that even for specimens and images without a scale-bar it is possible to define male and female of the species. A plot of *D. helmsii* abdomen ratios (Fig.6), including additional data from photographs of live (and museum) specimens without scale, shows two distinct groupings, with females (red) showing the smaller abdomen L:W ratio associated with short plump abdomens, and males (blue) showing larger L:W ratios associated with long thin abdomens. There is no overlap in the data, with mean L:W ratios of 3.54 +/- 0.59 for females and 8.10 +/- 1.39 for males. This, therefore, is undoubtedly a reliable way to distinguish male from female *D. helmsii*.

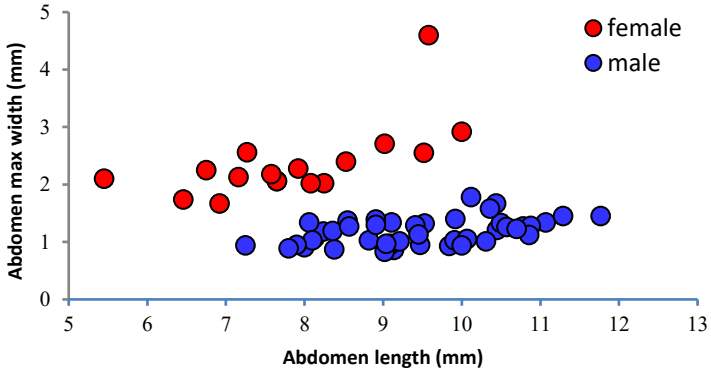

Figure 5: Forest Ringlet (*Dodonidia helmsii*) abdomen dimensions based on 62 museum specimens (male = 45, female = 17).

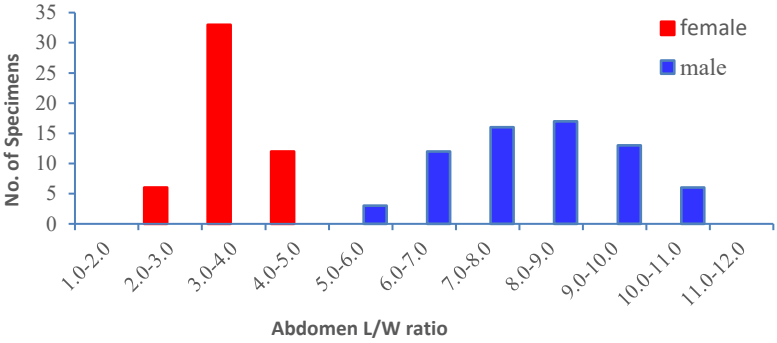

Figure 6: Forest Ringlet (*Dodonidia helmsii*) abdomen L/W ratios, based on 118 museum specimens and photographs (male = 67, female = 51).

Butterfly size (wingspan and forewing length)

Wingspan and forewing length are traditionally measured for butterflies. Of these, forewing length is perhaps the most reliable measurement for museum specimens as wingspan can vary slightly according to how the specimen is set. Based on measurements of 75 museum specimens or high-quality images with accompanying scale-bar, wingspans were found to vary from 39.77 - 57.56mm, with a mean of 48.13 +/- 4.00mm. Whilst Gibbs (1980) reports one specimen from the Waitakere Ranges, Auckland, with a wingspan of 64mm, this is exceptional. Those from Ruapehu and Wellington averaged 53mm, and those from Dun Mountain, Nelson, just 44mm, with a minimum of 40mm. Most specimens examined during the present study have wingspans in the range 41-56mm, with a maximum wingspan of c. 58mm. As expected, Figure 7 shows a strong linear relationship between forewing length and wingspan, but this is to be expected as they are not independent variables.

Having established that abdomen L:W ratios present a reliable way of distinguishing male and female *D. helmsii* (see Fig.6), the gender was thus defined for each museum specimen. A few specimens had gender defined on the specimen labels, and in all such cases this agreed with the definition based on abdomen L:W ratio.

Figure 8 plots the same data as Figure 7, but with male (blue dots) and female (red dots) plotted separately to allow an assessment of any differences. Whilst all bar two female specimens have wingspans >47mm (forewing length >25mm), there is so much overlap between male and female specimens that wingspan and forewing length are not going to be a reliable means of identifying male and female specimens. Intuitively, one might have expected that females would be larger, and whilst this might still be the case in specific geographic areas, it is not apparent from the presently available data. A greater sample size split according to different geographic areas will be required to resolve this.

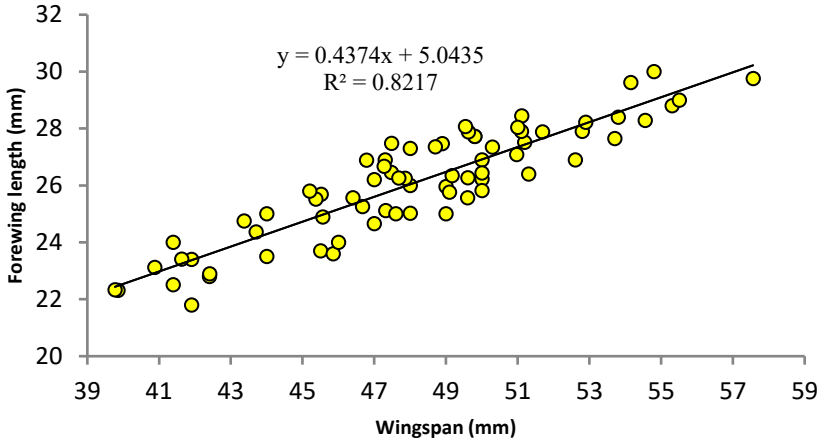

Figure 7: *Dodonidia helmsii* forewing length versus wingspan of both sexes for 72 museum specimens.

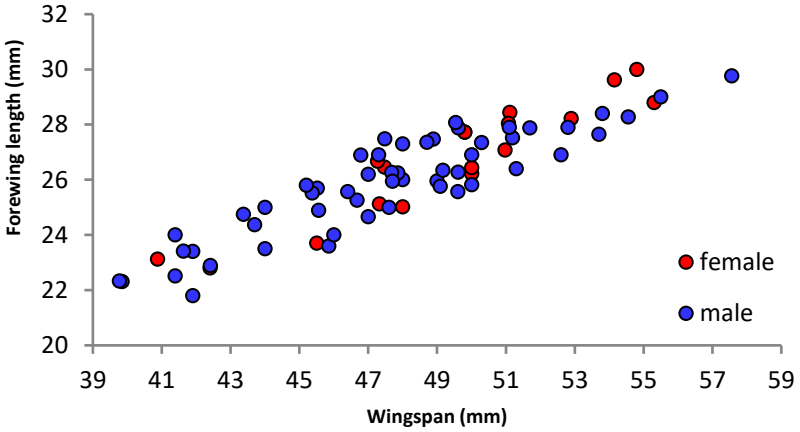

Figure 8: The same forewing length versus wingspan plot as Fig.7, but with the data set split according to male (blue dots; n=54) and female specimens (red dots; n=18).

Differences in forewing and hindwing markings

Having already considered differences in abdomen dimensions, wingspan and forewing length as potential ways to distinguish male and female *D. helmsii* specimens, it was concluded that only abdomen dimensions offer a reliable approach to distinguishing gender. Let us now consider, whether differences in forewing and hindwing markings might offer an additional means of confirming male or female specimens.

Forewing markings

Barker & Smerdon (2024) have already suggested that certain forewing markings may offer an additional way of distinguishing male from female, with males typically having the innermost orange band isolated from the remaining orange areas of the forewing by a central brown band extending from the costa to the very base of the forewing (Fig.9a-c). This is referred to as Forewing Type 1 (FW1). In the case of clear-cut females, all orange areas are connected, as the brown band terminates well before the base of the forewing (Fig.9g-i). This is referred to as Forewing Type 3 (FW3). There are a few examples that are less clear-cut and appear somewhere between the two end-members (Fig.9 d-f). These are referred to as Forewing Type 2 (FW2).

Hindwing markings

As well as variations in forewing markings, it has also been recognised that the hindwing markings of *D. helmsii* also show slight variation. Principally this is regarding whether the basal extensions of the "V" or "Y"-shaped orange area extends all the way to the orange of the termen ocellus. Again, there are three possibilities, the first being hindwings showing a clear gap between the termen ocellus and the orange "V" or "Y" area (Fig.9a,d,g), referred to as Hindwing Type 1 (HW1). The other end of the spectrum is hindwings showing a clear connection between the termen ocellus and the orange "V" or "Y" area (Fig.9c, f, i). This type is referred to as Hindwing Type 3 (HW3). The final hindwing category, referred to as Hindwing Type 2 (HW2) lies between HW1 and HW3 and exhibits a weak extension of the orange of the main area to the outer orange ring of the termen ocellus. This is often seen as a scattering of orange scales within the intervening brown area (Fig.9h).

Combined forewing and hindwing markings

Figure 9 illustrates a matrix of nine combinations of forewing-hindwing markings for *D. helmsii* specimens, ranging from Forewing Type 1 – Hindwing Type 1 (simplified notation 1-1), to Forewing Type 3 – Hindwing Type 3 (simplified notation 3-3). Having established this simplified notation, the forewing-hindwing combinations of each museum specimen and photograph were then determined by visual inspection. Assigning gender to each individual based on abdomen ratio, a tally of male and female wing markings in each of the nine possible types was then determined. The findings are summarised in Table 1.

Table 1: Number of upper side forewing-hindwing marking combinations as illustrated in Fig.9.

	Female				Male			
	HW1	HW2	HW3	N	HW1	HW2	HW3	N
FW1	0	3	2	5	40	11	9	60
FW2	3	6	3	12	6	0	2	8
FW3	4	9	21	34	0	0	0	0
N	7	18	26	51	46	11	11	68

Table 1 shows that the 1-1 combination (Fig.9a) only occurs in males, and 88% of males (60 out of 68) have the FW1 forewing type with the innermost lensoid patch of orange enclosed by brown and disconnected from the main outer Y-shaped orange band. Eight male specimens (12%) had the FW2 type of Forewing pattern, but no male specimens had the FW3 type with all orange areas fully connected.

Two-thirds of female specimens, 34 out of 51 (67%) had FW3 forewing type (not seen in males), with twelve (24%) showing the intermediate FW2 forewing type, and five (10%) showing the FW1 forewing type more characteristic of males (although none showing the 1-1 combination). With regard to hindwings, 26 of 51 females (51%) showed the HW3 hindwing type, compared with just 11 of 68 males (16%) and the combination FW3-HW3 (ie 3-3) was exclusive to females, being observed in 21 female specimens (41%) and not recorded in any male specimens.

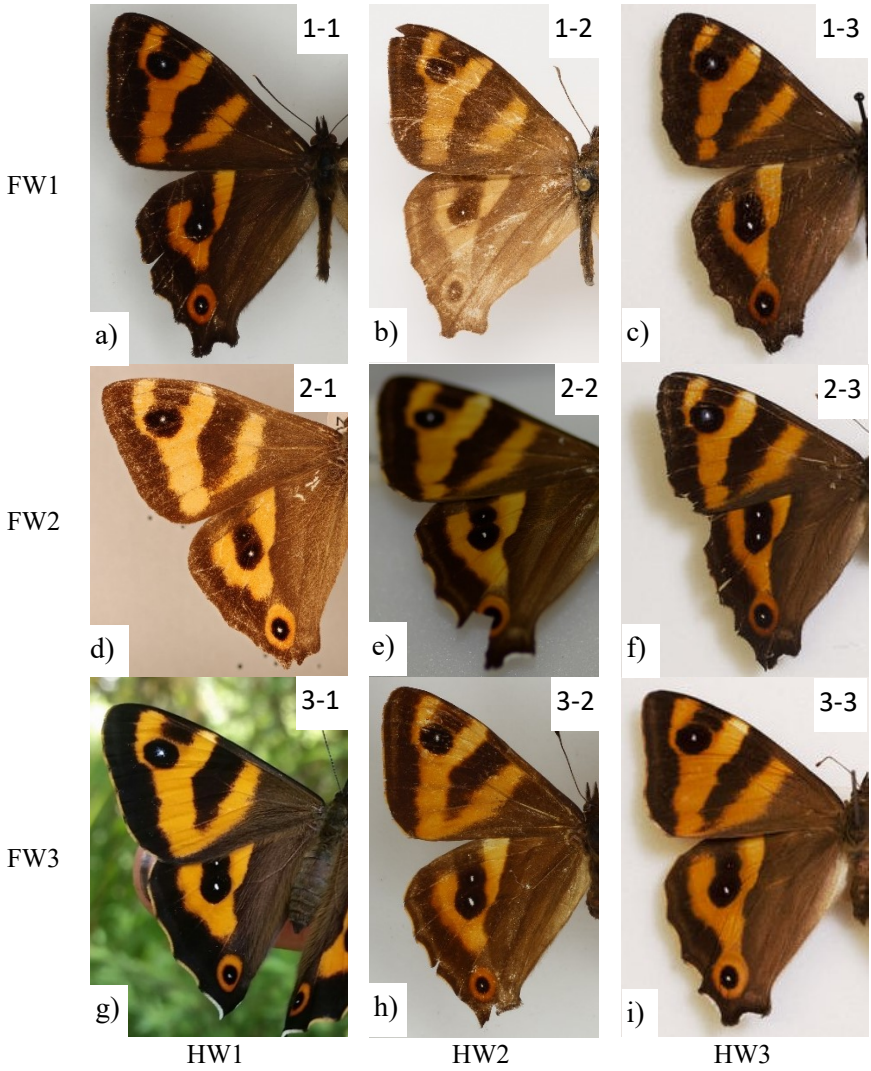

Figure 9: Differences in *Dodonidia helmsii* forewing and hindwing markings. The top row (a-c) is Forewing Type 1 (FW1) (forewing inner orange patch completely enclosed by dark brown. Second row (d-f) is Forewing Type 2 (FW2) (narrow connection between forewing inner orange patch and outer orange band). Third row (g-i) is Forewing Type 3 (FW3) (forewing orange patches completely connected). The first vertical column (a,d,g) in the grid is of hindwings with the main orange patch disconnected from the tornus ocellus (HW1). The second column (b,e,h) is of hindwings showing a feint connection of the main orange >>>

area with the tornus ocellus (HW2). The third column (c,f,i) is of hindwings with the main area of orange connected to the orange of the tornus ocellus (HW3). The grid of images shows nine possible combinations, from 1-1 top left (only seen in males) to 3-3 lower right (only seen in females). With rare exceptions, the FW1 types (top row) are only seen in males and FW3 types (bottom row) typify females. Specimen details and credits: **(a)** NHMUK 015199772 (Natural History Museum, London, UK) © Trustees of the British Museum [Creative Commons License 4.0 (https://creativecommons.org/licenses/by/4.0/]; **(b)** AMNZ22732 (Auckland Museum, NZ); **(c)** A102.357 (Puke Ariki, New Plymouth, NZ); **(d)** CMNZ 207.212.219 (Canterbury Museum, NZ); **(e)** Andy Barker ref: NZAC 01b (no institutional accession number), (New Zealand Arthropod Collection, Landcare Research, Auckland, NZ); **(f)** A102.358 (Puke Ariki, New Plymouth, NZ); **(g)** iNaturalist record (12 Feb 2015) © Catherine Beard; **(h)** NHMUK 015199779 (Natural History Museum, London, UK) © Trustees of the British Museum [Creative Commons License 4.0 (https://creativecommons.org/licenses/by/4.0/]; **(i)** A102.356 (Puke Ariki, New Plymouth, NZ).

Figures 10 and 11 present for the first time the only known examples of museum specimens of *D. helmsii* where the abdomens have been removed and slides made to confirm gender and illustrate genitalia characteristics of male (Fig.10) and female (Fig.11). Notice how the wing pattern of the male (Fig.10a) shows the typical 1-1 combination (Fig.9a & Table 1) that the present study has shown to be most characteristic of male individuals. The *Dodonidia hemsii* type specimen (Fig.1a), described by Fereday (1882) as a male, also shows the Type 1 forewing pattern that is characteristic of males.

In the case of the female specimen with abdomen removed (Fig.11), the wing pattern combination is Type 3-2 (bordering on Type 3-3). Forewing Type FW3 (Fig.9g-i & Table 1) is typical of females, and the hindwing Type HW2 is the second most common hindwing pattern of females, indeed, it is bordering on Type HW3, which is the commonest female hindwing pattern. The combination Type 3-3 is only seen in females.

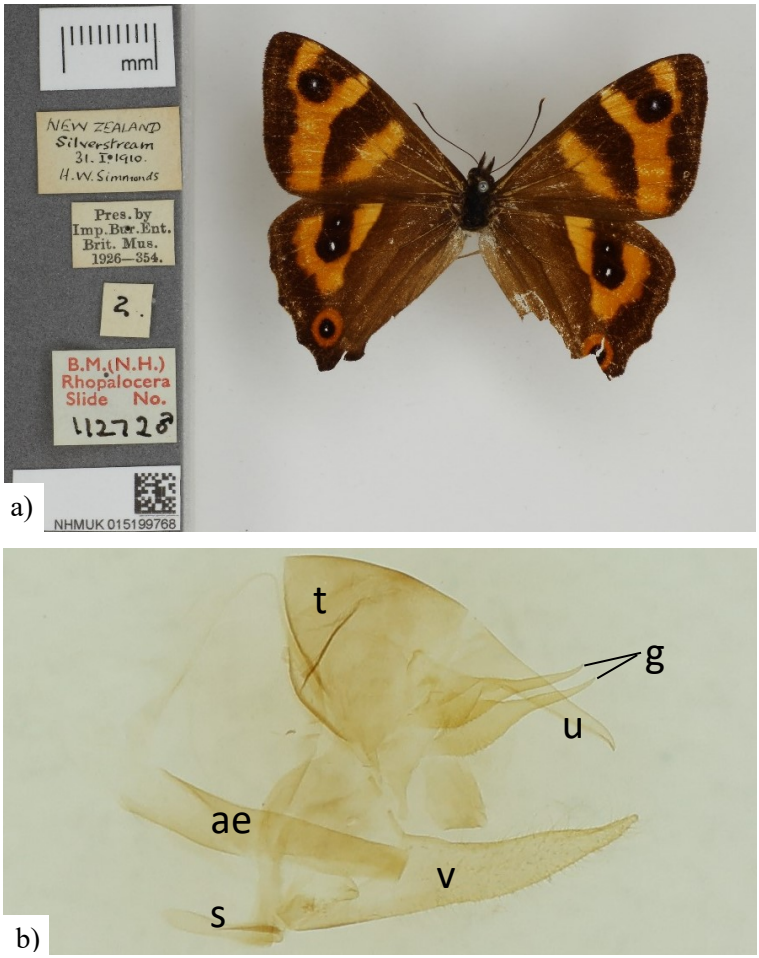

Figure 10: a) *Dodonidia helmsii* (male) (NHMUK 015199768); abdomen removed to make slide (BMNH 11272); **b)** lateral view of *Dodonidia helmsii* male genitalia [Slide BMNH 11272, stained] from abdomen of specimen shown in a). Images ©Trustees Natural History Museum, London) [Creative Commons License 4.0 (https://creativecommons.org/licenses/by/4.0/]. Labels added: ae = aedeagus, g= gnathos, s= saccus, t= tegumen, u=uncus, v=valva.

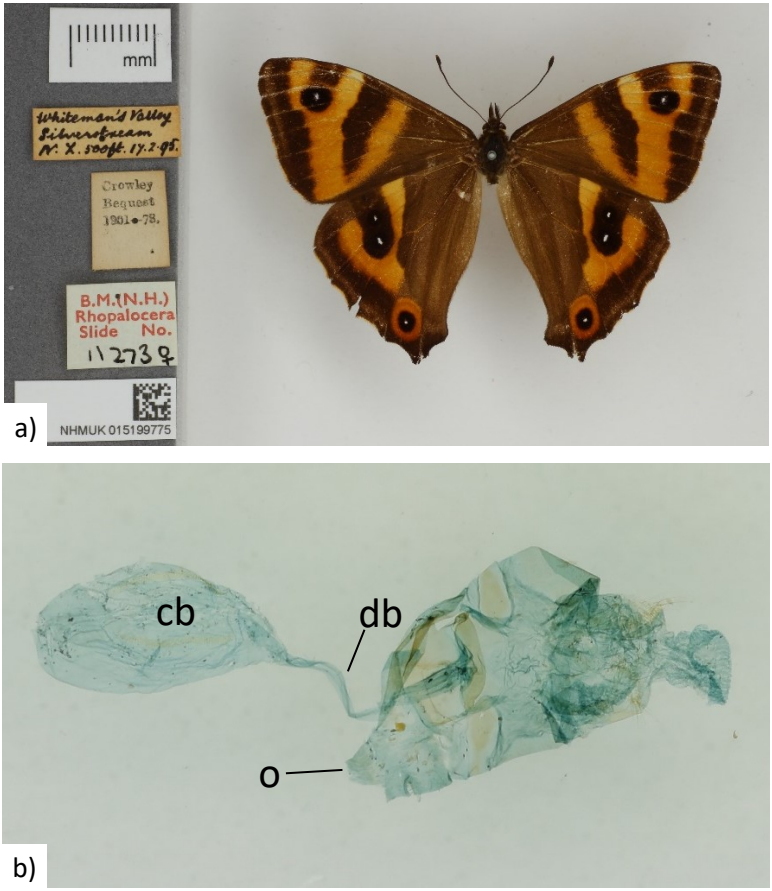

Figure 11: a) Dorsal view of *Dodonidia helmsii* (female) (NHMUK 015199775) with abdomen removed to make a thin-section slide (ref: BMNH 11273) for genitalia examination and gender confirmation; **b)** lateral view of *Dodonidia helmsii* female genitalia; Slide No. BMNH 11273 (stained with chlorazol black) from abdomen of the specimen illustrated in a). Images ©Trustees Natural History Museum, London [Creative Commons License 4.0 (https://creativecommons.org/licenses/by/4.0/]. Labels (added by present authors): cb = corpus bursae, db= ductus bursae, o= ovipositor.

Conclusions

Abdomen dimensions offer the most reliable way of determining male and female *D. helmsii*, with the male abdomen being long and thin with mean length:width (L/W) ratio of 8.10 +/- 1.39, whilst the female abdomen is short and plump with mean L/W ratio of 3.54 +/- 0.59. There appears to be no overlap.

Wing markings are less distinctive, but as first suggested by Barker & Smerdon (2024) forewing markings offer a degree of reliability with males almost always (88%) showing type FW1 with an innermost lens of orange separated from other areas of orange, and females mostly showing FW3 (67%) with all orange areas of forewing connected. No males have been found with FW3 type, but a few females were found with FW1 type, so forewing type alone should not be entirely relied upon. When looking at combined forewing-hindwing markings, however, it was only males that showed the 1-1 combination (Fig.9a), and only females that showed the 3-3 combination (Fig.9i). For these individuals, gender determination could be made with confidence on markings alone, but abdomen dimensions are advocated as the most reliable criteria for *D. helmsii* gender determination.

Being able to confidently identify the gender of *D. helmsii* based on abdomen L:W ratio combined with the pattern of wing marking means that good dorsal view photographs can now be used to record whether the observed individual is male or female. This can be important for understanding gender-related behavioural differences and advancing ecological knowledge of the species.

Acknowledgements

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Thanks, is also given to those individuals who have uploaded *D. helmsii* sightings and images to iNaturalist, or who have sent photos and observations directly to the authors. These and the authors' own photographs have formed the basis of the photographic images used for this research.

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