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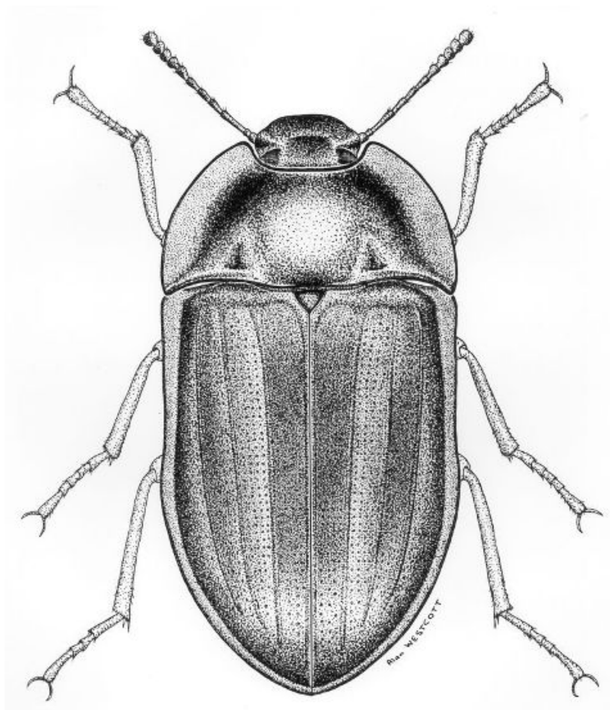

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UTILISING SWARM TRAPS TO EVALUATE AND CONTROL FERAL EUROPEAN HONEYBEE (*APIS MELLIFERA* L.) POPULATIONS

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Summary

In June 2022, Varroa mite, *Varroa destructor* was detected in New South Wales in sentinel beehives located in the Port of Newcastle area. The National Management Group agreed to attempt eradication of Varroa mite from New South Wales. Feral European honeybees were recognised as a threat to the success of the eradication campaign. Based on published international research, we deployed 281 swarm traps to assess Varroa mite infestation and to attempt control of swarming feral bees with the intention to contain the natural spread of Varroa mite. Only two traps caught swarms and no Varroa mite was detected. We discuss the possible reasons for our unexpectedly low capture rates and recommend the use of larger swarm traps with earlier deployment strategies for future trapping programs.

Keywords eradication, varroa, exotic incursion.

INTRODUCTION

In 2019/2020, Plant Health Australia (2021) valued Australian exports of agricultural commodities at more than Aus$25 billion. Pivotally, European honeybees (EHB) (*Apis mellifera* L.) and all pollinating insects contribute to the pollination many of these crops, which were valued at Aus$14.2 billion in 2017 (Karasinski 2018; Frost 2022; APAL 2022). Within the honeybee industry, honey and beeswax production was valued at Aus$162 million (Plant Health Australia 2021). In 2018, the Australian honeybee industry contained approximately 25,000 registered beekeepers managing about 672,200 hives (Plant Health Australia 2021). The industry continues to expand. By 2023, there were about 49,000 beekeepers managing 866,000 hives with a farmgate value of Aus$363.6 million (Clarke and Le Feuvre 2024). In summary, the honeybee industry is a significant contributor to the Australian economy.

The Australian honeybee industry faces many challenges, including exotic pest and disease incursions (Horwood *et al*. 1993; Gillespie *et al*. 2003; Hill *et al*. 2020). Until recently, Australia was the only major honey producing country in the world free from Varroa mite. Saunders (2022) predicted that the establishment of Varroa mite in Australia would cost more than Aus$70 million each year. Unfortunately, *Varroa destructor* (Anderson and Trueman) was detected in NSW near the Port of Newcastle on 22 June 2022 (Bourke *et al.* 2024). Varroa mites feed on larvae, pupae and adult honeybees causing malformation, weakening adult bees, and acting as a vector for many viruses affecting honeybees. During the Australian Varroa response, CCEPP (see Anderson *et al*. (2017) for details) supported several strategies with the intent of eradicating the NSW Varroa mite incursion. One such tool was the use of EHB swarm traps.

Swarm traps can be an effective tool for controlling feral EHB populations (Schmidt and Thoenes 1987, 1990a; Villa 1993; Oldroyd 1998). Population control by trapping and eradication of reproductive progeny is more easily achieved for species that have low reproductive potential. Therefore, feral EHB are ideal candidates for population control by trapping as colonies only produce one to three viable reproductive swarms per year (Schmidt and Thoenes 1990a).

Therefore, the use of artificial swarm traps can be an effective and cost-efficient method to capture and control feral honeybee swarms. This technique was used successfully by the United States Department of Agriculture to manage the arrival of Africanized bees in the 1980s and 1990s (Schmidt and Thoenes 1987; Villa 1993). In the USA and Mexico, over 40,000 swarm traps were utilised to monitor and stem the spread of Africanised honeybees at the peak of the honeybee control activities (Villa 1993).

Nest cavity selection by feral honeybees is a well-organised process involving scouting, evaluation of different cavities, and coordinated movement of the swarm (Seeley *et al.* 1979; Seeley and Visscher 2003). The physical characteristics of nests, such as cavity volume and entrance size, are critical components in the selection of cavities by scouts (Seeley and Morse 1976; Morse *et al.* 1993; Cunningham *et al.* 2022). The colour of the trap exterior is not important (Seeley *et al.* 1989), except that darker boxes can overheat in hotter conditions. Trap placement can influence occupation success (Seeley *et al.* 1989); a higher position above the ground around 5 m is preferred, and the trap should be highly visible to maximise the likelihood of discovery by scouts. A partly or fully shaded area is preferable in warmer climates to avoid overheating (Seeley and Morse 1976; Seeley *et al.* 1989).

Studies investigating the distance travelled by swarms from founding colonies have varying results but suggest that, if resources are present, a swarm will not travel longer distances than necessary. Consensus waggle dances prior to swarm dispersal indicate that most swarms move 500 to 1,500 m to natural, unbaited cavities (Seeley and Morse 1976). Jaycox and Parise (1981) found that artificial swarms preferentially occupied empty hives at distances of 200 m or less. Schmidt and Thoenes (1990a) demonstrated that the highest occupation rates by swarms (albeit for *A. mellifera* Africanised) were between 250-500 m, and that while occupancy did occur for shorter (100 m) and longer (1000 m) distances, they were at much lower rates. Also, Wenner *et al.* (2009) found that honeybee swarms normally move a geometric mean distance of 800 m from their parent colonies.

Previous studies found the addition of synthetic Nasonov gland components to swarm boxes significantly enhanced swarm trap occupation in various ecosystems (Free *et al.* 1981; Kigatiira *et al*. 1986; Schmidt and Thoenes 1990b). Additionally, the use of pheromones as an attractant would likely increase the distance a swarm is willing to travel. For example, Schmidt and Thoenes (1990b) used synthetic Nasonov and estimated that they captured 90% of swarms from a 38-colony apiary using 48 swarm traps at distances between 50 and 1000 m. Also, Schimdt (2001) demonstrated a significant hierarchical preference for swarm traps using Nasonov pheromone lures.Therefore, we considered that swarm traps could become an efficient and powerful tool to control feral EHB and aid in containing the natural spread of Varroa mite. The effectiveness was likely to improve when used in combination with bait stations. Here, we describe swarm traps used to trap swarms and hence to minimise the spread of Varroa mite by monitoring and eradicating reproductive swarms of feral EHB in Varroa Mite Eradication Emergency Zones (EEZs) in the Central Coast, Newcastle, Nana Glen, Gumble and Kempsey regions of NSW.

MATERIALS AND METHODS

We deployed 281 swarm traps starting on 16 August 2023, beginning with 7 traps within the Gumble EEZ. Swarm trap deployment continued along the designated feral bee containment line from late August to November 2023 as resources allowed. Inspections continued until 25 September 2023 following the NMG decision to transition from eradication of Varroa mite to management of the pest species.

Figure 1. Swarm trap (5-frame box) secure. to tree branches using ratchet strap.

Figure 2. Larger 40 L swarm trap with swarm prior to occupation.

Swarm boxes

Our swarm traps were approximately 21-41 L in volume and made of wax-dipped pine or marine ply (Figures 1 and 2). Most traps were based on a 5-frame Langstroth nucleus box, internal dimensions 46.5 (L) x 18.4 (W) x 24.2 (H), 20.7 L in volume, but with two circular entrances (2.5 cm in diameter) on both ends of the box (Figure 1). The other main box design was a larger, swarm trap specific design, with internal dimensions 46.0 (L) x 18.0 (W) x 49.0 (H), 40.6 L in volume (Figure 2). Each swarm trap was branded with DPI brand "A27" and National Varroa Mite Emergency Response labels applied to the outside for identification. Lid brackets were attached to each box with 15-20 mm screws and/or duct tape used to secure lid and weatherproof the gap between lid and box. Traps were elevated from the ground; higher placement was preferable with a safety step used to reach higher branches. We avoided the use of ladders on uneven ground for safety and operational reasons. Swarm traps were set up on metal posts (known as "star pickets") or attached to trees using screws, straps or hooks. Each swarm trap contained at least three frames (new or used, irradiated) with new wax foundation. We applied an attractant (Nasonov /Swarm Commander) to the underside of the trap lid, and also to the entrance or landing platform of the box. A small amount of melted irradiated hive wax was applied to the bottom of the box. Sites were within observed EHB activity and in part or full shade. Where possible, traps were installed at distances of 50 to 1000 m from likely feral EHB colonies.

Surveillance, sampling and euthanasia

Swarm traps were visited by field staff regularly (every 2-7 days) and were reported as "occupied" in MAX when bees were observed flying in and out. When boxes were deemed as occupied, an inspection was scheduled by a swarm team. Swarm teams used PPE (bee suit and gloves) to protect against bee stings and used a bee smoker to calm the bees.

If, upon inspection, the swarm trap was occupied by a colony of honeybees, a large tub (50-100 L heavy duty plastic container with lid) was prepared and soapy water added (up to 10 cm deep). Soapy water was made by mixing water with non-foaming dishwashing liquid detergent in accordance with APVMA permit PER90262. A 35 L bucket was prepared for sieving bees and mites by attaching a paint straining cloth (to collect Varroa mite) and securing a plastic sieve (to collect honeybees) over the straining cloth.

Swarm traps were removed from their housing (trees, metal posts) and placed on the ground for ease of management. The lid was removed and the trap and frames inspected. Where possible, the queen was captured and stored in a queen clip to better control the colony. Then, each frame was removed and the bees shaken from the frames into the soapy water. The lid was added and the tub was vigorously shaken until all bees were euthanised. After euthanasia, the bees and any Varroa mites present were simultaneously sieved through a large plastic sieve to remove bees, and paint straining cloth to remove Varroa mite. Subsequently, any bees caught in the strainer were placed back into soapy water and washed again. This process was repeated three times (three washes in total) to ensure any mites were dislodged. The abundance of euthanised bees was measured using a volumetric flask/jug. Euthanised bees were double bagged, sealed and frozen for at least 48 hours prior to disposal. Finally, the swarm trap was cleaned with ethanol, any removed frames replaced with new frames, and the swarm trap reset.

Spring Strategy Containment Line

Swarm traps were initially paired with existing bait stations deployed within a 'containment line', established at 2 km from the Varroa mite EEZ perimeter and extending for approximately 3km toward the centre of the EEZ (Figure 3). The proposed swarm trap site locations were set on a grid, with each site set 2 km apart. Swarm traps were not deployed along the coastal strips of EEZs.

A swarm trap pilot study was initiated on the 16 August 2023 in the Gumble EEZ to evaluate bee attraction to the traps, with early signs of scout bee recruitment to deployed swarm traps. Subsequently, deployment began within the main Varroa mite containment line extending from Topi Topi, west of Singleton, extending south to St Albans and Berowra (Figure 4), along with the Nana Glen, Gumble and Kempsey regions of NSW. Prior to deployment, permission was gained from landholders to install the traps. GPS coordinates were obtained for each swarm trap site. All swarm trap details were entered into MAX, an electronic biosecurity case management platform.

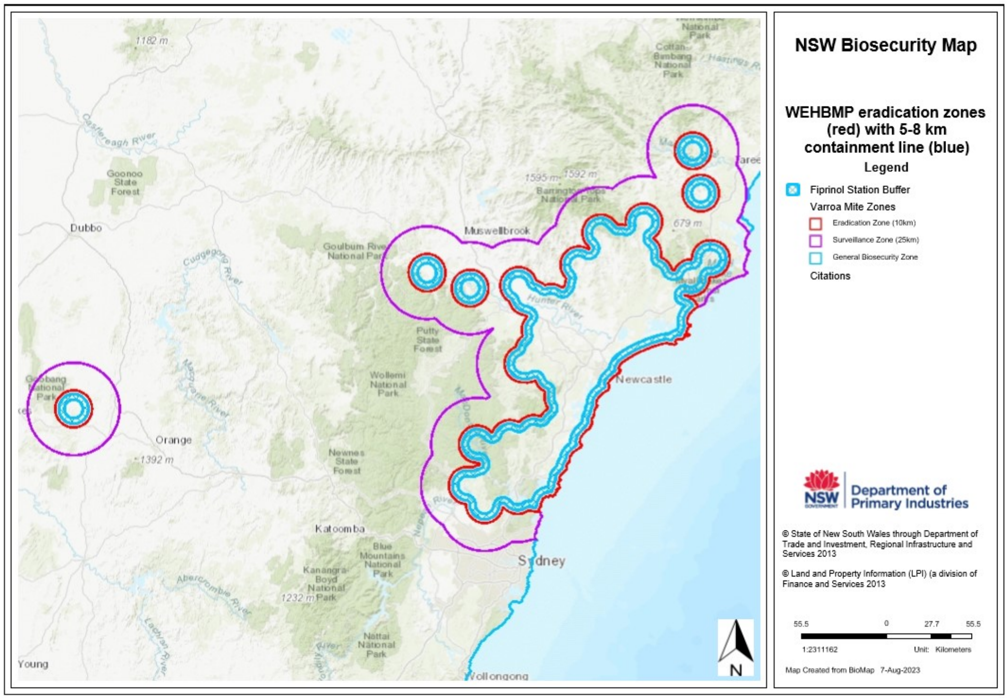

Figure 3. WEHBMP Emergency Eradication Zones (EEZs) in red with 5-8 km containment line (blue).

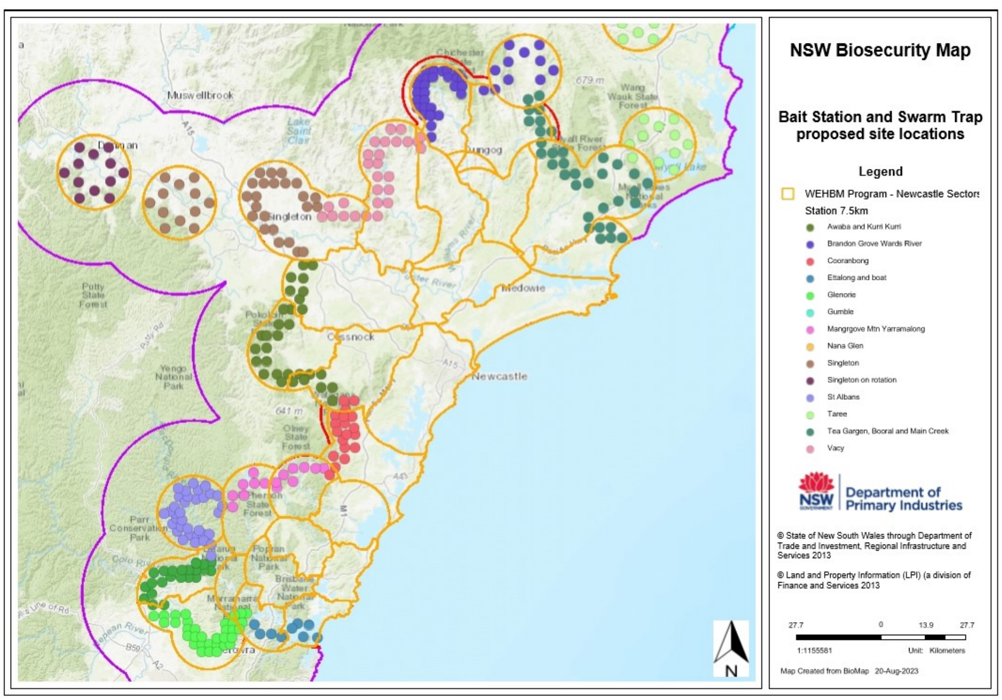

Figure 4. Proposed swarm trap (paired with bait stations) locations (grouped by locality) within the containment line in each Emergency Eradication Zone.

RESULTS

During the inspection period, swarms were detected in two traps (trap ID GUM-03-ST1 and GUM-04-ST1), both of which were in the Gumble EEZ. Both occupied traps were based on the larger volume box design (Figure 2).

Swarm trap GUM-03-ST1 was deployed on 16 August 2023, with subsequent bee activity observed at the entrance beginning on the 18 September 2023. Further observation on the 20 September 2023 confirmed bee occupation and the swarm was subsequently euthanised with the soapy water method described above on the 22 September 2023. The queen was identified and euthanised with the swarm which measured 1.5 L (wet bees) in volume. No Varroa mite was detected in the swarm.

As with GUM-03 ST1, swarm trap GUM-04-ST1 was deployed on 16 August 2023. Bee occupation was first observed on the 22 September 2023 and was subsequently euthanised on the same day with soapy water. The swarm measured 2.0 L (wet bees) in volume. No Varroa mite was detected in the swarm.

DISCUSSION

Contrary to expectations, we captured only two swarms from 281 swarm traps. Based on our results, we conclude that swarm boxes were not an efficient tool (during August-September) to capture swarms although there was no way of assessing how many feral swarms took flight during the assessment period. Encouragingly, neither of the swarms contained varroa. There could be several reasons for our results. Swarm boxes may be more successful later in the season as our boxes were deployed early in the season. Weather conditions may not have been conducive to feral EHB swarming in August-September. We have not way of knowing the number of alternative sites available in the surrounding environment that where attractive to feral swarms. Our swarm boxes had to be designed and placed to suit our labour force for safety reasons. Swarm traps of our design may not be attractive for feral honeybees in the Australia setting. Previous studies showed that nest cavity volume is a key component in the selection of cavities by scouts (Seeley and Morse 1976; Morse *et al*. 1993; Cunningham *et al*. 2022). Our two main trap designs measured 21 L and 41 L in volume which might be considered small when compared with natural nest sizes. Seeley (1977) found a wide distribution of natural nest volumes (12 to 443 L), but most nest volumes were clustered in the 20 to 100 L subrange. When presented with trap sizes of 10 L, 40 L or 100 L, swarms in Ithaca preferred 40 L boxes.

Notably in our circumstances, the two traps that captured swarms in Gumble were of the larger 41 L design, and we suspect that the swarming preference was for larger nesting cavities. Future trapping efforts may consider a larger trap volume to increase occupation rates by feral swarms. However, safety of field crews must be considered as the increased size and weight of the trap becomes unwieldy. This is particularly true when ladders are utilised for elevated trap placement, particularly on uneven ground.

The timing of our trap deployment may have been too late. Due to resource constraints, the swarm trap pilot trial in Gumble didn't start until mid-August 2023, with the main feral bee containment line deployed in the following months. Notably, the only traps that caught swarms were in the Gumble region. Gumble is situated in Central West NSW and lies in high altitude where spring conditions arrive later in the year. While there are some studies that suggest colonies may send out scouts to search for new nest sites several days before the entire swarm issues (Seeley *et al*.1989), there were recommendations from industry that this is too late for Australian conditions. Sydney Bee Rescue reports regular occupation in their swarm traps in spring and early summer when the traps are deployed in winter (June) in NSW, Australia (A. Wilson, personal communication, Jan 22, 2023). Additionally, they report that boxes that face south and are placed 2 – 4 m (or higher if safe to do so) above ground level have increased rates of occupation. In a long-term Australian study, Cunningham *et al*. (2022) reported nest box occupation by swarms after nest boxes were in place for at least eight months before the first surveys which were conducted in July 2010. In Cunningham study, the highest level of nest box use was recorded in spring 2011, and the following spring in 2012, possibly indicating that boxes that were in-situ for some time have higher rates of occupation success. Our data possibly supports earlier deployment of traps to increase occupation success.

During the Varroa Mite Emergency Response, there were multiple time-critical programs operating concurrently, including the euthanasia of both managed and feral honeybee hives. These programs aimed to destroy managed honeybee hives to firstly contain the spread of Varroa mite to EEZs and ultimately to eradicate it from the EEZ. The progression of these programs meant greatly lower numbers of honeybee colonies, both in managed apiaries and feral honeybee colonies within EEZs. Therefore, fewer honeybee colonies would result in fewer swarms to occupy traps. Furthermore, feral colonies that were destroyed in the eradication effort would leave an open nest site, such as a tree hollow, providing increased availability of natural nesting sites for swarming feral honeybees. While there was evidence of feral bee populations remaining in the wild, the success of the managed hive destruction and feral honeybee eradication programs may explain the low occupation rates in our swarm traps.

Despite our literature review of international studies, our swarm traps were not as successful as envisaged. Recommendations for future use of swarm traps would be to increase the box volume and to ensure early deployment of traps (in winter). Additional research is required to better inform the use of swarm boxes in our environment.

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Conflict of Interest

The authors declare no competing interests and there is no conflict of interest

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MOTH TRAPPING AT GOONELLABAH 2480, NSW 2023-2024

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Summary
Moth trapping in a Goonellabah garden near Lismore, NSW. Moth populations and diversity are discussed together with the impacts of climate change. Potential new species are noted.
Key words: Lepidoptera, moths, trapping, climate change

INTRODUCTION

We moved to Goonellabah in February 2023 after emigrating from the U.K. in November 2022. In the U.K. I had undertaken moth trapping on a regular basis in west Cornwall for 9 years from 2003 but with moth populations declining alarmingly during this time across the country I decided to stop trapping at the end of 2011. I employed two traps during this time: a large 'Heath Trap' with a 40W Actinic light in the back garden and a trap almost identical to that depicted below with a 22W Actinic black light which I placed on the roof of my house in Sancreed village.

The garden trap would always attract more moths but the roof trap would attract other species including migrants - which may have been attracted from further afield due to its elevation. By 2011, however, insect populations had crashed in the U.K and some nights I would not catch anything. Sadly, the situation is even worse now and for example, the organisation *Butterfly Conservation* declared 2024 to be "the worst year on record" for butterflies.

In 2023 I began trapping in my back garden in Goonellabah at the end of March and ran the trap every week until the end of the year. I missed a week (Week 46) but otherwise ran the trap once a week for 39 weeks.

During 2024, I undertook moth trapping once a week for the full 52 weeks of the year, commencing January 5th (Week 1) and finishing December 29th (Week 52). It has to be said, after witnessing the decline in insect (and bird) populations generally in the U.K., it was refreshing to move to a country where, at the moment at least, there are still healthy numbers of insects (and birds!) and although I won't be continuing my weekly efforts next year due to time constraints, it has been a welcome surprise to find there are still places in the world with large numbers of Lepidoptera…and exciting discoveries to be made.

METHODS

Unable to source a supplier in Australia for moth trapping equipment, I ordered it direct from the U.K.: a portable "Safari XL" cloth trap with a 22W circular actinic black light bulb. Ideally a bulb with light emissions in the 365nm UV-A spectrum works best (not the 'near UV' bulbs 395-405nm more usually advertised). I would deploy this once a week – preferably 7 days apart where practical – in front of white curtains to the entrance of my garage (Fig. 1). The trap would be left out overnight for approximately 12 hours, commencing an hour before dusk and switching off just after dawn. In the U.K. it is important to catch the first few hours at dusk for moths and I was puzzled initially why this strategy did not work here. I soon realised it must be connected to temperature: in the U.K. temperatures fall rapidly shortly after dusk so most activity is restricted to those early hours when the insects are most active. This clearly is not an issue here with moths apparently arriving all night.

After counting all the moths around the trap early the next morning I would then dismantle the trap and go through the moths contained within, photographing as many as practical before uploading the photos on to the iNaturalist website for identification. Initially I included many photos of micro moths but since I received no feedback – or even ID suggestions from iNaturalist with these – I gave up and only occasionally photographed those which stood out in some way. Hence the discrepancy in my overall total catch (which included micros) and numbers identified.

As in the U.K. I tried to avoid setting up the trap over a Full Moon. Damp, overcast skies even with light rain always yield higher numbers than clear, starry nights.

Fig. 1 Moth trap early morning, 9th September 2024 (week 37) when 822 individuals were counted inside and around the trap. This was the highest count of the year.

Results

Table 1. summarises the data for the two years. Note that a comparison between the two years is skewed in favour for 2024 due to the extra trapping weeks (52 instead of 39) included. For a ***direct*** comparison, by deleting those recorded during the period Jan – Mar (Weeks 1-13) 2024, when 4,699 moths were recorded this gives a total of 7,956 individuals for the remaining period in 2024 - which is 3,721 more than for the same period in 2023 – an increase of 88%. Clearly 2024 was a very good year for moths in Goonellabah at least and even though my garden is in suburbia it is surrounded by trees and a wide variety of gardens with bushes and shrubs which surely must help these insect populations. In addition to the 770 species attracted to the moth trap, two other day-flying moths, *Cephonodes kingii*, Gardenia Bee-Hawk and *Eudocema phalonia*, Common Fruit-piercing Moth have also been recorded.

Table 1. Moths trapped 2023 & 2024 at 5 Homestead Avenue, Goonellabah, NSW 2480.

	2023 (39 weeks)	**2024 (52 weeks)**	**Total**
No. individuals trapped	4,235	12,655	16,890
No. individuals identified	2,170	8,446	10,616
No. species identified	379	684	770

Table 2. Moth families recorded during 2024 ranked in order of abundance. Number of species per family in parenthesis

Rank	Family	Number of individuals
1	EREBIDAE (115)	1604
2	NOCTUIDAE (57)	1459
3	GEOMETRIDAE (104)	1262
4	CRAMBIDAE (68)	876
5	PYRALIDAE (49)	874
6	TORTRICIDAE (36)	658
7	OECOPHORIDAE (68)	445
8	NOLIDAE (33)	264
9	SPHINGIDAE (17)	165
10	STATHMOPODIDAE (3)	113
11	XLORYCTIDAE (13)	88
12	DEPRESSARIIDAE (15)	80
13	PLUTELLIDAE (3)	74
14	GELECHIIDAE (11)	60
14	TINEIDAE (13)	60
15	ETHMIIDAE (3)	59
16	NOTODONTIDAE (6)	50
17	LIMACODIDAE (5)	25
18	COSMOPTERIGIDAE (11)	24
18	PSYCHIDAE (8)	24
19	LYMANTRIIDAE (1)	19
20	BLASTOBASIDAE (2)	13
21	GRACILLARIIDAE (5)	11
21	LECITHOCERIDAE (4)	11
21	PTEROPHORIDAE (4)	10
23	ALUCITIDAE (2)	9
24	ANTHELIDAE (7)	7
24	EUTELIIDAE (3)	7
24	LASIOCAMPIDAE (3)	7
25	CARPOSINIDAE (2)	5
25	COSSIDAE (1)	5
26	CHOREUTIDAE (3)	4
26	OENOSANDRIDAE (1)	4
26	YPONOMEUTIDAE (2)	4
27	LACTURIDAE (2)	2
27	SATURNIDAE (1)	2
27	THYRIDIDAE (1)	2
28	ATTEVIDAE (1)	1
28	BEDELIIDAE (1)	1
28	GLYPHIPTERIGIDAE (1)	1

The most abundant moth trapped in 2024 with 745 individuals was the Noctuid *Spodoptera mauritia,* Lawn Armyworm Moth, followed by the Pyralid *Endotricha mesenterialis*, Snout Moth (517) and the Crambid *Herpetogramma licarsisalis*, Grass Webworm came in third place (212).

Of the 82 moth families in Australia, 40 (49%) were recorded in my garden during 2024. Table 2 ranks these in order of abundance with the Erebidae represented by 1604 individuals (115 species) in the lead.

Unexpectedly, three species were completely new for Australia (Fig. 2) and several more were the first ever recorded in the State of New South Wales. Table 3 below summarises those where there are <20 authenticated records for Australia on the iNaturalist database. However, except for those marked with an asterisk, many species are still awaiting confirmation. The three potentially new species for Australia may even be new for science and have only been confirmed (by others via iNaturalist) to generic level to date. These three are shown in the photos below (Fig. 2). [One of these, a Noctuid *Lophocalama* sp? may be *L. suffusa* although even the online expert admitted it does not really fit that species. If it is that species it would still only be the second record for Australia.]

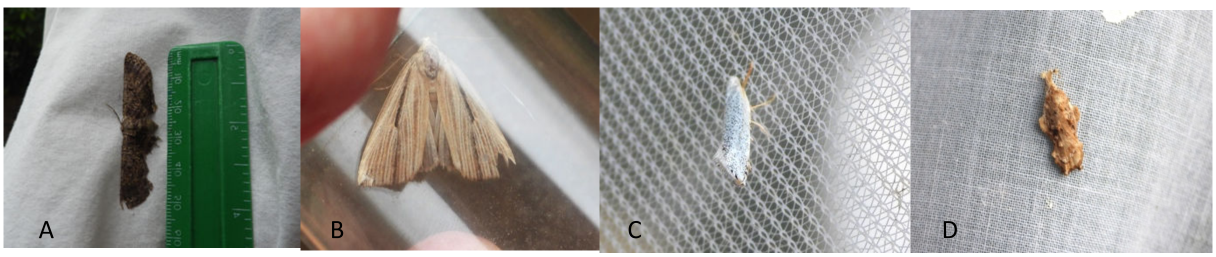
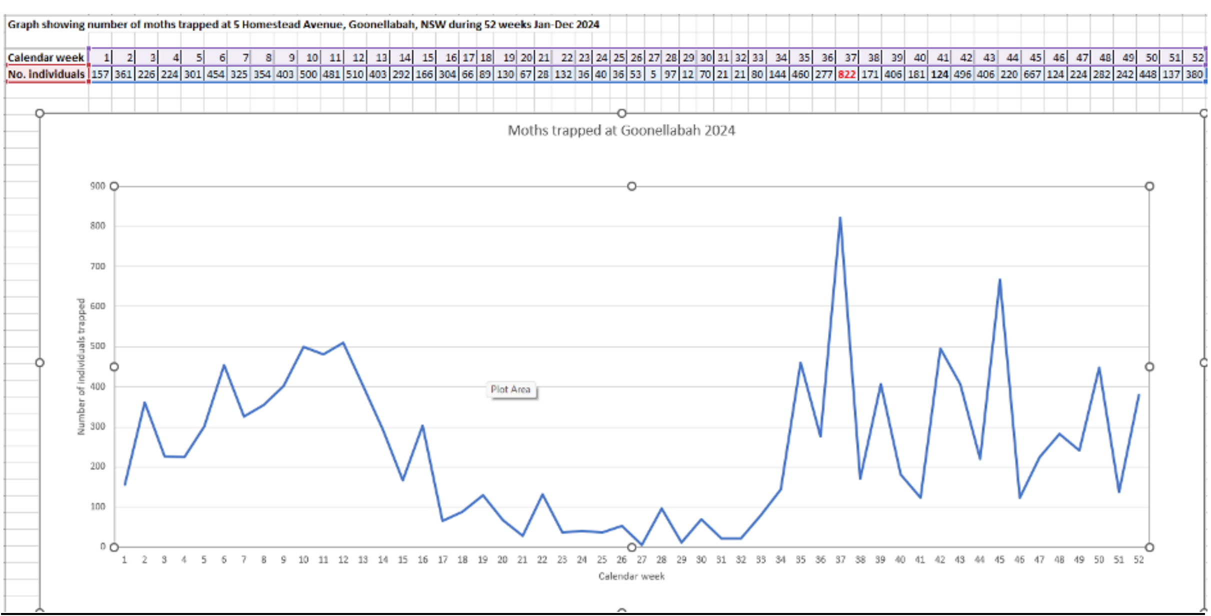


Fig.2 Potential new species. (A) *Ateloptila* sp? (Geometridae) Possibly an undescribed species. 30th November 2024. (B) *Lophogramma* sp? Possibly *L. suffusa* (Noctuidae) or an undescribed species. 30th November 2024. (C) *Lissochroa* sp? (Yponomeutidae). The 4th of its genus in mainland Australia & 1st for NSW. 2nd November 2024. (D) *Tonica flummoxed* (Depressariidae). Possibly an undescribed species.
13th February 2024.

Fig. 3 Number of moths trapped at Goonellabah each week Jan – Dec 2024



Graph showing number of moths trapped at 5 Homestead Avenue, Goonellabah, NSW during 52 weeks Jan-Dec 2024

Calendar week	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26
No. individuals	157	361	226	224	301	454	325	354	403	500	481	510	403	292	166	304	66	89	130	67	28	132	36	40	36	53

Calendar week	27	28	29	30	31	32	33	34	35	36	37	38	39	40	41	42	43	44	45	46	47	48	49	50	51	52
No. individuals	5	97	12	70	21	21	80	144	460	277	822	171	406	181	124	496	406	220	667	124	224	282	242	448	137	380

Note the peak count of 822 in Week 37, (coincidentally the same week in 2023 when numbers peaked too, albeit with fewer individuals, 385 moths).

Table 3. Exceptional records (<20 previous records) 2024.

	Species	Rank
1	*Acraephnes litodes*	15th for Australia
2	*Acropolitis canana*	8th for NSW
3	*Ateloptila* sp? (unnamed)*	1st for Australia
4	*Bastilla monogona**	1st for NSW
5	*Clytoscopa serena**	7th for Australia
6	*Compsotorna oligarchica**	1st for NSW
7	*Cyclopora nodyna**	1st for NSW
8	*Eublemma (Autobar) silicula**	2nd for NSW
9	*Eupselia callidyas*	1st for NSW
10	*Hemiscopis violacea**	6th for NSW
11	*Herpetogramma stultalis**	2nd for NSW
12	*Hippotion rosetta**	2nd for NSW
13	*Labdia leucombra**	3rd for NSW
14	*Lasiosticha canilinea*	19th for Australia
15	*Lophocalama suffusa*?/unnamed*	1st for Australia
16	*Macroglossum prometheus**	1st for NSW
17	*Metura phyllosacca**	18th for Australia
18	*Morophaga clonodes**	14th for Australia
19	*Opogona cleonyma**	5th for NSW
20	*Opogona* sp?*	Undescribed sp.
21	*Ortholomia moluccana*	2nd for NSW
22	*Oxyodes scrobiculata*	1st for NSW
23	*Pantydia metaspila*	1st for NSW
24	*Pherechoa crypsichlora**	17th for Australia
25	*Placocosma hephaestea*	9th for Australia
26	*Thallasodes pilaria*	2nd for NSW
27	*Lissochroa sp?**	1st for NSW
28	*Tisobarica jucundella**	3rd for Australia
29	*Tonica sp?* (*flummoxed*/unnamed)	1st for Australia
30	*Trachydora pygaea*	10th for Australia
31	*Xylorycta haplochroa**	1st for NSW

* Confirmed by iNaturalist

DISCUSSION

Moth trapping is becoming increasingly popular in the U.K. but even when I was active there 13 years ago most counties already had their own 'County Moth Group'. As a member of the 'Cornwall Moth Group' I would submit my monthly observations to the county recorder on a standardised recording sheet and would subsequently receive a quarterly newsletter summarising the data from dozens of observers throughout the county. This data would then be sent to a national recorder and incorporated with the datasets from other counties to produce a summary of the year for the entire U.K. This has shown that for many of the U.K.'s moths, there has been a gradual range expansion northwards, from the southern counties into Scotland over recent decades – linked to climate change. The northward range expansion found in the U.K. is not only confined to moths. Of its 69 butterfly species, 'ten have moved north over the last forty years' (ref. the U.K. organisation *Butterfly Conservation*) including two, Gatekeeper and White-letter Hairstreak recorded breeding in Scotland for the first time in 2024.

One has to ask if there is evidence of such a pattern in Australia linked to climate change? I think the answer to that question is unequivocally yes. Most of the species listed in Table 2 are from the tropics so it is not unreasonable to link their presence here with climate change; populations extending their range southwards due to a warming climate…or perhaps I am the only one looking!

Whether the 88% increase in numbers in 2024 compared to the previous year is related to climate change is perhaps less likely. Populations of Lepidoptera fluctuate wildly each year for a variety of reasons but particularly in relation to local weather patterns, farm chemical usage, disease and predation. No statistical analysis can be drawn from this limited dataset – which is just a snapshot in time – and I'm unlikely to be around to contribute annually for the next 30 years so will have to leave this for others to take up the challenge.

On a final note, when I started trapping here, I sought advice from others regarding recording sheets, moth traps, UV light bulbs etc but quickly discovered Australia has no such system in place. It is probably not surprising that with >22,000 moth species compared with the 2,500 or so in the U.K. there is not a single Field Guide covering any of them. I am perhaps unlikely to witness a dramatic upsurge in interest in Lepidoptera here in my lifetime but I would be delighted if these observations from Goonellabah (in just 12 months) might encourage others to take a look at what is going on in their own backyards on the moth-front one year.

ACKNOWLEDGEMENTS

I wish to thank all those from the global website iNaturalist, particularly Leon Crang for confirming many of my identifications. As already mentioned, there are no Field Guides covering moths in Australia so this online database proved invaluable for suggesting identifications after posting my photographs. Many of my posts still await confirmation but I keep a list of all those that have been confirmed by others to date. In addition, the two private Facebook websites *Australian butterflies & moths* run by Jackie Beer (with 8,100 members) and *Indo-Pacific Moths* (179 members) set up by Baz Hammer have also helped with specific enquiries. My thanks to Dominic Funnell for further help in connection with the latter. Locally I want to thank Hans Wohlmuth for comments on an earlier draft of this paper and fellow mother, Rose Wisemantel for her support and help with identifications. Several Australian birders visited to see the "catch" in 2024 – thank you all for your company. Finally, thanks to Dr Robin Gunning, Editor, *General and Applied Entomology* for help with formatting this manuscript for publication.

BOOK REVIEW

Australia Beetles Volume 3
Polyphaga (Part 2)

Editors: Hermes E. Escalona, Adam Slipinski
Publisher: CSIRO Publishing
Format: Hardback, 720pp
R.R. Price: $280

I have the pleasure of reviewing Australian Beetles Volume 3, but not without some trepidation as this seven hundred plus page tome is the third in a series of such an important group, of beetles, prompting me to more seriously evaluate the previous two volumes in the series for comparison and consistency.

The outstanding diversity of the Australian Curculionoidea, with 4200 species, is the primary focus of this volume. The volume also includes one subfamily of the Cerambycidae, the Lamiinae, in the 12 chapters of this book. It seems odd to include but one Cerambycid subfamily, however it certainly makes clear that subsequent Australian Beetle volumes are in preparation.

Of major importance to applied entomologists, specialist coleopterists and serious amateurs, the great scientific value of this work lies in the fact that for the first time complete and up to date morphological keys to all families, subfamilies, tribes and genera are available for the Curculionoidea. The coverage of the taxa in this volume replace, or are augmented by, the available species level accounts of (the incomplete) Zimmerman's 1990's Australian Weevils volumes. Incorporating up to date treatments, significant changes to the higher taxonomy and some generic concepts has been applied to the eight extant weevil families represented in this volume. Each family chapter provides a brief but concise nomenclaural history, an extensive morphological synopsis of both adults and larvae (where available), and a summary of ecological and biosecurity associations of the included taxa, all richly illustrated. Brief generic synopses include articulation of the type species and listing of all Australian species and their distributions. Remarkably, the many thousand of species of the Curculionidae are condensed to just 527 pages including text and illustrations.

This volume is extremely well illustrated with extensive high resolution electron micrographs, black and white illustrations and nearly 900 colour habitus and collection images that richly embelish the coverage of the target beetle genera. The synoptic nature of this work however precludes any sizing metric in order to accommodate the large number of illustrations.

The keys are excellent and (some) were tested out succesfully on a number of test targets by staff. The key characters are richly supported with illustrations making it suitable experts and non-experts alike. However on an operational note, the fine print, closeness of key couplets and length of ellipsis runs made using the keys for some a little challenging, (like me with sub optimal vision) without use of a ruler.

Each chapter is extensively referenced with many of the most significant and relevant taxonomic publications for the relevant Australian taxa. For a volume of such magnitude the indexing of taxa is valuable, necessary and comprehensive, however the use of emphasis attributes (perhaps bold or italics) within the indices to highlight important features like text sections or keys would greatly enhance the useability of the indices.

This particularly challenging volume is a testament to the tenacity & skills of the editors, authors and publishers in dealing with Australia's most speciose assemblage of related insect taxa. Like the Insects of Australia before, I am sure this series will be a landmark publication, standing as the long term reference text of preference for generatons to come.

I feel that this volume is a must have for proffessional collections/collectors, biodiversity and biosecurity officers, keen coleopterists and book collectors alike.

Peter Gillespie
Collections Curator, NSW DPIRD

SCIENTIFIC NOTE

LARVAL HOST PLANT OF *PHORACANTHA MITCHELLI* (HOPE) (CERAMBYCIDAE): A CURIOSITY AMONGST *PHORACANTHA*

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Summary

Phoracantha mitchelli (Hope) is an obscure species not well represented in insect collections. It is known from coastal Queensland and its range appears to mirror that of its larval host plant *Myrsine variabilis* R.Br. Two specimens of *P. mitchelli* were reared from the root balls of two young plants (ca. 2m tall) at Miranda (NSW).

Keywords: *Phoracantha mitchelli*, *Myrsine variabilis*, larval host

INTRODUCTION

The genus *Phoracantha* was revised by Wang (1995) including the synonymy of *Tryphocaria*, creating the largest genus in the tribe Phoracanthini. The larval hosts of *Phoracantha* spp. are largely restricted to *Eucalyptus* (Myrtaceae) (including *Corymbia* and *Angophora*) and *Acacia* (Fabaceae) (Duffy 1963, Froggatt 1907, McKeown 1947, Wang 1995, Webb 1987). Wang (1995) noted that the larval host plants of most species are unknown.

Phoracantha mitchelli (Hope) is known from various coastal localities in Queensland from the NSW-Qld border to Cape York. A single specimen from Camden, near Sydney, is housed in the Australian Museum collection. To my knowledge there are no larval host records for this species. *Myrsine variabilis* R.Br. (Primulaceae) (commonly referred to as muttonwood) is a shrub or small tree of rainforest or rainforest edges ranging from central coastal NSW to coastal north Queensland (Anonymous 2025). However, it does occur in small patches in wet sclerophyll forest around Sydney. This was the case for specimens described in this study.

OBSERVATIONS

On 6 November 2024, the damaged stems and root systems of several *Myrsine variabilis* plants (ca. 2m tall) were excavated from Kareena Reserve in Miranda (NSW) (-34.042740°S, 151.110995°E). The damage was superficially similar to that of *Aphanasium australe* (Boisduval) in *Grevillea* and *Hakea* (Webb 2023). However, on 10 January 2025 two specimens of *P. mitchelli* emerged. Both emerged from the remains of the root balls of two separate plants, one of which originally contained two larvae (the second did not survive). Damage was confined to the root ball and radiating main roots. No damage was evident in the stems above ground level.

One specimen maintained its bright colouration after death as seen for the type specimen of *Stenochorus mitchelli* Hope shown in Slipinski and Escalona (2016) (pg. 575) while the other darkened dramatically (Figure 1).

Wang (1995) described *Phoracantha cruciata* and noted the similarity to *P. mitchelli*. *Phoracantha cruciata* was distinguished by having the third antennal segment distinctly longer than the fourth. This is not the case for these two specimens.

ACHNOWLEDGEMENTS

One journal reviewer provided useful comments on the manuscript. This research was conducted under NSW Department of Planning, Industry and Environment license number SL102455

Figure 1: Specimens of *Phoracantha mitchelli* (both 28mm length – mandible to tip of elytra). NB. Third and fourth antennal segments similar in length.

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EVALUATING FIPRONIL RESIDUES AND RE-ESTABLISHMENT SUCCESS IN MANAGED HIVES FOLLOWING WILD EUROPEAN HONEY BEE (*APIS MELLIFERA*) ERADICATION IN NEW SOUTH WALES

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Summary

Following the incursion of Varroa mite (*Varroa destructor*) in New South Wales, Australia, in June 2022, eradication was attempted to protect the honey bee related industries. A Biosecurity Emergency Order was established to destroy managed and feral European honey bees within Emergency Eradication Zones, using specialised bait stations and fipronil. Our study aimed to identify managed hives within 1.5 km of a bait station and assess if they were impacted by fipronil application, to determine any residual fipronil concentrations within the hive and evaluate re-establishment success. We found eleven apiaries that met the study requirements, three of which reported colony loss during the eradication program. One apiary reported three destroyed hives that may be attributed to fipronil application at a bait station 3.08 km away (fipronil from closest bait station showed no impact). A very low concentration of fipronil was detected within the hive but was below the limit of quantification. Another apiary reported the loss of two hives, coinciding with the application of fipronil at a nearby bait station (387 m); the apiary has since successfully re-established hives. The third apiary experienced colony loss well after eradication efforts had ceased and, therefore, we consider the apiary was unlikely impacted from fipronil application; the colony was successfully re-established. The remaining eight sites were unimpacted and fipronil residues were not detected by HPLC. Further research is warranted to determine if re-introduced hives to eradication zones are impacted by residual fipronil in the environment.

Keywords: Varroa, emergency response, management.

INTRODUCTION

The Australian European honey bee (EHB) industry is a significant contributor to agricultural and horticultural production, and a major employer. Australian exports of agricultural commodities in 2019/2020 were valued at more than A$25 billion (Plant Health Australia 2021). Crucially, European bees (*Apis mellifera* L.) pollinate many of these crops, which were valued at A$14.2 billion in 2017 (Frost 2022; APAL 2022). Additionally, honey and beeswax production were valued at A$162 million (Plant Health Australia 2021). In 2021, the Australian honey bee industry contained approximately 25,000 registered beekeepers managing about 672,200 hives (Plant Health Australia 2021). The industry faces many endemic disorders (Bourke 2020) and must prepare for exotic incursions (Gillespie *et al*. 2003; Carnegie and Pegg 2018).

Varroa mite (*Varroa destructor* (Anderson and Trueman)) is an exotic external parasite of EHB and was first detected at the Port of Newcastle, NSW, on 22 June 2022 (McFarlane *et al.* 2024). An eradication program was initiated to protect the industries reliant on honey bees. The original 100 days of the response was described by Bourke *et al.* (2024), and Taylor *et al.* (2025) described the methods used to assess varroa populations in the response. The response plan had many components including attempting to trap swarms of feral bees, possibly infested with the mite (Nguyen *et al.* 2025), euthanasia of EHB and community awareness (Bourke *et al.* 2024). As part of the response, a *Biosecurity (Varroa Mite) Emergency Order* (*No 21*) *2022* was issued on 12 August 2022 which included specific clauses permitting their destruction by authorised personnel. An eradication program was established to destroy managed and feral honey bees within Emergency Eradication Zones (EEZs) (NSWG 2022). The Wild European Honey Bee Management (WEHBM) Program was initiated to control feral honey bees, operating from September 2022 until the Transition to Management was declared in September 2023 (Bourke *et al.* (2024).

Feral EHB colonies are more challenging to destroy than managed colonies, because their hives can be difficult to locate. Therefore, specialised bait stations (designed as detailed in the APVMA permit as a guideline) were deployed to attract local feral EHB to the bait station. A variety of species-specific food and attractants were used to actively attract and recruit foraging feral EHB to the bait station including: 1) 50-60% sugar and water (sugar syrup) solution contained within the bait station, 2) spraying an irradiated honey and water mixture around the bait station, and 3) heating a mixture of irradiated honey and irradiated, loose beeswax in the immediate area surrounding the bait station.

When enough EHB were observed to be feeding from the bait station (typically >300 bees recruited in the first baiting cycle), the sugar syrup solution was replaced with sugar syrup solution spiked with fipronil (10 mg fipronil/L of sugar syrup), in accordance with the APVMA permit, PER84929 (this permit has expired and is no longer available online) (APVMA 2017). Bait stations were raised off the ground and had grease strips to exclude ants and other non-target insects. Fipronil was applied only after EHB were seen feeding from the sugar syrup; the feeding was monitored throughout the spiking time to mitigate against non-target poisoning. The fipronil and sugar syrup solution was made available to the EHB for 2-3 hours until no more bees were seen foraging from the station. Then, the fipronil was removed and the cycle was repeated until no more bees were attracted to the bait station.

In March 2023, the option was made available to small apiaries (ie. apiaries with less than 10 hives) within EEZs to either euthanise their hives themselves without assistance from New South Wales Department of Primary Industries Surveillance and Destruction staff, or to leave their hives in situ. Hives left in situ were potentially exposed to fipronil bait stations deployed within the EEZ and had the potential to be impacted by fipronil.

The impacts of baiting and attempts to re-establish these hives is largely unknown, particularly those hives that were not destroyed, or disposed of. Also, the published literature on the impacts of fipronil residues and persistence is limited, particularly in the context of EHB (APVMA 2010). Therefore, our study aimed to identify those managed hives that were left in situ (within 1.5 km of a bait station) and determine concentrations of residual fipronil within managed hives following fipronil application.

MATERIALS AND METHODS

We used historical information obtained from the Varroa Mite and Wild Bee Management, MAX Plant Biosecurity records. MAX is a software program that contained data on registered beekeepers with hives in eradication zones (Figs. 1 and 2) that elected not to have their hives destroyed, either by DPI or themselves. We identified and contacted those beekeepers. If any hives were located within 1.5 km of a bait station and exposed to fipronil application, then we obtained permission from the beekeeper to sample the hive for nectar/honey, then a hive health assessment was conducted and the beekeeper was asked to complete a short survey. In addition to the survey, we asked beekeepers if any treatments were used in the hives to control pests (eg. fipronil used to treat small hive beetle (*Aethina tumida*)). We visited eleven apiaries and collected samples from November 2023 to January 2024 (Table 1).

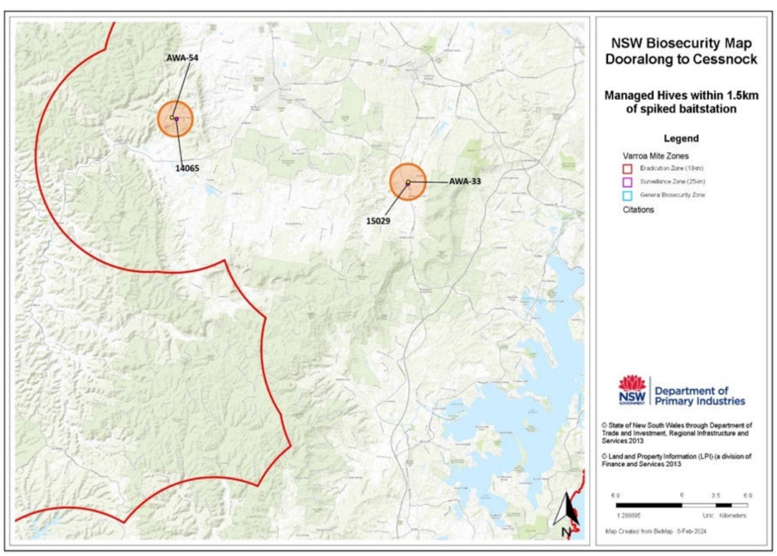

Figure. 1. Managed hives (pink) and bait station (yellow) locations, Northern Sector, Dooralong to Cessnock

Table 1. Study cases (hives) with MAX case IDs, nearest bait station/s within 1.5 km, distance from bait station/s, fipronil application (spike) dates, shake sampling date, beekeeper survey results, fipronil concentrations in shake/nectar samples and general hive health.

Max case	Nearest bait station/s (<1.5 km)	Distance from bait station/s (m)	Fipronil application dates	Bee mortality (Y/N)	Colony mortality (Y/N)	Date Colony Death	Shake Sampling Date	Re-introduced Hardware (Y/N)	Re-introduction Succesful (Y/N)	Pest Treatments (locally or in hive)	Fipronil Concentration (mg/L)	General hive health/notes at time of sampling
4088	MTA-101	1480	7/09/2023	Y (1-2 bees)	Y (colony lost due to AFB)	Sep-23	23/11/2023	Y	Y	N	ND	5 hives sampled; strong hive health (with a mixture of smaller and larger colonies); no major presence of pests.
7683	SOM-02 SOM-23	1030 1340	**SOM-02:** 21/12/2022 07/06/2023	N	N	NA	23/11/2023	NA	NA	Y (SHB treatment in hive)	ND	1 hive sampled; no impacts; good hive health; positive for varroa mite (alcohol wash) with low numbers of SHB. Low number of drone brood.
12063	MTA-104 MTA-78	894 1389	**MTA-104:** 23/08/2023 **MTA-78:** 07/09/2023 12/09/2023	N	N	NA	8/11/2023	NA	NA	Y (neighbour spraying for weeds)	ND	8 hives sampled (active hives only); signs of AFB (perforated brood, oily, smell); SHB observed; some hives with strong health.
12223	MTA-101	1078	7/09/2023	N	N	NA	11/12/2023	NA	NA	N	ND	2 hives samples; hive 1 (alive), Hive 2 (dead). Dead hive had AFB and SHB present. Hive 1 showed strong hive health.
12621	MTA-105	387	6/09/2023	N	Y	Sep-23	22/11/2023	N	NA	N	1.56	2 hives sampled; 2 hives impacted; both hives succesfully re-established with strong hive health; Indoxocarb used to treat SHB. Survey notes "DPI experience today very professional".
12630	MTA-105	826	6/09/2023	N	N	NA	19/12/2023	Y (requeenin g)	Y	N	ND	44 hives - 5 representative samples collected; all good hive health. Some SHB was present but manageable.
12696	MTA-103	1378	21/08/2023	N	N	NA	13/12/2023	NA	NA	N	ND	7 hives sampled; no impacts; Hives were healthy but showed early detection of varroa mite (bayvarol/sticky mats).
12800	MTA-105	487	6/09/2023	Y	Y	Dec-23 1/12/2023	13/11/2023	NA	NA	NA	ND	2 hives sampled; 1 colony impacted; loss occurred 3 months after last spike. 1 hive shows good hive health.
13958	ETT-IPU6 ETT-IPU1 ETT-IPU5	385 894 988	**ETT-IPU6:** 1/06/2023 28/07/2023 **ETT-IPU1:** 14/04/2023 22/05/2023 01/06/2023 **ETT-IPU5:** 03/05/2023 22/05/2023	Y (~1500-2000)	N	Oct-23 7/10/2023	7/11/2023	N	NA	Y (Neighbour accused of spraying poison)	ND	1 hive sampled; strong hive health at time of sampling; bee death attributed to neighbour spraying insecticide; low SHB numbers present
14065	AWA-54	370	18/05/2023	N	N	NA	20/11/2023	NA	NA	N (possible treating/spot spraying St Johns Wort)	ND	1 flow hive sampled; no impacts; moderately healthy but with high SHB numbers in honey super; no SHB traps present.
15029	AWA-33	176	09/06/2023, 1/08/2023	Y	Y	Apr-23	15/11/2023	N	NA	N	~0.25 (< LOQ)	3 flow hives sampled: hives impacted; reports weak and dead bees outside hive entrance. Colony lost before first spike. Re-establishment not attempted.

Abbreviations: SHB, Small Hive Beetle; AFB, American Foulbrood; ND, Not Detected; LOQ, Limit of Quantification; N, no; Y, yes; NA, not applicable.

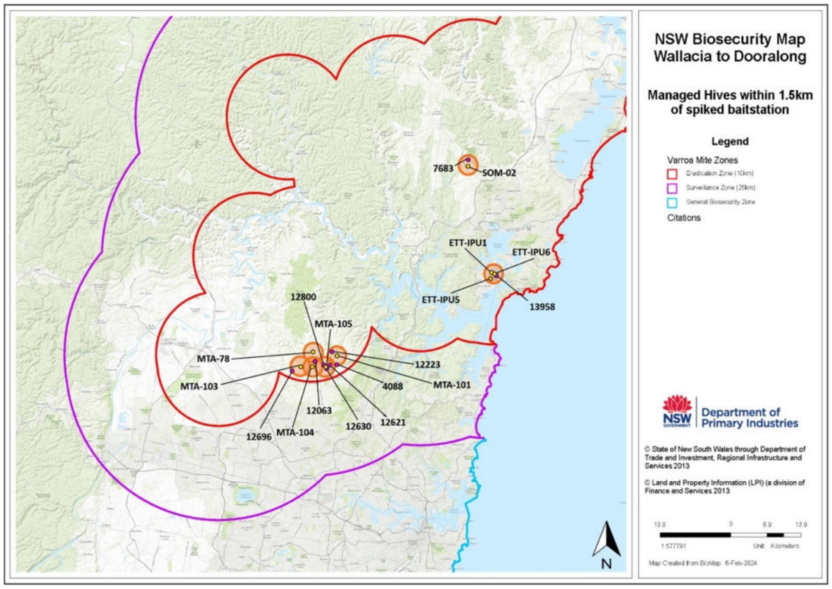

Figure. 2. Managed hives (pink) and bait stations (yellow) locations, Southern Sector, Dooralong to Wallacia.

Figure. 3. Visible nectar/honey within open cells of a frame, deposited by foraging EHB.

Figure. 4. Frame containing nectar/honey were held flat and vigorously shaken until the nectar/honey solution fell from the cells into the collection tray.

To quantify the concentration of fipronil within the hive, we targeted nectar and shake samples. EHB are highly sensitive to fipronil, with a reported LD_{50} of 4.17 ng/L (Johnson, 2015). Previous assessments of bait station efficacy found that EHB succumb to fipronil 31.6 ± 9.5 min after first forage (Nguyen et al., in prep.). Consequently, colonies affected by fipronil would likely collapse shortly after exposure, precluding the conversion of contaminated nectar or syrup into capped honey. Nectar/shake samples, therefore, represent a more reliable matrix for analysis, as they are expected to contain the highest detectable concentrations of fipronil. Therefore, we collected shake and uncapped honey (typically comprising nectar or sugar syrup) and analysed these samples using high-performance liquid chromatography (HPLC) following established methods (Tomasini et al., 2011).

To collect the shake solution, a large clean tray was set up near the hive and covered with baking paper. Frame/s with open cells containing nectar/sugar syrup were held flat and horizontally to the tray and vigorously shaken until the nectar/sugar syrup solution fell from the cells into the tray (Figs 3 and 4). If the solution was not readily shaken from the frames, then we used a syringe to extract the sample. When a representative sample was collected, the solution was transferred to a vial, placed on ice in a dark container, immediately transported to a freezer and to the University of Newcastle laboratories for analyses. All samples were stored at -18 °C until required for analysis.

The nectar/honey samples were analysed for fipronil using a modified QuECHeRS method (Quick, Easy, Cheap, Effective, Rugged, and Safe) for determination by HPLC, as described by Tomasini *et al*. (2011). The QuEChERS method is recognised as an official method in the detection of multiple pesticide residues in fruits, vegetables and honey (Anastassiades *et al.* 2003; Lehotay 2007; Tomasini *et al*. 2011). This method is particularly useful for the determination of polar, middle polar and non-polar pesticide residues in various food matrices because it is robust, inexpensive and suitable for determination of pesticides in complex samples (Tomasini *et al.* 2011). In optimal conditions, the limits of quantification of fipronil using the QuEChERS method is 0.6 mg/kg.

Briefly, the QuECHERS procedure involves initial extraction of honey with acetonitrile, followed by liquid-liquid partitioning formed by the addition of $MgSO_4$. Nectar/honey (2.0 g) samples were weighed and placed into a 50.0 mL PTFE centrifuge tube. Deionized water (4.0 mL) was added and the mixture vortexed until it became homogenous. Then 4 mL acetonitrile was added and the sample homogenized for 1 min. After adding 1.6 g anhydrous magnesium sulphate, the mixture was vortexed again for 1 min, and then centrifuged for 5 min at 5000 rpm. Finally, 20 µL of the upper acetonitrile layer was filtered (0.45µm) into 2mL vials before injection in HPLC system for analysis.

RESULTS AND DISCUSSION

We identified eleven viable cases that met the criteria and where a beekeeper agreed to participate in the study (Table 1). Of the eleven cases, three showed signs of potential impacts from spiking as "colony loss" was experienced during the baiting program. The first case reported colony loss in three flow hives in approximately April 2023. The nearest bait station (bait station ID AWA-33, distance of 176 m) was first spiked on 9 June 2023 and subsequently on 1 August 2023. The initial spike date occurred two months after the observed colony loss date (based on the recollections of the beekeeper) although the report of hundreds of dead bees outside hives was typical of a fipronil impacted hive. We deduced that the observed colony loss was not due to fipronil application at AWA-33. However, we found that a bait station (AWA-34) 3.08 km away was spiked on 15 May 2023, closer to the reported colony loss date. Bait stations may be most effective up to 1 km. Numerous studies have shown bees can travel much longer distances when foraging for food resources (Couvillon *et al*. 2015; Beekman and Ratnieks 2000; Visscher and Seeley 1982). Therefore, the reported colony loss for Case 1 may be due to fipronil application at AWA-34, which may also explain the very low concentration of fipronil detected in the shake sample, as determined by HPLC. Re-establishment of these hives was not attempted by the beekeeper.

Case two reported colony loss in two hives in September 2023, coinciding with the nearest bait station (MTA-105, 387m) fipronil application on 6 September 2023. Since the time of spiking and colony loss, both hives had successfully re-established with healthy colonies and with no signs of fipronil poisoning during re-colonisation (hives re-assessed for hive health on 22 Nov 2023). HPLC results showed 1.56 mg/L of fipronil, however, the beekeeper reported use of traps containing oil and diatomaceous earth, to treat small hive beetle and other pests, along with chemicals including Indoxacarb, an oxadiazine pesticide. Indoxocarb has similar topological polar surface area to fipronil (NCBI 2024), and we suspect that the presence of indoxocarb might interfere with the detection of fipronil as determined by HPLC. EHB are very sensitive to fipronil with a reported LD_{50} of 4.17 ng/L (Johnson 2015), well below the concentration reported by HPLC. Given that the hives were healthy during the time of sampling, therefore, we think that it is unlikely the reported figure was an indication of fipronil, but more likely confounded by the presence of indoxocarb or its metabolites.

Case three reported colony loss on 1 December 2023 with the nearest bait station (MTA-105, 487m) having fipronil applied on the 6 Sep 2023. The reported date of colony loss falls well after the spike date and after eradication efforts had ceased. On September 2023, the National Management Group decided that eradication was no longer feasible (Bourke *et al.* 2024) and the eradication response program transitioned to management of the pest, following the National Transition to Management Plan which was initiated on the 19 Sep 2023. HPLC results determined no detectable concentrations of fipronil from the hive samples. Therefore, we consider it was unlikely that the fipronil application in September at MTA-105 was the cause of colony loss in December. The lost colony was successfully re-established.

We did not detect fipronil at the remaining eight sites and all sites that attempted re-establishment were successful according to the survey results. We think that these results are not surprising given that most hives showed no impacts from fipronil during the eradication effort. Furthermore, most hive samples were taken some time after the nearest bait station/s were spiked, and the hives were healthy or active when samples were taken. If the hive survived, then there was little chance that there was fipronil within the honey, given the high sensitivity of EHB to fipronil. It should be noted that our study was not indicative of all hives that were exposed to fipronil because most of these hives were destroyed during the program. Therefore, we proposed that any fipronil that may have been present was likely either removed or degraded. Also, we think that it was unlikely that fipronil would be detected within the hive samples, considering that the hives were healthy or active at the time of sampling and that EHB are very sensitive to fipronil.

Based on our findings, we suggest that most apiaries that met the criteria for the study showed no impact from fipronil application from WEHBM bait stations. We recognise that our study had some limitations. Our study sites may not be representative of all apiaries that may have potentially been affected by the WEHBM eradication program. For instance, hives that were exposed to and impacted by fipronil during eradication were probably destroyed, owner reimbursement costs claimed, and hives subsequently disposed of. Therefore, these fipronil impacted hives were not available to be included this study, limiting the sample subset to mostly non-impacted hives. Furthermore, commercial beekeepers were asked to opt in and agree to sampling, with many declining to participate in the study. This applied especially for several commercial apiarists with large amounts of hives which may have been impacted. This opt-out option may have substantially decreased our sample size. Conversely, recreational beekeepers, with much smaller quantities of hives, were much more agreeable to participate in the study. Additionally, unregistered beekeepers were not included the study, as the keeping of bees without registration was against current NSW law.

We were heartened by the conduct of the survey and by the response from most beekeepers. We think that the beekeeper survey was effective, not only to collect qualitative information, but also to engage with beekeepers, allowing them to voice their experiences with the program. There were several comments expressing the professionalism and care taken by DPI staff.

CONCLUSION

Our study aimed to investigate the impact of residual fipronil and re-establishment success of managed hives that were exposed to fipronil application from a WEHBM bait station. This aim is distinct from the assessment of residual fipronil in the environment (ie. foraged from wild hives) and, therefore, our research does not entirely address the safety of re-introduced hives. However, we did not find much evidence to support the theory that re-established hives would be adversely impacted by the fipronil used in WEHBM bait stations. Further research is needed to determine if re-introduced hives to eradication zones are impacted by residual fipronil in the environment.

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FABACEAE AS LARVAL HOSTS OF *URACANTHUS BIVITTA* NEWMAN (CERAMBYCIDAE, CERAMBYCINAE, URACANTHINI).

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Summary

Uracanthus bivitta Newman is known from largely coastal areas from central Queensland through to South Australia and Western Australia. Aside from a few historical records, all larval host plants for this species appear to be from the family Fabaceae. In this study a range of Fabaceae species from southern Sydney were identified as larval host plants.
Keywords *Uracanthus bivitta*, Fabaceae, larval hosts

INTRODUCTION

Historical records indicate that *U. bivitta* has been reared from a number of host plants (mostly Fabaceae) (Dixon 1908, Duffy 1963, French 1900, 1911, McKeown 1947). French (1911) did not specifically identify *U. bivitta* as infesting any nominated species but rather linked some of these larval host plants to a group of three *Uracanthus* spp. including *bivitta* and McKeown (1947) followed this notion. However, French (1900) did identify *Ulex* sp. as a larval host of *U. bivitta* and Duffy (1963) referenced an unpublished record for *Pultenaea stipularis* Sm.. Dixon (1908) was not explicit in nominating whether the listed food plants were larval or adult food plants but it is generally accepted that these were larval food plants. These were *Ulex europaeus* L., *Cytisus scoparius* (L.) Link, *Daviesia latifolia* R.Br. and *Viminaria juncea* (Schrad. & J.C.Wendl.) Hoffmanns (all Fabaceae).

Duangrat Thongphak (Thongphak 2007; Thongphak and Wang 2007) revised the genus *Uracanthus* and therefore there is some uncertainty about larval host associations listed in the literature prior to this review. They listed specimen label data for *U. bivitta* which indicated larval host associations with *C. scoparius, Pultanaea* spp., *Jacksonia sternbergiana* Benth. and *Jacksonia* sp. (all Fabaceae). With the exception of a few historical records all larval host plants appear to be from the family Fabaceae.

Infested stem material from various species of Fabaceae were collected from various locations in southern Sydney (New South Wales) during 2021 and maintained in plastic tubs under semi-controlled conditions. Progressively over time, adult specimens of *U. bivitta* emerged. All specimens of *U. bivitta* from this study were checked against specimens labelled by Duangrat Thongphak in the Australian National Insect Collection in Canberra.

RESULTS AND DISCUSSION

***U. bivitta* records from Fabaceae**

(RNP = Royal National Park)

***Bossiaea heterophylla* Vent.**

1. NSW, RNP, Bottle Forest Fire Trail, collected on 30 June 2021, cut live 30 June 2021 (2 specimens).
2. NSW, RNP, Bottle Forest Fire Trail, collected on 7 September 2021, emerged 2 July 2021 (1 specimen).

***Daviesia alata* Sm.**

1. NSW, RNP, Curra Moors Fire Trail, collected on 2 September 2021, emerged on 18 December 2021 (1 specimen).

***Daviesia ulicifolia* Andrews**

1. NSW, Menai, Heathcote Rd., collected on 6 September 2021, emerged on 17 December 2021 (1 specimen).

***Dillwynia retorta* (J.C.Wendl.) Druce**

1. NSW, RNP, Winifred Falls Fire Trail, collected on 2 January 2021, emerged on 7 March 2021 (2 specimens).
2. NSW, Kurnell, Caltex boundary fence, collected on 26 July 2021, emerged on 24 December 2021 (1 specimen).

***Dillwynia sericea* A. Cunn.**

1. NSW, RNP, Grays Point, collected on 4 January 2021, emerged on 16 March 2021 (1 specimen).

***Gompholobium grandiflorum* Sm.**

1. NSW, RNP, Curra Moors Fire Trail, collected on 2 September 2021, emerged on 29 November, 14 and 24 December 2021, 2 and 8 January 2022 (5 specimens).

***Phyllota phylicoides* (DC.) Benth.**

1. NSW, RNP, Curra Moors Fire Trail, collected on 28 January 2021, emerged on 7 November 2021 (1 specimen).
2. NSW, RNP, Loftus, collected on 3 May 2021, emerged on 23 November 2021 (1 specimen).
3. NSW, RNP, Curra Moors Fire Trail, collected on 26 August 2021, cut live on 26 August 2021 (1 specimen).
4. NSW, RNP, Curra Moors Fire Trail, collected on 26 August 2021, emerged on 12, 29 December 2021 (2 specimens).

***Pultenaea tuberculata* Pers.**

1. NSW, RNP, Anice Falls Fire Trail, collected on 28 April 2021, emerged on 10 October 2021 (1 specimen).

With the exception of the dubious records of *Ozothamnus ferrugineus* (Labill.) Sweet (Asteraceae) and *Acacia longifolia* (Andrews) Willd. (more recently considered to be Fabaceae) (French 1911, McKeown 1947), all other larval host records including those provided here were from species of Fabaceae. To my knowledge there are no other published records from other plant families for *U. bivitta,* although other species of *Uracanthus* are known to breed in plant species from a variety of other families (Dixon 1908, French 1900, 1908, Hawkeswood 2002, Thongphak and Wang 2007, Webb 2020 and others).

Uracanthus bivitta typically infests a single stem but may infest the root system of some host species where multiple larvae may reside. Larvae cut the typical V-shape slit in a stem above which the plant material snaps off. They then feed downwards to the base of the stem or root system to pupate and later emerge through the V-shape slit or close to. A pre-emergent adult, larvae and pupae are shown in Figures 1 and 2.

So far there is no evidence of *U. bivitta* utilizing any plant species other than members of the Fabaceae family.

Figure 1: Pre-emergent *U. bivitta* in hollowed out stem of *Phyllota phylicoides*.

Figure 2: Larvae and pupae of *U. bivitta* in hollowed out stem of *Phyllota phylicoides*.

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A REVIEW OF THE UNIQUE NESTING CHARACTERISTICS OF *ROPALIDIA PLEBEIANA* (WHITE-FACED BROWN PAPER WASP) – AN ETHOLOGICAL PERSPECTIVE

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Summary

This study examines the nesting characteristics of *Ropalidia plebeiana* (White-faced Brown Paper Wasp), a species found in southeastern Australia. The species is unusual amongst social vespid wasps in the variety of nesting styles that it exhibits.

This study reviewed scientific and citizen science information, conducted field observations on nest forms, and investigated the possible ecological and behavioural factors that might account for the nest variability across the full geographic range of the species.

In New South Wales (NSW) and southeast Queensland, nesting in large aggregations of nests, or "cities", is common with northern populations forming the largest aggregations. In contrast, solitary nests are more typical toward the south and in the Australian Capital Territory (ACT) where large aggregations are not present. Aggregation sites appear to be influenced by nesting site availability and the availability of food resources for adults and larvae; warmer regions offer more abundant resources, supporting larger groupings.

Nests exhibit either a stick-style or plate-style configuration and may be suspended in a pendent form or attached flush to the substrate. Non-pendent nests predominate in the northern parts of the range; further south, pendent nests are more common. The form and attachment method of each nest are shaped by its unique developmental progression across multiple seasons. Additionally, nest architecture correlates to location, substrate type, exposure to weather and spatial constraints in the immediate environment.

The observed variability in the construction of various nest types is primarily the result of five main innate behaviours; each being expressed to the extent permitted by prevailing conditions. This results in diverse nesting outcomes across time and geographical locations.
Keywords: *Ropalidia, plebeiana*, ethology, nesting.

INTRODUCTION

Ropalidia plebeiana (White-faced Brown Paper Wasp) is one of 36 species of social wasps of the subfamily Polistinae in Australia (Australian Faunal Directory, Harris 2025) including two introduced species. It is the only temperate climate species of *Ropalidia* with a distribution spanning the east coast of Australia between Brisbane and Melbourne - the others are all mainly tropical or sub-tropical. They are a small wasp at about 10-12 mm long (Figure 1). They are generally not aggressive and, individually, have only a mildly painful sting. The species has a large window of seasonal activity, emerging early from winter dormancy and rapidly commencing the rearing of young by re-using old nests.

Much of what we know of the species is based on work carried out in the 1970s by Richards and by Itô and others in the 1980s, who studied nests in the South Coast area of New South Wales (Richards, 1978; Itô, 1987).

Historically, the species was found in aggregated nest sites in rocky outcrops. Following the arrival of man-made structures, the species is more likely to be found nesting under more plentiful human constructs such as bridges, making the species an apparent beneficiary of the Anthropocene.

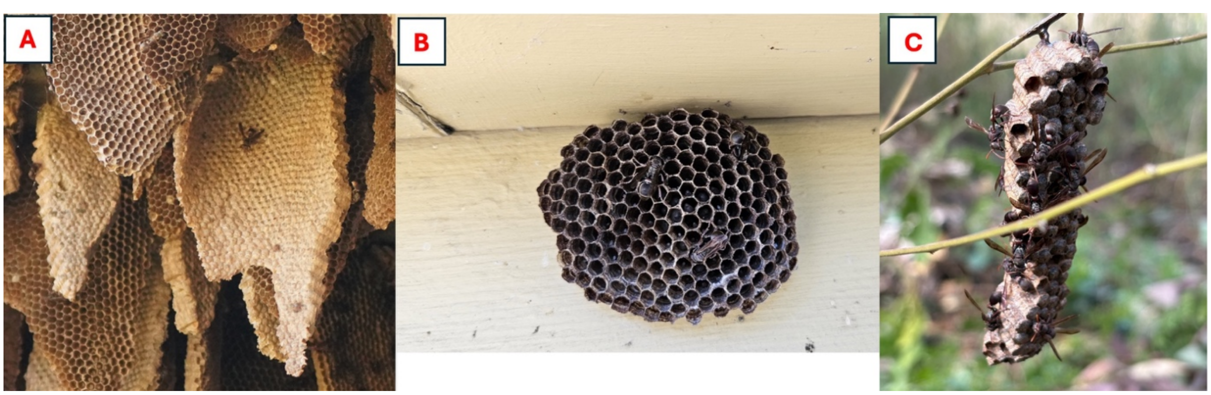
Ropalidia wasps, like other Polistinae, are eusocial. Typically, a single reproductive female gyne (or "queen") is present per nest, though larger colonies may temporarily have several. Workers, also female and similar in appearance to gynes, handle nest building and caring for young. The establishment of a female in the role of the reproductive gyne, whilst other remain as workers, is mainly determined by size and the process is conducted largely without violent interactions (Fukuda, 2003). Males are often found at the nest but only rarely assist; their main purpose is to seek mates (Kojima 1993).

Figure 1. ***Ropalidia plebeiana*** **at Ulladulla, NSW. Photo credit: P. Warburton**

Ropalidia plebeiana are also notable for their large, aggregated nesting sites. Such sites are not "super-colonies" but an aggregation of multiple independent nests, usually numbering in the hundreds (Itô 1987). These aggregations, or "cities", are often in out-of-the-way locations, such as under bridges over creeks and on rocky overhangs on cliffs. Occasionally, these cities are abruptly abandoned, and the wasps relocate to a new nearby site. The preference for nesting in aggregations occasionally extends to *Ropalidia plebeiana* sharing a nesting site with another *Ropalidia* species (Hook 1982).

Most nests are found in the aggregation sites, but there are also some solitary nests positioned apart from others. The nests typically consist of a single layer of cells arranged in a comb. They may be suspended pendulously from an overhead structure with horizontally oriented cells, in an irregular plate-like form (Figure 2A) or instead attached directly to the substrate, with cells oriented horizontally, vertically downward, or at various angles (Figure 2B). Nests may also be in a stick-like form (Figure 2C), again either in a pendent style or attached flush with the substrate.

Figure 2. Three nest styles. Photo Credit: P. Warburton (A) Morpus (B), Chris William (C)



This diversity in nesting styles distinguishes the species, as most other wasp species generally display a consistent nest architecture with limited variation. It seemingly represents an unusual degree of plasticity in nest making; insects are more notable for rigidly programmed, innate behaviours in nest building (Sane 2020).

With the modern availability of a wider base of observational data from citizen science (also known as community science) observations, there is now an opportunity to study a larger number of sites and to review the nesting habits over the full geographic range. This study seeks to address that opportunity. The availability of high-resolution cameras and drone technology has allowed a photographic examination of wasp behaviours over a larger number of sites. We address the question of why individuals of this species have multiple different styles of nest building and explain their preference for living in large nest aggregations in northern locations, with a lower propensity for aggregation in more southern locations and highland locations. We consider what leads them to periodically abandon these "cities" and relocate nearby. We address the question as to why they sometimes construct solitary nest sites. We consider these characteristics in light of the innate behaviours of the species.

METHODS AND MATERIALS

Records from the Global Biodiversity Information Facility (GBIF) were examined and taken for analysis in September 2025. A total of 373 photographic observation records of *Ropalidia plebeiana* were extracted and analysed along with records from 28 personal observational visits to 15 sites in September to November 2025. Of those records analysed, 42 aggregated nesting sites and 94 solitary nesting locations had adequate associated photographs for this study. Identifications to species were confirmed by at least two expert identifiers in each case.

The aggregated nesting sites that were visited were at Brogo at the southern-most extent of where aggregations are found, several near Batemans Bay on the New South Wales south coast, Tahmoor in the mid-part of the range, and Coffs Harbour in the northern part of the range. The photographic observations from community science databases provided further information across the entire range of the species from around the Brisbane area to southern Victoria and the Australian Capital Territory (Appendix 2, Figure 15). The 94 solitary nests that were studied were across the range of the species (Appendix 3, Figure 16).

GBIF gathers data from various sources. For *Ropalidia plebeiana*, this includes museum collections and citizen science platforms - predominantly iNaturalist, but also Naturemapr, Questagame, Naturewatch, and Insect Investigators. These records and their associated photographs were reviewed, the data was checked for accuracy, and each observation was classified according to nesting behaviours. Additional information on sightings was obtained, when necessary, through communication with observers, analysis of Google Maps, and site visits.

Pendent nests were defined to be those that were hanging by one end of the comb from an overhead substrate; the cells of the comb were horizontal or nearly horizontal.

Non-pendent nests were those that were constructed in the plane of the substrate, usually with multiple peduncles throughout the comb. In this case, the cells were either horizontal or pointing vertically downwards, with some at varying angles between the two.

The nests were categorised into groups based on shape. Many of the nests were of an irregular "plate" shape ranging from roughly circular to roughly ovoid, with a flat edge at the point of attachment to the substrate. "Stick" nests were defined to be when the length was greater than 3 times the width.

To avoid disturbing the wasps, and to allow a safe study of the nests, the site activities were studied from about 2 to 5 metres to the nest aggregations using a remotely operated, high-resolution camera with an 800 mm telephoto lens. Activity and location data were logged. A drone was employed to view sites that would otherwise have been inaccessible.

Records were prepared of the details of the nest sites, surrounding vegetation, and topography of the area. Note was taken of any historical information about the nesting site. Botanical studies were carried out in the area around some of the aggregated nesting sites.

Samples of fallen nest combs were obtained from wind damaged nesting sites during the winter dormancy period.

RESULTS AND DISCUSSION

The analysis revealed the very strong tendency to nest in aggregation but with some wasps instead making solitary nests. Nest types vary over time and geography.

Table 1 shows the estimated split between the nest types in the 42 aggregations that were studied (Figure 15 in Appendix 2). Note that these are approximations based on the estimated numbers of nests at each site and the estimated split between the type in each case.

Table. 1 Estimated number of nests by type in the aggregation sites studied.

		Number of nests	%
A)	Pendent plate nests	2,000	*19%*
B)	Non-pendent plates	4,600	*42%*
C)	Pendent Sticks	4,200	*39%*
D)	Non-pendent sticks	0	*0%*
		10,800	*100%*

The analysis confirmed the paucity of large, aggregated nesting sites in cooler climates; Victoria had just four small aggregation sites that had been recorded on the community science databases, and there were no observations recorded in the Australian Capital Territory. The largest aggregation sites are in Queensland and northern New South Wales. Observations of solitary nests are much more common in Victoria and central/southern New South Wales than in the northern areas of the range.

Aggregation sites in the northern parts of NSW and Queensland were more likely to be relatively exposed to the elements, compared with aggregations in central and southern areas of New South Wales, where overhanging rocks, and similarly sheltered structures under bridges, were strongly preferred.

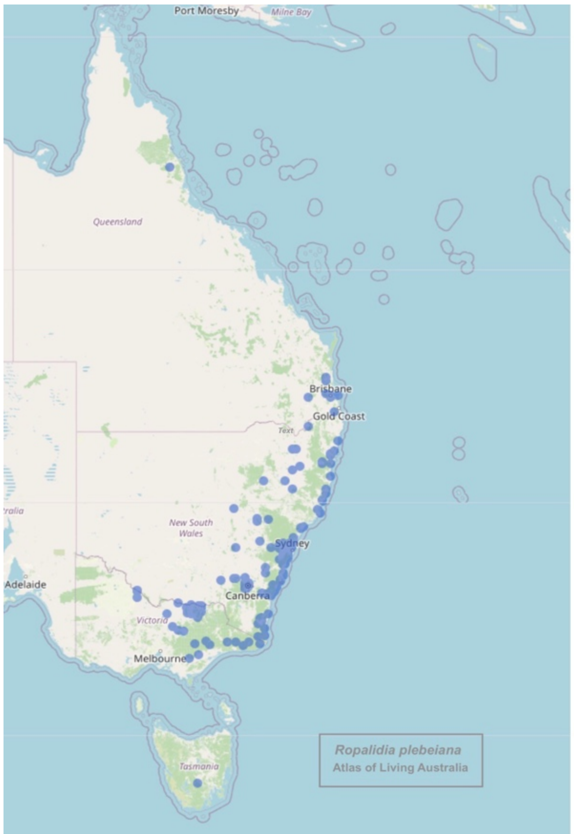
Botanical surveys of large, aggregated nesting sites confirm the richness of the plant life around large aggregation sites. Eight such surveys were carried out and show that the area within 50 metres of each large site contained many trees, shrubs and smaller plants. These usually included at least a dozen different flowering species that would sustain the colony with nectar and foraging opportunities throughout the active months (Appendix 1).

The timing of the wasps' emergence from dormancy coincided with flowering times for several of the plants mentioned in the surveys, especially Acacia species. The emergence from dormancy, in the central areas of New South Wales, lags the northern part of the range by about two weeks. Canberra and the southern parts of the range, in south coast areas of New South Wales and Victoria, lag by a further two weeks. The flowering of Acacia and other plants in those areas follows a similar pattern. A minority of the nests studied were solitary nests, constructed outside of aggregations (Table 2).

Table 2. Number of nests by type in the solitary sites studied.

		Number of nests	%
A)	Pendent plates	28	*30%*
B)	Non-pendent plates	39	*41%*
C)	Pendent Sticks	24	*26%*
D)	Non-pendent sticks	3	*3%*
		94	*100%*

Figure 3. Range of *Ropalidia plebeiana* (left) compared to climatic regions (right)
Ref: Atlas of Living Australia and Australia Bureau of Meteorology



Ropalidia plebeiana is the only temperate climate species in the genus (Figure 3) - the other members are mainly confined to tropical and sub-tropical regions.

The species has been able to exploit the arrival of man-made structures as advantageous sites for their nest cities. Several of the sites we observed were mentioned in a 1985 study, indicating that such nest aggregations have survived for decades (Itô 1985), though the exact site of the aggregation within a larger location sometimes changes over time; they may move from one side of a bridge to the other for example.

The nesting aggregation behaviour of this species is a kind of defensive cooperation of independent reproductive units that is rarely found in the animal kingdom.

Importantly, females oviposit in old cells i.e. combs are used for more than one year. This behaviour contributes significantly to the reproductive efficiency in the species, increasing the potential for successful egg development (Itô & Higashi 1987).

A contributing factor in the success of the species is that the active nesting period is long; about 10 months in the northern parts of the range and about 8 months in the southern-most areas, which is longer than typical nesting periods for related temperate polistine wasps (Richards 1978). Most new foundresses are observed to reappear from dormancy at their natal nests in early spring (mid-August to mid-September) to quickly establish colonies by reusing existing nest combs for ovipositing. The pre-emergence stage, referring to the duration before the first adult brood appears, lasts around 3.5 months (Saito 2005).

In Tables 1 and 2, we quantified the three nesting styles that predominate in aggregation sites as well as in solitary sites. These nesting styles are as follows:

A) Pendent-plate nests

Often the pendent nests are in an irregular plate-shape, suspended from an overhead structure, such as the lip of a bridge. Plate-shaped nests typically develop to about 10-20 cells wide and 25-35 cells long, about 450 cells in total, but they can sometimes be as much as six times that size. The orientation is usually with the cells laid horizontally (Figure 2A). This is the style most common on bridges over creeks between Sydney and Brogo in south New South Wales, for example the aggregation at Cabbage Tree Creek, on the Kings Highway near Batemans Bay, NSW (Figure 4).

Figure 4. A Nest aggregation at Cabbage Tree Creek, NSW which is mainly pendent plate form nests. Photo credit: P. Warburton

B) Non-pendent plate nests
In most aggregated nest sites, there is a vertical surface that does not lend itself to pendent style combs. In these cases, the combs are attached in the plane of the substrate with multiple peduncles (Figure 2B). In some cases, this is the predominant form in the aggregation. This type of aggregation, on more exposed flat surfaces, is more likely to be found in the northern parts of the range where it is the preferred style on highway overpasses and vertical rock faces (Figure 5).

Figure 5. An aggregation at Coffs Harbour which is predominantly non-pendent in style. Photo credit: L. Copleand

C) Pendent stick nests
Some pendent nests are more stick-like in shape, and they are found particularly in older, long-established nests. This is the result of a unique process of nest fission. In spring, new foundress females often use their mandibles to cut a prior season's nest-comb, dividing it into two or more independent nests. These reproductive gynes will often do this with subordinate female helpers (Tsuchida, 2022).

The fission of nests into new independent communities is handled with only limited conflict between the females; some pecking and barging but no obvious attempts to sting, confirming prior observations (Fukuda, 2003). This relatively moderate confrontational behaviour is key to the successful operation of such large, cooperative, multi-nest communities (Itô 1985).

Before the nest division, each principal egg-laying female and her subordinates occupy a distinct region of the same comb. These females gnaw at cells within the intermediate zones separating such "territories," eventually resulting in the division of the comb into independent combs, which they simultaneously secure to the roof with new attachment peduncles (Figure 6). The formation of new nests via comb-cutting has been shown to account for as much as a third of the total increase in nest numbers. This unique form of colony fission has, so far, only been documented in *Ropalidia plebeiana* (Yamane,1991; Makino,1994; Oberprieler & Spradbery 2009).

Where nest expansion by fission of old nests predominates, this results in additional nests within the same confined space. The divided nests are longer and narrower than the original nest and there is only room to expand them by extending them in the vertical plane. The result is stick-like nests of variable dimensions but typically 6 to 10 cells wide and 10 to 30 cells long. At the same time, the other females that are starting new nests overwhelmingly do so within the perimeter of the existing aggregation area when there is any space to do so (Figure 8) and so are constrained by congestion into building stick nests that expand in the vertical plane. As time passes, older aggregations increasingly consist of stick type nests if the site has been spared from wind damage for an extended period (Figure 7).

Figure 6. Nest fission taking place. The yellow arrows show where females are dividing older nests. The nest division at the top of the photograph appears to be itself the product of a prior division that included the two segments above it. Photo credit: P. Warburton

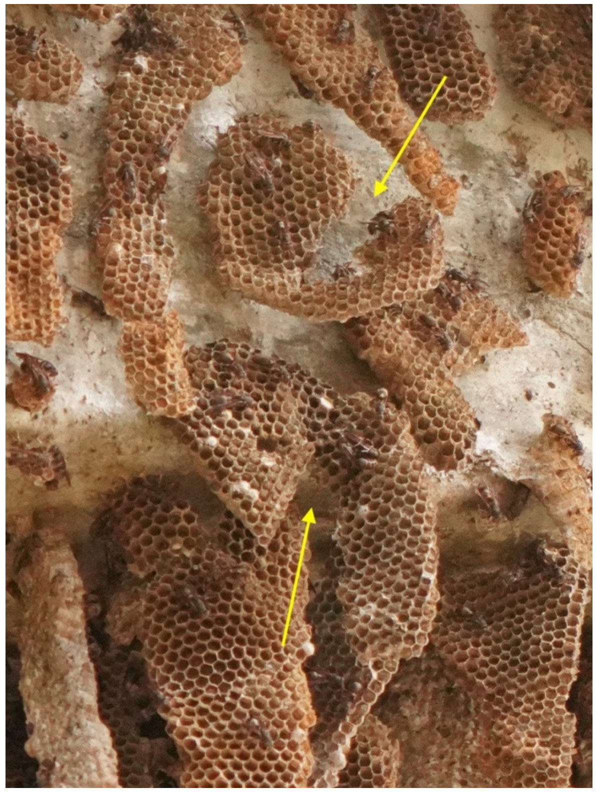


Figure 7. An older aggregation at Tahmoor, NSW with a predominance of stick-like nests. Photo credit: P. Warburton

Apart from wind damage, the nests are well suited to usage over more than one season. The combs are hydrophobic and resistant to water damage. In this study, one of the fallen (unoccupied) nests was immersed in water for 24 hours and then left to dry out. It suffered no visible damage to the structure, and the cells were completely intact.

Pendent stick nests also occur in solitary nest locations. An example is shown in an observation in Prestons, NSW (Figure 2C). Pendent stick nests are usually found in vegetation where there is only a single point of attachment and where obstructions such as twigs constrain development of the nest laterally.

Preference for Aggregated Nesting

Several contributing factors may account for the tendency towards aggregated nesting in *Ropalidia plebeiana*. Aggregations of nests may allow for shared communication about the location of food sources (Richards 1972). The joining of nests in an aggregation provides cooperative protection, for a species possessed of only limited defensive capability at an individual level, making aggregation a successful evolutionary strategy. Certainly, wasps from multiple nests, will fly out to attack any person who gets too close to the aggregation site (Lin and Mitchner 1972).

The species displays notable philopatry, with new adults typically nesting at their natal site. It has been theorised that, throughout the evolutionary history of *Ropalidia plebeiana*, nesting sites under cliff overhangs of a size that facilitates aggregation, may have been relatively limited, making the search for new sites a significant investment. Remaining at known sites could thus have conferred a selective advantage. This pattern appears to persist even though suitable man-made structures are now more widely available (Itô, 1987). This innate behaviour of philopatry leads to pendent nest structures as the method of using the limited space to the maximum, until resource constraints lead to exploration of alternative sites.

We suggest that the historically limited number of rocky outcrops, that could sustain an aggregation, made this niche too small to support aggregation as an evolutionary strategy more widely in the social wasps.

Aggregated nesting sites in *Ropalidia plebeiana* wasps reflect the "selfish herd" instinct, as individuals prefer central locations in the aggregation for protection, leaving those on the edges at greater risk (Hamilton 1971; Itô 1988). The centripetal tendency to build well-within the existing boundaries continues until available inner space is exhausted. At Tahmoor, about 95% of new nests in early spring were built within existing boundaries to fill gaps from lost nests. Similarly, Nelligen Creek saw early-season nest rebuilding focused within the existing aggregation perimeter, after a gale in late winter 2025 caused the loss of about 10% of the nests. (Figure 8).

The periodic abandonment of sites due to excessive wind damage is, in turn, increasingly likely as aggregations age, due to the greater vulnerability of the long, thin stick nests. These increase in prevalence over time due to the nest splitting and congestion withing the perimeter of the aggregation.

Figure 8. New nest start-ups predominantly exploited gaps within the existing aggregation perimeter. Photo credit: P. Warburton

Nest city location

Historically, nest aggregations would have been found under overhangs on cliff faces. With the advent of man-made structures, bridges and other similar constructs are more plentiful and have also become preferred nest aggregation locations. Concrete or rock are the preferred substrates for anchoring nests with other materials only occasionally being used. In this study, 22 of the 42 aggregation sites were described as "medium" (100-300 nests) to "very large" (>500 nests) and all of these were attached to rock or concrete (though there were three of those sites that were mainly concrete but included a section of galvanised steel). Metal surfaces are not usually favoured – apparently because nests are more easily blown off when the attachment points are to these materials. Where metal surfaces are used, the nest aggregations are small, and the nests are usually located flush with the substrate with many peduncles used for attachment. On one of the bridges we studied at Nelligan Creek, a small section of steel girder was within the perimeter of the aggregation, the rest of the area being concrete. This area of nest aggregation on steel substrate was impacted disproportionately by wind damage. However, after the winds the wasps set to work to fill the gap, presumably with a future risk of them being blown down again. Apparently, the instinct for centripetal nest building over-rides the aversion for attaching nests to a smooth steel surface.

Botanic surveys indicate that, where there are large aggregations, the surrounding vegetation is consistently rich in flowering plants and provides ample food resources (Appendix 1). A large aggregation of wasps means competition for nectar resources for feeding and for prey resources for the larvae. A study of the site at Tahmoor in early spring revealed that most wasps feeding on nectar were found within 50 m of the nest site; none were found at 100 m from the site, despite the large population of thousands of wasps. This site was typically well provisioned with many flowering shrubs and trees, comprised of over a dozen species within the 50 m radius. Such well-provisioned sites are often available in the warmer northern regions and are fewer in the south of the species' range. Later in the nesting season, when the wasps focus more on finding prey insects for feeding larvae, wasps are found more widely looking for food in the surrounding area.

In the northern parts of the range, there is a clear tendency to nest on vertical surfaces, particularly on concrete bridges including highway overpasses, with non-pendent nests. By contrast, nests in the southern parts of the range tend to be pendent style.

To understand the reasons for the different styles across the range, it is essential to consider the reasons for the nest site development. We propose that the ethological mechanism for the development of aggregations is simply one of trial and error, coupled with complete abandonment of failed sites. Nesting locations are tried, and aggregations develop and become larger over time if the site is successful. Periodically there may be a catastrophic event that destroys a large part of the aggregation. When this occurs, the wasps often completely abandon the site and re-establish in a new location and try again.

The factors that contribute to a site being successful in the north differ from the ones that contribute in the case of more southern locations. In the south, well-sheltered locations with plentiful plant resources nearby are often bridges over creeks with a rich riparian habitat in a sheltered gully. Such sites are relatively scarce and high-density aggregations with pendent nests are prevalent.

Northern regions, with their warmer climates, offer more nesting sites near adequate food sources and these northern areas have more extensive highway infrastructure than the south, hence with more plentiful man-made nesting sites in the form of bridges. However, nests in the flat terrain associated with highway bridges are highly exposed to wind; pendent nests are usually destroyed, while sturdier, non-pendent nests, often attached to a vertical surface, survive. These surviving nests define nest aggregation perimeters and hence define where new nests are built; the wasps' centripetal instincts lead them to expand within the remaining site perimeter rather than into new areas, leading to a predominance of non-pendent forms of the nests.

Most independent solitary nests are amongst vegetation, for example under the shelter of leaves or a branch, and bushfires are a threat to such locations. The extensive 2019/2020 bushfires devastated much of the geographic range of the wasp (Figure 9). However, the preference for nesting in sheltered locations under concrete bridges and under rocky outcrops is clearly beneficial in mitigating fire impacts. In this 2025 study most of the decades-old nest aggregations in the Eurobodalla area of the New South Wales south coast, listed by Itô and Higashi in 1987, were still present and were thriving, despite this area being particularly impacted by the fires. For example, there are still two aggregations under a concrete bridge across Cabbage Tree Creek on the Kings Highway, NSW (Figure 4) that were studied in 1987.

The successful aggregation locations are usually ones that are sheltered from weather with an overhead roof. Usually there is a vertical structure, immediately adjacent to the aggregation site, to provide wind protection laterally. Additional adjacent structural elements, that add further weather protection, are clearly beneficial. Where aggregations are found on bridges, the nest is usually under the lip of the bridge and adjacent to a structural beam providing lateral wind protection.

A representative site is a bridge in Brogo on the far south coast of NSW. The structure provides wind protection with concrete beams on two sides of the nests, and which also offers some shelter from frost. The location also offers wind protection arising from the local topography; the bridge is in a valley surrounded by hills on all four sides. Observations indicate no evidence of damage from strong gales which took place towards the end of the winter dormancy, and the nest aggregation at this site is large and thriving. The wind and frost protection offered by this location may account for its suitability at the southernmost extent of large nest aggregations (Figure 10).

Figure 9. National Resource Management (NRM) map of the areas of greatest fire intensity in the 2019/2020 bushfires showing the NSW south cost as particularly severely impacted (DAWE, 2020).

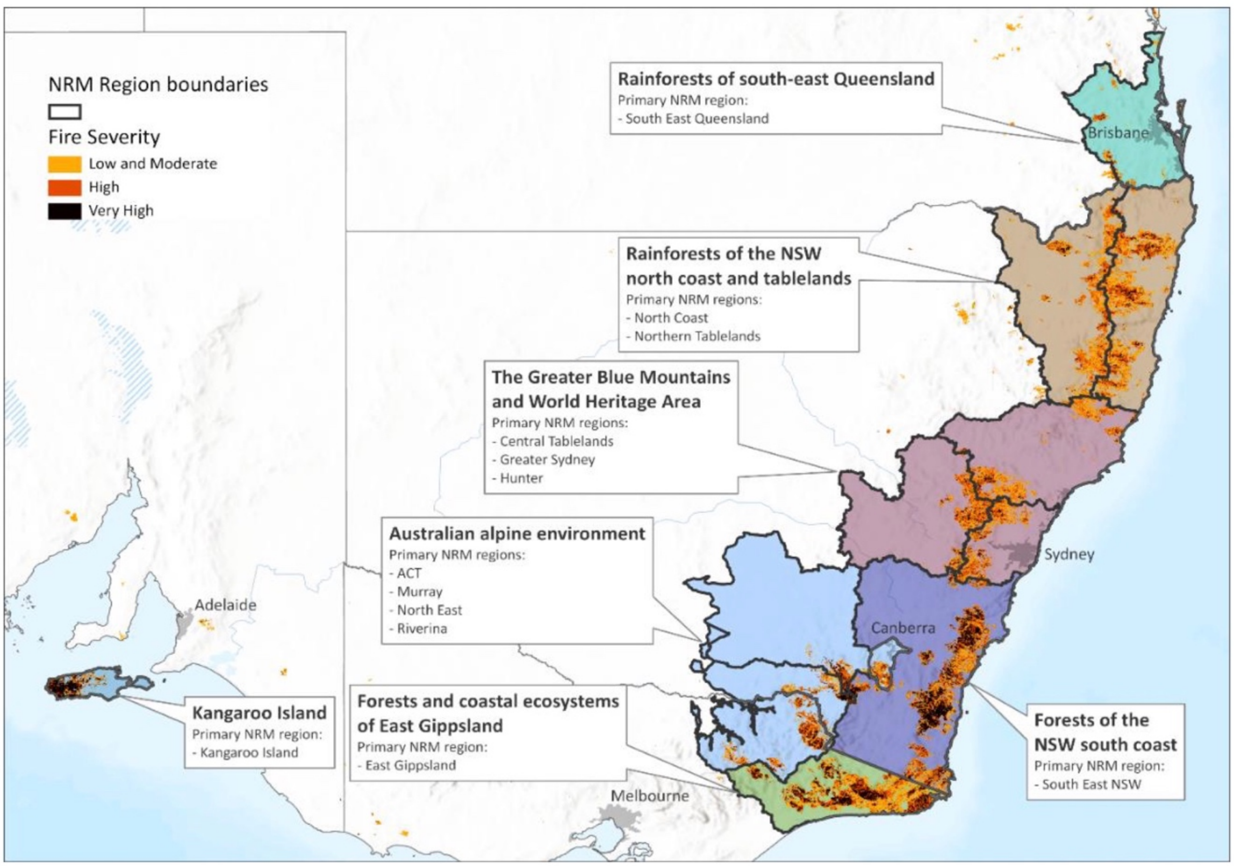


Figure 10. A particularly well sheltered site at Brogo, NSW. Photo credit: P. Warburton

Large nesting groups are mainly found in the northern range, with up to 2,000 nests, while the Australian Capital Territory has none and Victoria has only small aggregations. Solitary nests are more frequent in southern New South Wales and Victoria; 87% of the observations of solitary nests in public databases are south of the mid-point of the species range compared to 13% in the north. Aggregation size decreases further south, giving way ultimately to more solitary nests. Nesting patterns correlate with resource availability and climate.

City layout

Pendent nests near the city's edge are often seen to flap in the breeze, while those farther inside remain protected by outer nests.

The layout of the nests within the city often reflects the degree of wind exposure. Nests at the back of the aggregation and closest to a wind barrier, such as a concrete beam, are often the largest pendent nests. The nests reduced in size towards the weather-exposed perimeter. In many cases, the pendent style of nests gave way to nests comprised of a single layer of cells in the plane of the substrate, where wind exposure was greatest. For sites that feature pendent nests, the result is a tiering of the nests progressively from the front to the back (Figure 11).

For sites with a high exposure to wind, pendent nests do not persist, and the non-pendent nest form predominates.

Threats to aggregated nesting sites

The main threat to the nests, particularly those at the peripheries of the wasp cities, is wind (Itô, 1987). Significant damage from wind was evidenced in several of the sites studied.

Attacks on the larvae by the ichneumonid parasitoid wasp *Arthula plebeja*, have been reported previously (Ubaidillah et al. 2009). This parasitoid wasp was photographed during our study, apparently for the first time, after it was reared from parasitised *Ropalidia plebeiana* larvae (Figure 12).

Figure 11. At Nelligen Creek - showing that the nests with the least exposure to weather are larger than nests near the perimeter. Photo credit: P. Warburton

Figure 12. The ichneumonid parasitoid, *Arthula plebeja*. Photo credit: P. Warburton

Most observed sites had some spider webs at the edges of the aggregations. In such cases, we observed that the nests with spider webs on them had been abandoned; a single web impacting as many as 6-8 nests. Up to 10% of the nests were abandoned in active aggregations because of spider webs. The spiders were clearly having a significant impact on the wasps confirming earlier reports where spiders were seen capturing wasps (Itô & Hignashi, 1987).

In the case of the Nelligan Creek aggregation, a whole aggregation site had been abandoned and was covered in extensive spider webs (Figure 13). We propose that the most likely explanation for the abandonment was excessive levels of spider webbing and spider activity. No other explanation was apparent as the nests were not significantly damaged. Whilst it is possible that the webs were constructed on the aggregation after abandonment, this seems implausible because there would not have been prey insects around the nests to warrant the spider investment in such extensive webs. This impact by the spiders is unusual as the predation relationship between spiders and wasps is generally asymmetric and favours the wasps. A likely explanation is that the spiders establish themselves with well-developed webs, prior to the wasp's emergence from the dormancy period and have some success in trapping wasps, making the location worthwhile. The extensive webs provide the spiders sufficient protection to sustain this choice of a web location. The web style of these spiders indicates that they are probably in the *Badumna* genus.

A small spider was reared from the nests which blew down, emerging 23 days after the nests were collected. This was identified as *Intruda signata*. It is possible that this is a second species of spider that is predating on the wasps, though this may have been introduced after the nest fell to the ground. An adult dermestid beetle, *Anthrenus verbasci*, was also found in a fallen nest and was presumed to be feeding on nest material and contents. It is not known if these beetles represent a threat to nests that are in use, however dermestid beetles are a known threat to other hymenopteran nests (Motyka 2022).

The spider threat may be partially reduced by the presence of other wasps. Mud wasp nests, including those of Sphecidae like Sceliphron, were often found close to the paper wasp aggregations. These spider predators may benefit from some protection for their larvae by the proximity of the paper wasps, whilst the mud-dauber adult females contribute to constraining the number of spiders threatening the nests of *Ropalidia plebeiana.*

Figure 13. Extensive spider web encroachment of the abandoned aggregation site at Nelligen Creek. Photo credit: P. Warburton

Birds are also a threat to the wasps. For example, the Nelligen Creek site transitioned from dormancy to an active nest site in the period between the 10th and 12th of September 2025, coinciding with the arrival of Welcome Swallows from their migration. These swallows commenced the construction of about a dozen nests in the bridge structure. The birds were making continuous feeding flights around the wasp nest aggregation and certainly would have been making a significant impact on wasp numbers. A Grey Fantail was seen visiting the nests at Nelligen Creek.

The various threats faced by the nesting sites illustrate some of the reasons why the defensive strategies of aggregation and centripetal development have evolved.

Site abandonment

There are several instances where aggregation sites are completely abandoned following a catastrophic event. At Sheep Station Creek, an older aggregation had been abandoned following damage from inundation during heavy rain. Nelligan Creek had two old aggregations that had been abandoned, apparently due to wind damage and a third abandonment was possibly the result of excessive spider incursion. Gales appear to have impacted the aggregation at Tahmoor, causing a relocation of the aggregation to a new position on the bridge.

Eventually, after multiple relocations, a site is found where the risks are minimal, and the site is unaffected by any catastrophic event for an extended period. Some locations on bridges, for example, continue to have nest aggregations for years and even decades, with periodic relocations a few metres along the bridge.

The process of site abandonment appears to be an innate behaviour and an important component of the process for the development of aggregations sites. Over an extended period, the abandonment of unsuitable sites represents a selection process that eventually results in nesting sites with all the ideal characteristics to support large aggregations.

Some questions remain unanswered in this process and require further investigation. It is notable that there do not seem to be any wasp stragglers that remain at the old nest aggregation sites. When a whole site is abandoned, what is the mechanism that causes the aggregation of wasps to collectively move elsewhere without exceptions? The answer may relate to the centripetal instinct. It maybe that when a majority leave, the remainers find their innate requirement to be at the centre of an aggregation - effectively a herd instinct - is no longer being met. Further study may clarify the mechanism in more detail.

CONCLUSION

Ropalidia plebeiana is an iconic species in temperate regions of Australia. It exhibits some remarkable characteristics in variability in its nest building. This study has clarified the nest behaviours over the full geographic range which had not been possible when the species was studied in the 1970s and 1980s.

The apparent plasticity in nest-type variability is a manifestation of several innate behaviours that are followed as rigidly as circumstances allow. The most important of these innate behaviours are:

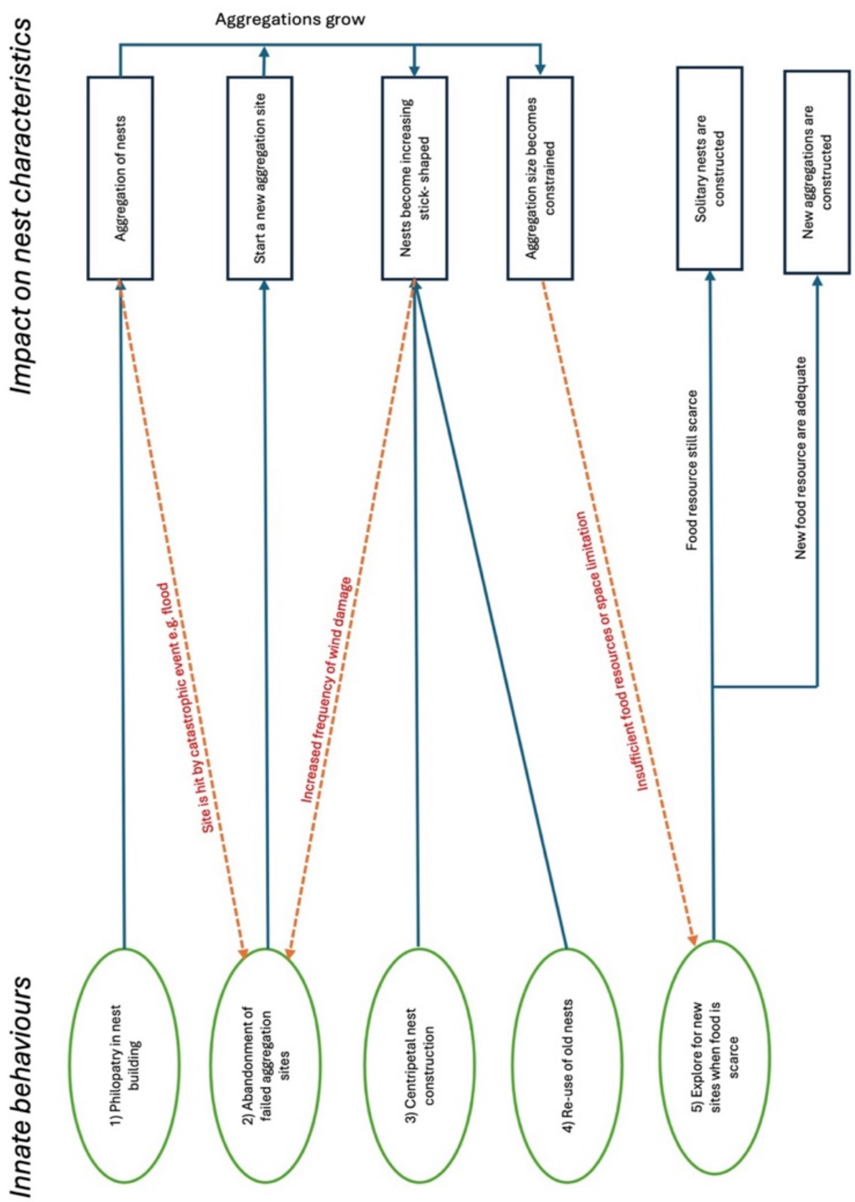
* the instinct for philopatry in nest choices as the evolutionarily proven method for best dealing with the limited number of suitable sites,
* the abandonment of a site when hit by catastrophe,
* the instinct for centripetal nest building for protection,
* the reuse of old nests when they are available, for reproductive efficiency, and
* the need to seek new nesting sites when food resource constraints prevent natal site expansion.

These key innate behaviours impact on nest structure, size, location and its development pathway and these are summarised in Figure 14.

We propose that the different nesting styles are simply a reflection of the extent to which these innate behaviours can be acted upon, given the constraints of the wasps' local environment over time and across its geographic range.

Future studies may cast further light on these other innate behaviours that are part of the ethological mechanisms for nesting characteristics.

Figure14. The impact of innate behaviours on nest characteristics



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Appendix 1 – Survey of plants within 30m radius of the aggregated nesting sites

The botanical surveys can be summarised as follows:

Survey site 1 -Tahmoor, NSW
Many flowering plants including a dozen species within 30 m, particularly Acacia decurrens and Acacia longifolia. Other flowering plants at that location included: Indigofera, Philotheca, Clematis, Hardenbergia violacea, Pomaderris, Pseudanthus, Dillwynia as well as some introduced species such as Senecio madagascariensis and Gerbera/Gazania.

Survey sites 2,3 and 4 on the Pacific Highway in northern NSW
The main plant species flowering during the visit were introduced weeds (e.g. Lantana camara and Ageratum houstonianum) but there were plenty of native flowering plants (e.g. Eucalyptus saligna, E. microcorys, Glochidion ferdinandi, Acacia melanoxylon, Callicoma serratifolia and Synoum glandulosum) also present, within 30 m of the site, during the September survey and would flower well at other times throughout the year.

Survey site - 5 Brogo in south coast NSW
In the immediate vicinity there were Acacia, several Eucalyptus and Casuarina that were yet to flower and introduced species such as Senecio madagascariensis and Trifolium. There were a dozen or more flowering Prunus cerasifera bushes 100 m away.

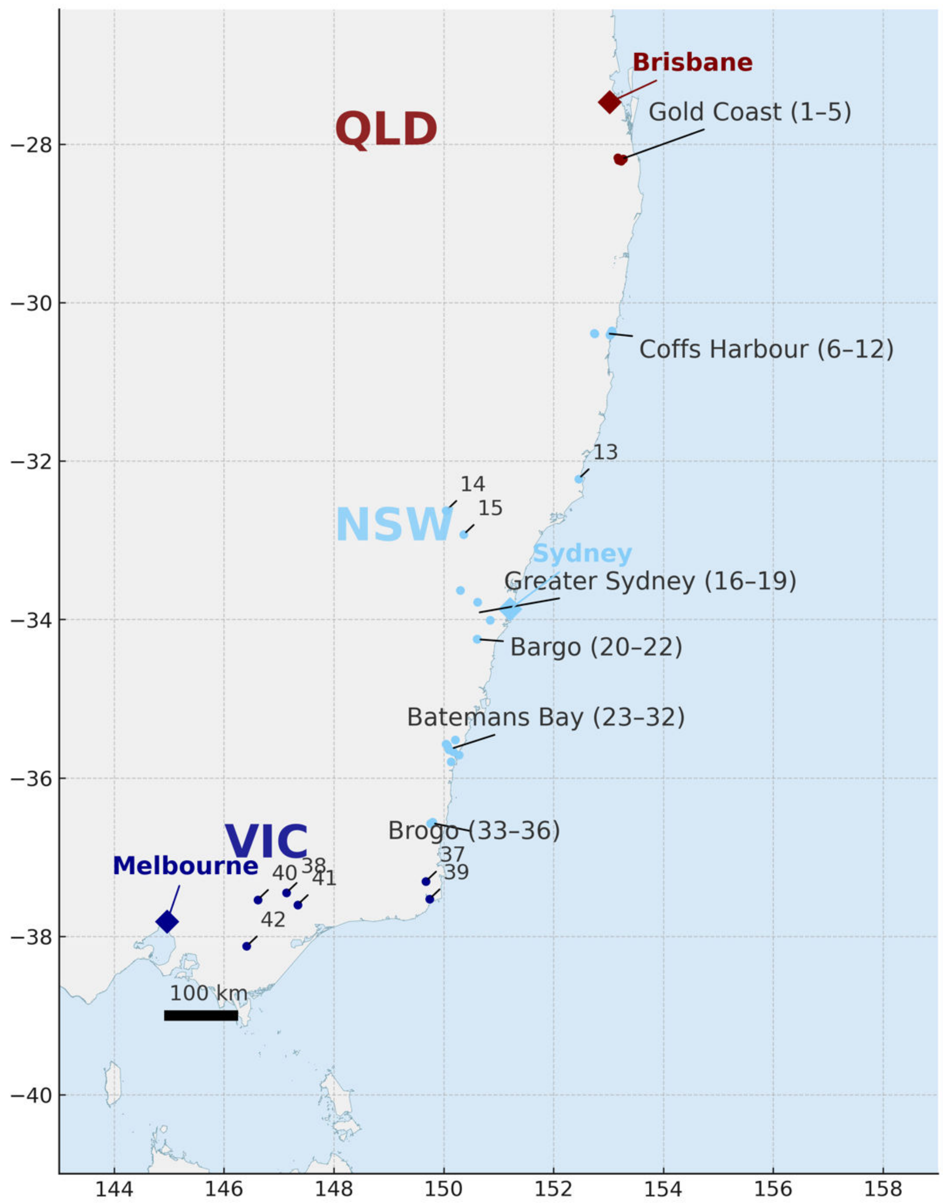
Survey site 6 - Nelligen Creek, South Coast NSW
Acacia mearnsii, several Eucalyptus species, Casuarina spp., Bursaria spinosa, Solanum aviculare, Myoporum bateae, as well as some introduced species including Acer negundo, Lilium formosanum and Prunus cerasifera.

Survey site 7 - Jeramandra Creek, Mogo, South Coast NSW
Acacia mearnsii, several Eucalyptus species, Casuarina, Gaudium trinervium and some weed species including Solanum mauritianum. There is a domestic garden 100m away with many cultivated flowering shrubs and smaller plants.

Survey site 8 - Lyons Rd overpass over Pacific Hwy, Coffs Harbour
The main plant species flowering during the visit were introduced weeds (e.g. Ageratina adenophora, Ageratum houstonianum and Ipomoea cairica) but plenty of native flowering plants (e.g. Melaleuca quinquenervia, Polyscias sambucifolia, Pittosporum undulatum, Lomandra longifolia) are also present within 30m and would flower well at other times throughout the year.

Appendix 2 – Aggregation locations that were studied (see also key below the map)

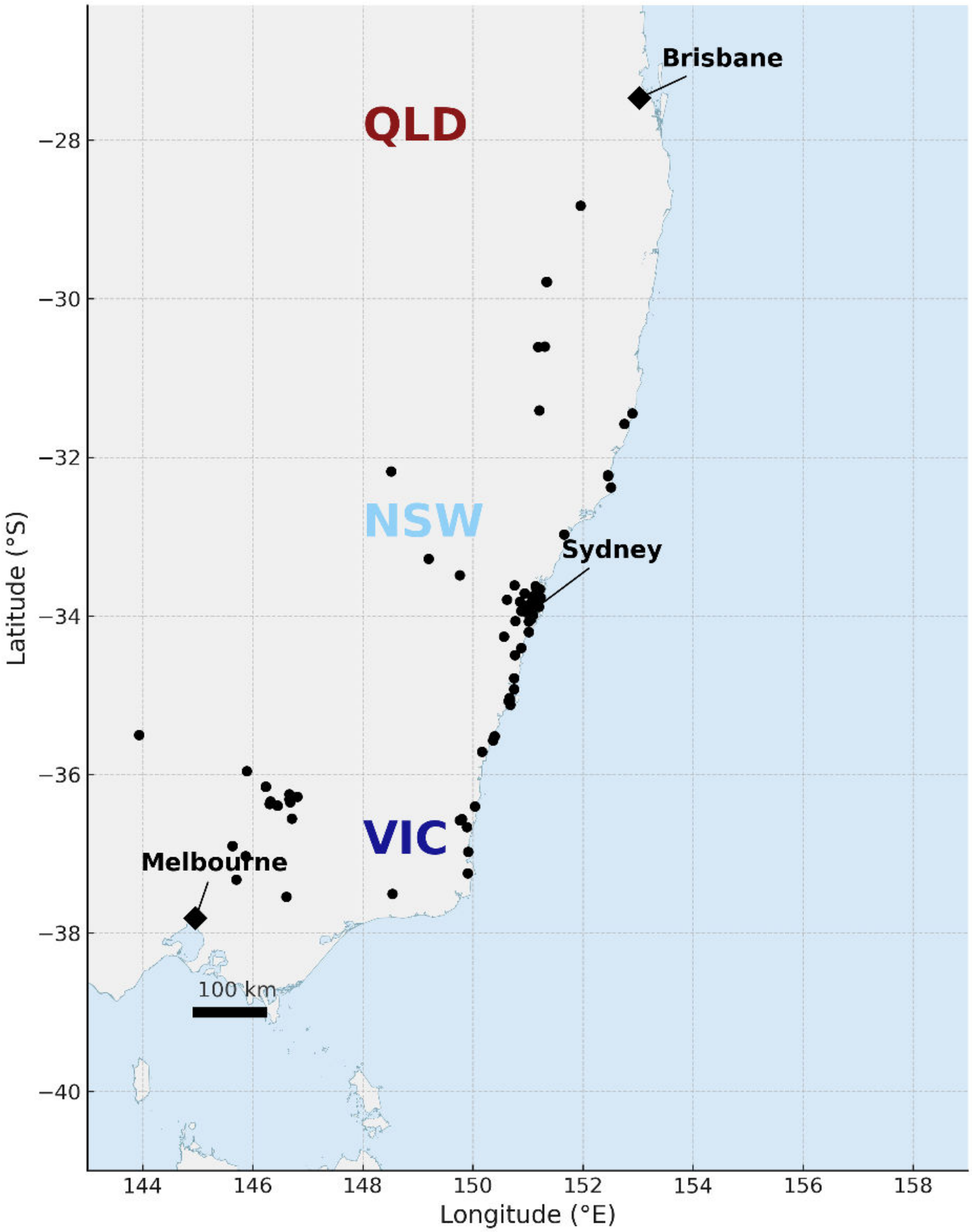
Figure 15. Aggregation locations (see key below).



Appendix 3. Solitary nest site locations

	Location	State	Latitude	Longitude
1	Binna Burra	Queensland	-28.169958	153.169206
2	Springbrook	Queensland	-28.186817	153.269502
3	Guanaba-Springbrook	Queensland	-28.187241	153.267439
4	Lamington National Park, Binna Burra	Queensland	-28.199550	153.182810
5	Nerrang	Queensland	-28.211660	153.233950
6	Lyons Rd Pacific Highway	New South Wales	-30.354597	153.063179
7	Bonville	New South Wales	-30.376288	153.067135
8	East Bonville	New South Wales	-30.385424	153.042517
9	Dorrigo Mountain	New South Wales	-30.387628	152.740750
10	Dorrigo Mountain	New South Wales	-30.394163	152.745536
11	Northbound Pacific Hwy Coffs Harbour	New South Wales	-30.407588	153.027441
12	Southbound Pacific Hwy Coffs Harbour	New South Wales	-30.408658	153.027571
13	Coomba Park	New South Wales	-32.226185	152.454800
14	Growee	New South Wales	-32.626147	150.044311
15	Wollemi	New South Wales	-32.928911	150.360977
16	Blackheath, Australia	New South Wales	-33.631857	150.304845
17	Near Red hands cave walking track. Blue Mountains	New South Wales	-33.779695	150.616897
18	Obscured	New South Wales	-34.010778	150.844855
19	Bargo River, Rockford Road Bargo	New South Wales	-34.249504	150.607112
20	Rockford Rd Bridge at Tahmoor,	New South Wales	-34.249589	150.607097
21	Rockford Rd Bridge at Tahmoor	New South Wales	-34.249589	150.607097
22	Bargo River, Rockford Road Bargo.	New South Wales	-34.249610	150.607204
23	Mogood	New South Wales	-35.520798	150.208314
24	Cabbage Tree Creek on Kings Highway (1)	New South Wales	-35.572795	150.040711
25	Cabbage Tree Creek on Kings Highway (2)	New South Wales	-35.572795	150.040711
26	Dinner Creek on the Kings Highway, Currawan	New South Wales	-35.591285	150.064419
27	Nelligan Creek Bridge on Kings Highway (1)	New South Wales	-35.641475	150.093876
28	Nelligan Creek Bridge on Kings Highway(2)	New South Wales	-35.641475	150.093876
29	Nelligan Creek Bridge on Kings Highway (3)	New South Wales	-35.641475	150.093876
30	Sheep Station Creek	New South Wales	-35.670084	150.173359
31	Honeysuckle Beach, Murramurang	New South Wales	-35.710838	150.276425
32	Jeremandra Creek on the Princess Highway near	New South Wales	-35.795869	150.133981
33	Brogo , bridge over Alsops Creek on Hawks Head	New South Wales	-36.560541	149.799268
34	Brogo bridge over Alsops Creek on Hawks Head Rd	New South Wales	-36.560541	149.799268
35	Brogo (3)	New South Wales	-36.577283	149.762578
36	Domestic Porch, Brogo (4)	New South Wales	-36.577324	149.762560
37	Timbillica	New South Wales	-37.306579	149.674346
38	Hawkhurst	Victoria	-37.449134	147.137334
39	Mallacoota	Victoria	-37.530123	149.740175
40	Licola	Victoria	-37.538136	146.614766
41	Mitchell River	Victoria	-37.603000	147.339000
42	Yallourn North	Victoria	-38.125036	146.412214

Figure 16. Solitary nest site Locations



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REVIEW OF THE GONDWANAN APHIDS

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Summary

Endemic and indigenous aphid species are rare in most Gondwanan countries, being greatly outnumbered by aphids accidentally introduced from Europe or North America, or spreading naturally via Southeast Asia. Our limited knowledge of Gondwanan endemic and indigenous aphids is summarised here, with emphasis on Australia and New Zealand. Many undiscovered species are almost certainly present. Barriers to their discovery are discussed. Associations of aphids with ancient and endemic plants are considered in relation to continental history. Aphids have close functional relationships with a range of other organisms, including ants, wasps, flies and bacteria and some of the obligate relationships may also be Gondwanan. *Neophyllaphis*, *Neuquenaphis* and *Sensoriaphis/Taiwanaphis* have an unquestionable Gondwanan origin, as do southern *Aphis* and *Paradoxaphis* (Australia, New Zealand, South America). The subfamily Lizeriinae may have been the first to diverge from the basal Aphididae and has an exclusively Gondwanan distribution. The subfamily Greenideinae is another early group and is composed of genera with a Gondwanan distribution. Species in these genera occurring in both Australia and Asia/Southeast Asia can be considered native, but not endemic to these areas. *Greenidea* and *Schoutedenia* are thus Australian natives. *Aphis clerodendri* and (less convincingly) *Aphis eugeniae* have a Gondwanan distribution. The genus *Sitobion* (Macrosiphini) has many species probably endemic in various Gondwanan countries and most likely has a Gondwanan origin. Aphidiine wasps (Braconidae), primary parasitoids of aphids, have a well-supported southern origin, and the hyperparasite *Alloxysta*, by virtue of its host-specific relationship with southern aphids and aphidiine wasps, may also have southern roots.
Keywords

INTRODUCTION

Aphid species in Australia have been listed by Eastop (1966), Carver (1998, pers. comm.), Hollis and Eastop (2019) and Brumley (2020). These lists cover all Aphidoidea. The families Phylloxeridae and Adelgidae in Australia each have few species, all of which are introduced to Australia or are cosmopolitan, so the emphasis in this paper is on the remaining family, the Aphididae, made up of 25 sub-families with 30 tribes (Favret, *Aphid Species File).* Relatively few of these sub-families contribute to the extant aphid fauna of Gondwanan countries.

In this paper I use the term "endemic" for taxa that occur in a particular country but not elsewhere. The term "native" refers to taxa that occur naturally, but are also found in other geographic areas. "Southern" is used as meaning Gondwanan, in all its scattered parts including south and Southeast Asia, but not the Kazakhstan, East Turkey and Saudi Arabia elements which separated earlier.

The total number of aphidoid species known in Australia is approaching 200, but the 20+ endemic species are far outnumbered by adventive and cosmopolitan species. Some known but undescribed species are included among the listed endemics. The situation is similar in New Zealand (Teulon and Stufkens 1998, Teulon *et al.* 2002, Teulon *et al*. 2013) with 15+ endemic species in a total fauna of about 135 species. *Neophyllaphis* occurs in both countries and has been thought to be closest in morphology to the earliest aphids, although some recent molecular studies place the Lachninae as an early branching lineage (and no endemic or native lachnine species are known in Australia or New Zealand). It is considered that the

endemic aphids of these and other southern countries originated in Gondwana before its break-up. In this paper, I collate information on the aphids, their host plants, and some of the endoparasitoids and hyperparasitoids of the aphids, as well as the relevant changes in the geographic environment.

THE CONTINENTS

This section and the following two are not intended to be anything more than a background to the time and place of different events.

The landmass of Pangaea, which coalesced during the Early Carboniferous about 335 mya, began to break up in the Triassic Period (252 to 201 million years ago) leaving Gondwana (the name comes from India) in the south and Laurasia in the north. At this stage, Laurasia was mostly north of the Equator and Gondwana mostly south of the Equator but north of the South Pole. There was climate variation across the supercontinent in terms of temperature and rainfall and a widespread but spatially differentiated fauna, with regions marked by patterns of precipitation. There was also a complex flora, again with some division into provinces, and including cryptogams as well as both gymnosperms and angiosperms (see "The Plants" below). During the Jurassic, about 180 mya[1], Gondwana began to divide forming the current Gondwanan continents, including Australia, Te Riu-a-Māui /Zealandia, Africa, South America, India and some fragments now in the Middle East,

[1] mya= million years ago

western Asia and east and Southeast Asia. Metcalfe (2006, 2013) discussed the origins of southeast Asian areas, including South China, Vietnam, Laos and Thailand, which broke away from Gondwana in the Devonian and Permian, but in more recent times were adjacent to later Gondwanan arrivals such as India and Myanmar. Notably, the Devonian is before the time of land animals, or most seed plant lineages, but they are there by the Permian. Part of Indonesia is Gondwanan. Japan's origin is different, being derived from remnants of the early supercontinent Rodinia. The island of Taiwan formed only 4-5 mya, so its flora and fauna are necessarily recent arrivals, either naturally via land bridges to South China or the Philippines during the Pleistocene glaciation which resulted in periods of low sea level, or introduced by human activity. An approximation of these events is shown in Table 1.

The first separations were in Western Gondwana, with India/Africa and South America dividing from each other and from the remainder of Gondwana. India began to separate from both Africa and Gondwana at as late as the Cretaceous and collided with Asia only 50 mya (Chatterjee *et al*. 2006). Australia separated from eastern Antarctica at about 132 mya. At about 84 mya Zealandia separated. Zealandia has recently been accepted as a continent although now largely submerged. It includes New Zealand and islands such as Lord Howe, Chatham, Norfolk and New Caledonia. Compression forces during the early Eocene pushed the continent upwards and inwards and hence there was a continuous exposed south to north land mass connecting all the components that we know today as widely separated. About 25 mya, the weakened crust sank back again, with the exception of the current emergent elements. The geology and history of Zealandia have been detailed by Mortimer *et al*. (2017).

During the history of continental movement, dramatic changes in climate occurred both globally and locally, leading to mass extinction events in each of which well over 50% of insect species are thought to have become extinct (e.g. Crowley and North, 1988). Major extinctions of land plants may not have accompanied the late Permian extinction of many land animals (Nowak *et al*. 2019, McElwain and Punyesena, 2007). It has been suggested that aphids may have suffered disproportionately. Johnson (1992) showed that there was a substantial loss of plants in north America during the Cretaceous extinction event and Donovan *et al*. (2017) discussed the rapid establishment of insect-plant communities in Patagonia after the Cretaceous extinction. The same might be true for other southern continents. Rehan *et al*. (2013) elucidated the diversification of bees following the Cretaceous extinction. Aphids, like bees, are plant dependent and are considered to have diversified strongly during the Cretaceous (von Dohlen and Moran, 2000) and Miocene, especially in the north. I have attempted to summarise major events in Table 1.

FOSSIL RECORD

The earliest known fossil in the Aphidomorpha is *Triassoaphis cubitus* from pre-Jurassic deposits around Ipswich, Queensland (Evans, 1956). From around the same period, *Creaphis theodora* was described from middle Asia by Scherbarkov and Wegierek (1990). The oldest known aphid fossil from Europe is *Leaphis prima* from the Vosges area of France dating back to 174-163 Mya (Jurassic) (Shcherbakov 2010)). *Koonwarraphis* from early Cretaceous Victoria is another fossil from Australia but it is a relative latecomer (Martin *et al*. 2016). Raynor and Waters (1989) described a fossil aphid from South Africa, but it, too, is a Cretaceous latecomer. They discussed other palaeontological evidence and concluded that the origins of aphids were as far back as the Carboniferous.

Szwedo *et al*. (2013) took the origin of Aphidomorpha back to the Middle Permian following discovery of aphid-like fossils in Southern France. Ole Heie (e.g. 1967, 1987, 1994, 2015) (Denmark) and Żyła and colleagues (Poland) have done extensive work describing new families, genera and species of fossil aphids from Europe (e.g. Żyla and Wegierek 2020, Żyla *et al*. 2020).

THE PLANTS: HISTORY AND EARLY APHID ASSOCIATIONS

The earliest land plants were algae and evolved in the late Silurian, from about 420 mya. The late Devonian saw the development of ferns, seed ferns, horsetails and lycopods. Mosses are known from about 400 mya (Liu *et al.* 2019). McLoughlin (2001) and Kustatscher *et al.* (2018) summarised the flora of Gondwana in relation to geological and climatic events, pointing out that the basic flora, generally associated with lands derived from Gondwana, was present before the breakup of Pangaea, and is assumed to have become extinct in "Laurasia", following climatic extremes. Aphids occur on present-day mosses and horsetails, *Aphis equiseticola* being specific to its horsetail food plant. A range of species in the Northern Hemisphere feed on mosses (e.g. see Müller 1973, Pike *et al*. 2012). Some aphid species are specific to ferns: for example, Robinson (1966) listed seven indigenous and four adventive species in North

Table 1. Approximate chronological sequence of palaeogeography and life forms

Period/Epoch	Time span (Mya)	Geography	Plants	Aphids	Extinctions
Devonian	419-359	North and south China, Indochina split from Gondwana	Horsetails, mosses, seed ferns, ferns, lycopods, tree-like plants	(Earliest land arthropods)	Late Devonian
Carboniferous	359-299	Assembly of Pangaea		Insect diversification	
Permian	299-251	West Burma and Lhasa terranes split from Gondwana	*Glossopteris*, podocarps, early angiosperms	Earliest aphidomorph fossils	End Permian
Triassic	251-200	Split of Gondwana and Laurasia		Earliest aphid fossils	End Triassic
Jurassic	200-145	Separation of India-Africa-South America from Gondwana			
Cretaceous	145-65.5	India separates from Africa, Australia and Zealandia separate from Gondwana, Africa and S. America separate.		Many aphid fossils	End Cretaceous (K-T)
Tertiary/ Paleocene	65.5-55.8				
Tertiary/Eocene	55.8-34	India collides with Asia			
Tertiary/Oligocene	34-23				
Tertiary/Miocene	23-5.3		Radiation of northern angiosperms	Radiation of northern aphids. European fossils of Greenideinae	
Tertiary/Pliocene	5.3-2.4	Taiwan formed			
Quaternary	2.4-present				

America. The seed ferns are extinct. The only record of aphids on a lycopod is of the banana aphid, *Pentalonia nigronervosa* (Singh and Srivastava 2022). It seems unlikely that this is a regular host association.

High forest trees, including the extinct *Glossopteris*, and tree ferns, conifers, ginkgos and podocarps date from the early Permian, *ca* 299 mya, and have living descendants today. Blackman and Eastop, in *Aphids on the World's Plants*, did not record any aphid species on ginkgos. I have used this reference extensively without further attribution for notes on host-aphid relationships and aphid distributions, both online (Favret, 2025, https://aphidsonworldsplants.info/) and in the original books, *Aphids on the World's Crops*, *Aphids on the World's Trees*, and *Aphids on the World's Herbaceous Plants and Shrubs*, the latter typically abbreviated by Victor Eastop to *Aphids on the World's Whatsits*. I have used Colin Favret's *Aphid Species File* (https://aphid.speciesfile.org/) for questions of aphid taxonomic detail. I use the abbreviations B&E and ASF occasionally in the text for these resources, but I have relied heavily on B&E for information on aphid host-plant relationships and distribution, and ASF for nomenclature.

There are extant aphids on *Araucaria* (Araucariaceae) and *Podocarpus* (Podocarpaceae) as well as numerous northern conifers. The Araucariaceae is the oldest family of living conifers, with 13 species of *Araucaria* endemic to New Caledonia and others spread across the southern continents and introduced elsewhere. They are particularly favoured for beachfronts. *Neophyllaphis araucariae* lives on several species with a wide geographic spread including Mauritius (first description), Java, New Guinea, Australia, Hawaii, Costa Rica, Bahamas, Mexico and USA, but it was introduced to Hawaii and probably other North American sites. Surprisingly, it has not been recorded in New Caledonia despite the range of potential hosts (Mille *et al*. (2020). B&E give information for five species of *Araucaria* and *Neophyllaphis araucariae* occurs on all of them. Although *Araucaria* had a world-wide range in the Jurassic and Cretaceous, it became extinct in northern continents. However, it occurs as a cultivated plant in many countries, often taking its aphids with it. In Australia, *Neophyllaphis araucariae* occurs on Norfolk Island pine (*Araucaria heterophylla*, endemic to Norfolk Island but widely cultivated elsewhere including Australia) but there are no records from hoop pine (*A. cunninghamii*) or bunya pine (*A. bidwillii*), both of which are endemic to Australia.

In a sister group to *Araucaria*, *Wollemia* (wollemi pine, recently discovered in New South Wales) has not been reported to host aphids. In the same family is *Agathis* (kauri): *Neophyllaphis rappardi* has been found on *Agathis labillardieri* in West Papua (Indonesia) but surprisingly there are no records from Australia or New Zealand, both of which have kauri forests. The wollemi pine is sometimes described as "a living fossil" but equally, the araucarias and kauri are living fossils. Chen *et al.* (2022) provided a molecular analysis clarifying the relationships of these ancient tree genera.

For Podocarpaceae, 22 species of *Podocarpus* trees harbour 18 different *Neophyllaphis* spp., in Australia, New Zealand, Papua, China, Peru, Africa, Argentina, Brazil, Venezuela, Taiwan, Vietnam, Java, Malaysia. These (except the newcomer Taiwan) are Gondwanan countries. *Neophyllaphis* occurs in other countries where it has been introduced with its host. There is some overlap of aphid species between hosts. On four species of *Afrocarpus*, we have three species of *Neophyllaphis* overlapping with the *Podocarpus* list, *N. varicolor*, *N. viridis* and *N. grobleri*. *Neophyllaphis* also occurs in Japan, on *P. macrophylla.* Possibly both plant and aphid were introduced there. *P. angustifolia*, widespread in South America, is critically endangered by logging but is not reported to have any aphids. But there may be many more podocarp-feeding aphids. Khan *et al*. (2023) have listed 208 extant species of podocarps ranging through the Gondwanan countries. One genus, *Dacrydium* (rimu in New Zealand), is spread through the islands of Zealandia, New Guinea, Indonesia and Southeast Asia, but is extinct in Australia. No aphids are known from *Dacrydium*, but it seems a likely and accessible host.

Based on a time-calibrated molecular phylogeny, Salomo *et al.* (2017) argued that the angiosperms probably arose in the Late Permian, about 275 mya, rather than the later estimate of around 130 mya suggested by fossils alone. Hence, we have a well-developed flora of both angiosperms and gymnosperms before the separation of Pangaea, and thus also before the breakup of Gondwana. And (see above) we have an aphidomorph fossil history in Europe that is older than expected, i.e., preceding the breakup of Pangaea.

Prominent among the Gondwanan angiosperms, the southern beech (*Nothofagus*, Nothofagaceae) occurs in Australia, New Zealand, New Guinea, South America and New Caledonia. Steed-Mundin (2025) has given an overview of the genus, and discusses a "disputed split" proposed by Heenan and Smissen (2013) who raised four well-defined subgenera to generic status. In this paper I will follow Steed-Mundin's example and stick to the original generic name. 36 species are known: 12 from New Guinea, 11 from South America, five from New Caledonia, five from NZ and three from Australia. The New Caledonia and New Guinea species are all in subgenus *Brassospora*; the subgenus *Nothofagus* includes only South American species, and the other two subgenera, *Lophozonia* and *Fuscospora*, each occur in Australia, New Zealand and South America. *Nothofagus* spp. are host to *Taiwanaphis/Sensoriaphis* (Taiwanaphidinae) in South America, Australia, New Guinea, and New Zealand. Three species of *Taiwanaphis* on diverse host plants (not *Nothofagus*) are listed in India by Singh *et al.* (2018). These three species are supported in B&E but not listed in ASF. *Neuquenaphis* spp. (Spicaphidinae) are widespread on *Nothofagus* spp. in South America, but only one species in the Taiwanaphidinae is found there. *Nothofagus* does not occur in India or Africa and there is no fossil evidence (including pollen) for its past occurrence. No aphids have yet been recorded from *N. gunnii* (Tasmania). Plant scientist David Tng (2012) has written a thoughtful account of *Nothofagus*.

The other host plants of presumed southern endemic aphids are dicotyledonous shrubs and herbs, with the possibility of some on native grasses. The hosts (other than podocarps and *Nothofagus*) include small trees, shrubs and herbaceous plants. In New Zealand, *Aristotelia* (Elaeocarpaceae), *Plagianthus* (Malvaceae), *Coprosma* (Rubiaceae), *Hebe* (Plantaginaceae), *Olearia* and *Ozothamnus* (Asteraceae), *Carmichaelia* (Fabaceae, New Zealand broom) and the native climbers *Muelenbeckia* (Polygonaceae) and *Clematis* (Ranunculaceae)are all known to host aphids.

The smaller aphid-bearing plants include *Platylobium* and *Gompholobium* (Fabaceae, yellow bush peas, Australia), low herbaceous plants like *Epilobium* (Onagraceae) (Australia and New Zealand) and *Acaena* (Rosaceae) (Australia and South America). *Aciphylla* (Apiaceae) is a hard leaved rosette plant producing a long flower spike, in Australia, New Zealand and Antarctic islands, but aphids are known on it only in New Zealand. *Dracophyllum* (62 spp., Ericaceae) has a distribution in Australia and the elements of Zealandia but aphids are known from it only in New Zealand. See Table 2 for the Australian aphids and their host plants.

THE APHIDS

Aphids are not ordinary insects. Not for them is the simple life cycle of other hemimetabolous insects: egg, larva (eating and growth), adult (fly around, mate, lay eggs). The complexity of their annual cycles and the terminology of different forms often deters non-specialists from reading or thinking about them, but aspects of their biology form essential components of their ability to colonise new territories.

1. Aphids are parthenogenetic and viviparous, most of the time. They are born with embryos already developing inside them, so can rapidly build up new colonies.
2. The parthenogenetic aphids come in two forms, wingless and winged. The winged ones develop in response to environmental cues, especially crowding. It is an epigenetic process triggered by environment and modified via neural, neurohormonal and hormonal and epigenetic pathways, and enables dispersal.
3. Aphid species may be restricted to a narrow set of host plants (e.g., a single species, genus or family), or they may be able to use a wide range of plants ("polyphagy").
4. Aphids have a sexual generation, usually invoked by thresholds of photoperiod, or more accurately, scotoperiod (dark phase). A consequent epigenetic change causes production of males and females (gynoparae) whose parthenogenetically-produced female offspring will be capable of mating and laying eggs.
5. Many aphid species include clones that are continuously parthenogenetic, i.e. do not and cannot produce sexual forms.
6. The sexual females may occur on the regular host plant/s or may be deposited on a special winter host, e.g. the peach aphid *Myzus persicae*, found on a wide range of plants in summer, but returning to peach to produce overwintering eggs. This is known as host alternation. Usually the sexual females are wingless and the males are winged, but not always. Non-native aphids in Australia, including *M. persicae*, have clones that skip the overwintering process and reproduce parthenogenetically throughout the year.
7. The sexual females mate with males and lay eggs that diapause, avoiding adverse seasons, winter or sometimes summer.

Moran (1988), von Dohlen and Moran (2000) and others have discussed the evolution of host plant alternation in aphids. Hales *et al*. (1997) reviewed evolutionary and genetic aspects of aphid biology including migration and annual cycles.

These unusual biological features can be both advantageous and limiting when considering the ability of aphids to expand their geographic range. Polyphagy is an asset for expansion of range, and diapausing eggs can get a species through extremely cold winters or hot summers. Both strategies can give flexibility to a species. Host alternation has a downside, as not one, but two essential host plants must be found for a coloniser to succeed. Sexual reproduction, of course, also gives the opportunity for genetic recombination, again providing flexibility and potential new opportunities. Genetic change by mutation in parthenogenetic lines also gives opportunities for adapting to new environments and potentially new host plants (e.g. Wilson *et al.*, 2003). Alien aphids can establish on sub-Antarctic islands, where polyphagy and parthenogenesis have been recognised as enabling factors (Hullé *et al*. 2003).
An essential consideration in studying aphid distribution is dispersal. Aphids are small, fragile, soft-bodied insects. Yet winged aphids are capable of wind-assisted flights of over 1000 kilometres. In the 1950s Bruce Johnson, later professor of zoology at the University of Tasmania, worked on *Aphis craccivora* (cowpea aphid) and its migration on high altitude winds from crops in north-western NSW (Coonamble, Walgett, Moree) to the east coast (Johnson, 1957). In August 1966, on a trip from Adelaide to Alice Springs, I observed *A. craccivora* on desert chenopods such as saltbush and bluebush at nearly every stopping point, raising the possibility of even longer flights to the east coast. Few other aphids were seen (White, 1967). European and North American reports were summarised by Robert (1987) including dispersal over 1000 km. Hill *et al*. (2020) proposed that the giant willow aphid *Tuberolachnus salignus* arrived in mainland Australia and Tasmania from infestations in New Zealand, a distance around 1500 km. With these capabilities, aphids would laugh at Wallace's Line, the distance from China to Taiwan, and the distance from Australia to the closest historically exposed parts of Zealandia. Successful establishment at the destination is another matter and depends on ability to find a suitable host plant in the limited time available. Aphids survive only a day or so without feeding, but a single successful female migrant can start a new population, because of the parthenogenetic mode of reproduction.

Aphids contain populations of a symbiotic bacterium, *Buchnera aphidicola*, in specialised cells known as bacteriocytes (= mycetocytes in older literature). The bacteria are essential to the aphid's survival, as they are capable of synthesising amino acids lacking in the aphid's diet of sugar-rich, but nitrogen-poor, phloem sap. The bacteria are passed from mother to embryo (or egg) during development. Older work on bacteriocyte and bacterial structure was summarised by Houk (1987), though unfortunately he does not mention work by Hinde at Sydney University (Hinde, 1971, a, b c.). See Wilson *et al*. (2010), Pers and Hansen (2021) and Hansen and Moran (2011) and references therein for more recent detailed accounts. The relationship of aphids and *Buchnera* dates back to the earliest days of aphids, and bacterial molecular markers can throw light on aphid phylogeny e.g., Martinez-Torres *et al*. (2001). There is extensive literature on the molecular, biochemical and physiological aspects of *Buchnera* and its interaction with its aphid host. One interesting feature is its ability to synthesise Vitamin B5 (pantothenic acid. The holidic[2] aphid diet developed by Dadd and Mittler in the early 1960s (see Dadd and Mittler, 1966) enabled studies of essential nutrients for aphids, and further developments using aposymbiotic aphids (i.e. those with bacterial symbionts killed by antibiotics) allowed recognition of nutrients supplied by the bacteria. Initially it was supposed that the bacteria provided aphids with sterols, as insects cannot synthesise them, but aphids feeding on plants can modify plant sterols to produce their sterol requirements (e.g. the moulting hormone ecdysone is a steroid). On defined artificial diets lacking any sterols, however, only a limited number of generations survive, suggesting a possible role for *Buchnera*, although it is known that most bacteria do not synthesise sterols. Aposymbiotic aphids on sterol-free diets were not able to reproduce (Douglas, 1988), so the role of the symbionts in sterol production is supported.

Sometimes aphids contain additional bacterial symbionts. One is *Hamiltonia*, which appears to deter parasitic wasps from attacking its host aphids (Rothacher *et al.* 2016). Others may support recovery after heat stress of both *Buchnera* and its host aphid (Heyworth *et al*. 2020).

Some aphids have mutualistic relationships with ants. Most of these associations are facultative but a few are obligate (reviewed by Stadler and Dixon, 2005). Unidentified ants were observed forming a cover of vegetation fragments over colonies of the endemic Australian *Aphis acaenovinae* on its ground-hugging host, *Acaena ovina*, in the

[2] A holidic diet is one prepared from defined chemicals, e.g. measured amounts of specific amino acids, sucrose, vitamins, minerals, water. Aphids feed on the diet through a stretched Parafilm membrane.

Table 2. Australian endemic aphids with host plants and distribution

Subfamily	**Genus**	**Species**	**Plant**	
Neophyllaphidinae				
	Neophyllaphis	*araucariae*	*Araucaria heterophylla*	(see text)
		brimblecombei	*Podocarpus elatus*	NSW, QLD
		lanata	*P. spinulosus*	NSW, QLD
		gingerensis	*P. lawrencei*	ACT, NSW
Taiwanaphidinae	*Taiwanaphis*	*tasmaniae*	*Nothofagus cunninghamii*	TAS
		furcifera	*Nothofagus moorei*	NSW
		melaleucica	*Melaleuca lanceolata* (Myrtaceae)	WA
Greenideinae	*Meringosphon*	*paradisicum*	*Gastrolobium dilatatum* (Fabaceae)	WA
	*Anomalaphis**	*casimiri*	*Leptospermum scopiarum* (Myrtaceae)	ACT
		comperei	*Agonis flexuosa (Myrtaceae*	WA
Aphidinae	*Aphis*	*acaenovinae*	*Acaena anserovina* *Geum* (both Rosaceae)	NSW
	Aphis	*carverae*	*Epilobium labillardiereanum* (Onagraceae)	NSW
	Aphis	*platylobii*	*Platylobium formosum* NSW, *Daviesia mimosoides* ACT (Fabaceae)	NSW and ACT
	Casimira	*canberrae*	*Epilobium junceum***	NSW and ACT
Lizeriinae	*Ceriferella*	*leucopogonis*	Leucopogon	NSW
		dossuaria	"native flowers"	WA

* Additional species known but not described

** Very widespread, similar to *E. billierardereanum*.

Brindabellas, but the relationship was not obligate as colonies without ants were common (Hales 2008). Aphids have various general predators such as coccinellid, neuropteran, chamaemyiid and syrphid larvae, and coccinellid and neuropteran adults, and any of these can extinguish local populations.

Northern ladybirds have been introduced from time to time as potential biological control agents to Australia and New Zealand, for example *Adalia bipunctata*, on several occasions dating back to 1895. Its first intended target was the cabbage aphid, a particularly unattractive meal as it has a dense coat of wax filaments and smells like cabbage. It has not been successful (Ślipiński *et al*. 2020). The exotic ladybird *Hippodamia variegata* arrived in Australia seemingly unassisted some 25 years ago (Franzmann, 2002). It is known to prey on pest aphids but could feasibly also attack endemic ones. More insidious, and more successful in biological control, are endoparasitic wasps in the Aphidiinae and Aphelinidae. Some cecidomyiid midges also parasitise aphids. As well as the primary parasites, there are hyperparasites.

More details of life histories of the parasites will be discussed in a later section, as will the proposition that some of these groups have a southern background.

Adventive aphids can cause a definite threat to native aphids, e.g., *Aphis oenotherae* that competes with *Casimira canberrae* and other endemic aphids feeding on *Epilobium*.

ENDEMIC AND NATIVE APHIDS OF AUSTRALIA

Autobiographically, this should be the beginning of the story. In 1965 I had started my PhD on the physiology of polymorphism in aphids, with the cabbage aphid as the experimental animal. I was sharing a flat with friends and we wanted to augment our basic cookery with some herbs, so had small pots of mint and parsley on our 3rd floor windowsill. The parsley got aphids on it. The mint got aphids on it. They were different aphids! So began my interest in aphid diversity, supported initially by Cottier's 1953 book on the aphids of New Zealand.

Eastop's comprehensive work on the Australian Aphidoidea (Eastop 1966) recognised 119 species altogether, 51 genera with 101 species being in the Aphididae as then defined. Eleven species out of the total, all in Aphididae, were noted as native (=endemic). The endemics included two species of *Neophyllaphis*, one *Taiwanaphis*[3] two *Ceriferella*, one *Meringosiphon*, one described and two undescribed *Anomalaphis*, *Aphis acaenovinae*, and *Casimira canberrae*. *Neophyllaphis podocarpi*, known from Asia, was included in the list, but the Australian version has since been described as a separate endemic species *N. brimblecombei*. *Ceriferella* is now classified in the subfamily Lizeriinae, possibly the most ancient subfamily of the Aphididae, and one with an exclusively Gondwanan distribution (see below). The Lizeriinae includes just three genera. *Paoliella* has about 22 species distributed across Africa and one in India and one in Brazil. *Ceriferella* has two species in Australia, *C. dossuaria* and *C. leucopogonis*, both described by Carver and Martyn in 1965. The genus has not been found elsewhere. The third genus, *Lizerius*, has eight species and is confined to South America and the Caribbean.

Mary Carver (pers. comm., 1998) listed 171 species of Australian Aphidoidea including four undescribed *Anomalaphis* species which are probably endemic, and an undescribed *Taiwanaphis* from Western Australia, since described by Quednau (2010) as *T. melaleucica*. Brumley (2020) updated Eastop's list in work based on the aphids in the Australian National Insect Collection plus 24 known species not in the collection, with endemics *Aphis carverae*, *A. platylobii*[4], *Neophyllaphis lanata* and an additional species of *Taiwanaphis* (as *Sensoriaphis*, Carver and Hales, 1974) and of *Anomalaphis*. Brumley (2020) also provided identifications of some specimens in the collection, recognising non-native species and genera not previously recorded here. So the number of recognised Australian endemic aphids has almost doubled since 1966. A species of *Sitobion* on *Smilax* spp. in Australia was listed by Hollis and Eastop (2006) as *S. smilacifoliae*, but morphometric studies did not support this (Hales *et al*. 2010), leading B&E to suspect that it was a new species. *Sitobion* has high numbers of probably endemic species in Africa and India (B&E).

The number of exotic arrivals - at least 18 since Carver's checklist - has easily outstripped the number of newly recognised endemics. Two recent newly-found exotics were *Megoura crassicauda*

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[3] It is ironic that the aphids on *Nothofagus* in Australia and neighbouring countries are now named for an island that did not exist when they evolved.

[4] Hille Ris Lambers (*in litt.*) called it "a horrible aphid" because of its differences from northern *Aphis* spp.

and *Sarucallis kahawaluokalani* from the author's northern Sydney garden.

The Australian endemic aphids are shown with their host plants and other data in Table 2. Some species not listed can be considered naturally occurring in Australia ("native" or "indigenous") by way of a Gondwanan linkage via Southeast Asia, particularly Indonesia. These include *Aphis clerodendri*, *Aphis eugeniae*, and Greeneidinae (*Greenidea*, *Schoutedenia*).

Of these, *Aphis clerodendri* is present through east Asia, New Guinea and Australia on *Clerodendrum* (Lamiaceae), a genus found in Africa, South America, India, East and South-East Asia, New Guinea and Australia (but not New Zealand). (https://powo.science.kew.org/taxon/urn:lsid:ipni.org:names:30002447-2). On the basis of distribution, it is tempting to think of *Aphis clerodendri* as a Gondwanan aphid but it is very similar in morphology to the world-wide highly polyphagous *Aphis gossypii*. Polyphagy leads to easy natural and human-assisted invasion of new areas. *A. gossypii* is said by Blackman and Eastop to be "particularly abundant and well-distributed in the tropics". A southern origin for *A. clerodendri* is probable but more work on *A.gossypii* would be needed before claiming southern origin.

Aphis eugeniae (= *A. hardyi* in Eastop,1966) was found by the author in Sydney on cheese tree (*Glochidion ferdinandii*: Phyllanthaceae). It has other hosts, often in the Euphorbiaceae, and is widely distributed across Asia, including India, east and south-east Asia and Australia, New Guinea and New Caledonia. Like *A. clerodendri*, it could make a hopeful claim to be Gondwanan.

A better claim can be made for the two subfamilies Greenideinae and Lizeriinae. Liu *et al*. (2014) provided an interesting molecular phylogeny showing the relationships among Greenideinae and to other aphid subfamilies, essentially supporting the division of Greenideinae into three tribes, Greenideini, Cervaphidini and Schoutedeniini.

The Greenideini includes *Greenidea* (61 spp. with wide Asian distribution, especially India and east and Southeast Asia, one sp. in Australia), *Allotrichosiphum* (4 spp. from India, South China and Japan), *Eutrichosiphum* (54 spp. mainly India, east Asia including Japan, Indonesia), *Greenideoida* (12 spp., east and southeast Asia), *Mesotrichosiphum* (3 spp., East China, Philippines, Indonesia), *Mollitrichosiphum* (18 spp., India, China, Taiwan, Japan, Indonesia), and *Tritrichosiphum* (1 spp., described from Thailand, listed in *Aphid Species File* but not *Aphids on the World's Plants*). Gao *et al.* (2017) discussed species richness and endemism of the Greenideinae, recognising 192 species/subspecies and found the greatest endemicity was correlated with concentrations of taxa. The distribution has a Gondwanan nature - Himalayas, Southern China, Malay Peninsula and Java. Australia has *G. ficicola* on native figs such as Port Jackson fig *Ficus rubiginosa* (noted in Eastop 1966 as *Ficus hillyi*, a misspelling of *Ficus hillii*).

Cervaphidini includes five extant genera, *Anomalaphis, Brasilaphis, Cervaphis*, *Meringosiphon* and *Sumatraphis* plus the fossil *Quisqueyaaphis heiei* from the Caribbean in Dominican amber (Wegierek, 2001). *Anomalaphis* consists of two species limited to Australia. Carver (pers. comm. 1998) knew of 4 undescribed species, all from Australia. *Brasilaphis* consists of a single species, unsurprisingly in Brazil. *Cervaphis* contains five species mainly in Southeast Asia with *Cervaphis rappardi* reported from Papua New Guinea (PNG) (Lamb,1971). *Meringosiphon* is monotypic and known only from Western Australia. The hostplant, not known at the time of description, has been confirmed as *Gompholobium dilatatum* (Fabaceae). *Sumatraphis* is also monotypic, and known from Indonesia, India, Nepal and Yunnan province in Southern China (B&E, ASF).

The Schoutedeniini contains three genera. *Schoutedenia* has two species, *Schoutedenia emblica* from India, Pakistan, Nepal, China, and Thailand, and *S. ralumensis*, recently revised to include a range of species including *S. lutea*, which is common in eastern Australia on *Breynia oblongifolia* (Euphorbiaceae). *S. ralumensis* is widespread in Southeast Asia, India, and Africa. *Eonaphis* has four species, all in Africa, while *Paulianaphis* is monotypic with a single species in Madagascar. A Gondwanan origin is likely, and was recognised by Remaudière (1988): "Les Schoutedeniini ont une distribution typiquement Gondwanienne." Perkovsky and Wegierek (2018) argued that thermal sensitivity of the bacterial symbiont *Buchnera* restricted aphids, following the northern Tertiary expansion, from crossing the tropics. They took the Greenideinae as an example of the few groups to have made the crossing, but may not have considered their likely southern origin or the extent of Gondwanan fragments in Asia. However, the resolution of Greenideinae origins is equivocal because there are several known European fossils (Wegierek and Peñalva 2002).

The subfamily Lachninae is generally associated with Pinales, another group with early Pangaean ancestors. Eastop (1966) listed six species in Australia, but considered them to be of European or North American origin. There are no known endemic or native species in Australia. The subfamily appears to be an ancient lineage of the Aphididae, supported with molecular studies by von Dohlen and Moran (2000), Martinez-Torres *et al*. (2001), Ortiz-Rivas, Martinez-Torres (2010) and Rebijith *et al*. (2017) using mitochondrial and nuclear genes, as well as Owen and Miller (2022) in a phylogenomic study, although there is variability among the topologies. Heie (2015), however, questioned these conclusions, proposing possible evolutionary mechanisms compatible with his cladistic determinations suggesting that the family is relatively young. None of the analyses using genes of the obligate bacterial endosymbiont *Buchnera aphidicola* found Lachninae to branch near the base of the tree (Nováková *et al*., 2013). Lachninae has no endemic or native species in Australia or New Zealand. Several are present in India. The family thrives in the conifer forests of North America and Europe and several species, some of aboricultural significance, have been accidentally introduced to Australia. It seems our *Callitris* (Cupressaceae) woodlands support no endemic aphids. Lachnines almost certainly evolved during Pangaean times and either inhabited only the sections of the supercontinent that became Laurasia and India, or were generally dispersed throughout Pangaea but became extinct in the rest of the Gondwanan section.

Jacksonia papillata (Aphidinae: Macrosiphini) is known from Macquarie Island but is widespread in the Palaearctic and found particularly on etiolated parts of plants especially grasses (Eastop 1966), but also on mosses (Müller 1973). Despite being found in an Australian Antarctic territory it can be presumed to be an accidental introduction.

In New Zealand, possible additions to the list have been found in native grass turfs, an environment not so far studied in Australia. New Zealand endemic aphids are reported in detail by Teulon *et al*. (2013) and are summarised in the next section.

Singh and Srivastava (2022) listed aphids on various ancient plants in India, including horsetails, ferns and podocarps, but their data seem to include accidental presence on plants and throw no light on the relationship of aphids to plants of early or southern origins. For example, *Idiopterus* on horsetails and *Myzus ornatus* on *Podocarpus* are unlikely relationships. *Cinara atrotibialis* on an unidentified *Araucaria* is more likely but more information is needed.

ENDEMIC AND NATIVE APHIDS OF NEW ZEALAND

Work on the New Zealand endemic aphids has been more comprehensive than that in Australia and at least 15 endemic species are now known out of a total fauna of 135. The endemics are mostly in the Aphidinae, but there are two species of *Neophyllaphis* and one of *Taiwanaphis*. The aphidine species are each specific to particular endemic plants.

Cottier (1953) recognised two species of *Neophyllaphis* (*N. araucariae* and *N. totarae*), *Aphis coprosmae*, *Aphis healyi*, *Aphis nelsonensis*, *Sensoriaphis nothofagi* (*gen. et sp. nov.*) feeding on *Nothofagus truncata* on Little Barrier Island, and *Thripsaphis foxtonensis* from *Carex*. *Aphis nelsonensis* on *Epilobium* may now be rare following introduction of *Aphis oenotherae*, as with *Casimira canberrae* in Australia (Hales *et al*. 2014).

More recently, Teulon and colleagues have written extensively on endemic aphids in New Zealand, e.g. von Dohlen and Teulon (2003), who used molecular phylogeny to establish the early divergence of the NZ aphidines, and Teulon *et al.* (2013) who gave a detailed account of endemic aphids and their biology in NZ. They generally use the term "native aphids", but the species listed are most likely endemic. Their list (see Table 1 in Teulon *et al*. 2013) includes undetermined/undescribed species (*Aphis* on *Hebe*, *Aphis* on *Olearia*, *Aphis* on *Samolus*, *Aphis* on *Clematis*, *Casimira* on *Ozothamnus*, *Schizaphis* on *Aciphylla*, *Schizaphis* on *Dracophyllum*, *Melanaphis* on ?grass, three undescribed *Rhopalosiphum* from grass and cereals. Podmore *et al*. (2019) investigated the genetics of undescribed *Schizaphis* species using mitochondrial markers which supported the distinction between the *Aciphylla* and *Dracophyllum* populations and showed the presence of potentially new species. Teulon (2021) gave details of the biology of *Aphis healyi* and an undetermined *Schizaphis*.

Other species in New Zealand are well-known and described in Teulon *et al.* (2013). They include *Paradoxaphis arisoteliae* and *P. plagianthi*, and *Aphis cottieri* on *Muehlenbeckia* in Aphidini, with *Megoura stufkensi* on Fabaceae being the only apparently endemic member of the Macrosiphini (but see *Sitobion* below.) Specimens of *Carmichaelia* (aphid host plant in NZ) found on Lord Howe Island were noted as probably accidentally transported, but recognition of past land connections presents another interpretation.

Taiwanaphis (*Sensoriaphis*) *nothofagi* is listed on three species of *Nothofagus*. *Neophyllaphis totarae* is listed from four species of *Podocarpus*, and another undescribed *Neophyllaphis* from *P. nivalis* is known in the South Island. This one (collected by author on 4 February 1972 at Arthur's Pass, about 3000 ft altitude) was to have been described by Mary Carver and named *N. sinzi*, following tourist pamphlets advertising the South Island as SINZ. The material consisted of one aptera vivipara, one male, and one ovipara, which was not enough for a full description (White, collection notes). They were scattered on young shoots, not part of a colony. Some Australian *Neophyllaphis* on *Podocarpus* diapause over summer as eggs, (Hales 1976, Hales and Lardner 1988), so the discovery of parthenogenetic and sexual morphs in February is a point of interest. I also collected a *Sitobion* (Aphidinae: Macrosiphini) from *Rhipogonum* (Smilacaceae) at Lake Mapourika on 7 February 1972, which may fall into the same category as the Australian one from *Smilax* - probably native if not endemic.

ENDEMIC AND NATIVE APHIDS OF NEW GUINEA.

Lamb (1971) published a checklist of the 57 aphids known at that time from PNG, based on Moericke yellow trap samples from several contrasting localities (*ca* 6000 specimens) and others in collections, including the British Museum (Natural History). It included cosmopolitan aphids like *Aphis gossypii*, and widespread aphids that were not known in Australia until later such as *A. glycines* and *Hysteroneura setariae*. It further included Asian genera still not known in Australia (*Indomegoura* widespread in Asia, *Macromyzus* which has 5 species on ferns and is widespread in Asia. *Anomalosiphum pithicolobii* and *Cervaphis rappardii* (Cervaphidini) were listed and also occur in Asia. *Greenidea*, discussed above, has species in PNG that have not reached Australia. Lamb (1974) went on to compare the aphid fauna across various countries using numerical analysis and concluded that the PNG aphid fauna was most similar to that of the Philippines and not closely related to the Australian fauna. Probably several of the species listed by Lamb could be regarded as "native", having a continuous distribution from PNG through the Gondwanan countries of India and East and Southeast Asia.

Taiwanaphis niuginii on *Nothofagus carrii* is an endemic addition to the fauna (Carver, 1978, as *Sensoriaphis*). There may be others.

ENDEMIC AND NATIVE APHIDS OF SOUTH AMERICA

The continent of South America consists of multiple countries. There are many species of endemic *Neophyllaphis* on *Podocarpus*. One on *Araucaria* is widespread in Gondwanan countries. *Neuquenaphis* is a genus endemic to South America on *Nothofagus*, and some species may be threatened by incursions of human populations and logging (Figeroa *et al*. 2017). See earlier account for these ancient genera.

Fuentes-Contreras *et al*. (1997) listed 31 native species from Chile alone: of these, four were *Aphis* spp., 10 *Uroleucon* and 12 *Neuquenaphis*. Nieto Nafría, Mier Durante, Ortego and colleagues have written numerous papers on newly-discovered South American aphids, particularly in Chile and Argentina - too many to reference individually, but easily found. Taking a single genus as a proxy for the South American aphid fauna, I went through the genus *Aphis* in Blackman and Eastop's *Aphids on the World's Plants*, and found over 50 recently described species of *Aphis* presumably endemic to South America. Ortego *et al*. (2021) raised the number just of *Aphis* endemic to South America to 56. As in Australia and New Zealand, *Aphis* and related genera are significant among the endemic aphid fauna. Like Australia, South America has an *Aphis* on *Acaena*, but it is not closely related to Australia's *A. acaenovinae* (Mier Durante and Ortego, 1998). South America has not only large numbers of endemic *Aphis* spp., but also endemic genera such as *Delfinoia* (Nieto Nafría *et al*. 2017). Unlike Australia, South America has at least one macrosiphine genus, *Uroleucon*, with 25+ endemic species. *Uroleucon* is otherwise widespread in Europe, Asia, and North America. Two species have recently arrived in Australia. The genus in South America was reviewed by de Carvahlo *et al.* (1998), who described four new species.

It has long been a question for aphidologists "Why are there so few aphids in the tropics"? Dixon, Kindlmann *et al*. (1987) suggested that it might be a question of plant apparency, i.e. the flora is so complex that host-specific insects have (or would have) trouble finding their host-plants. This is especially so if there is a dense canopy obscuring the understory. Like Australia, South America has both rain forest and desert regions which allow few opportunities for aphids. The further question, posed by Heie (1994), is "Why are there so few aphids in temperate areas of the southern hemisphere?" There are more than he thought, and without doubt there are more undiscovered, particularly in South America and Africa.

However, the native vegetation in much of the non-desert area of Australia is dominated by *Eucalyptus* and *Acacia*, neither of which support populations of aphids, although ephemeral colonies of polyphagous non-native aphids are occasionally found on them. Further, agricultural and grazing land are unlikely to support a diverse range of aphids.

In contrast to the aphids, other members of the Sternorrhyncha and of the Auchenorrhyncha are well-adapted to eucalypts and acacias and have radiated extensively in Australia, with some tribes endemic to Australasia. The moss bugs (Peloridiidae) are recognised as a Gondwanan group (Fletcher, pers.comm., see Burckhardt 2009, Ye *et al*. 2019).

ENDEMIC AND NATIVE APHIDS OF AFRICA

The continent of Africa consists of about 48 separate countries, and contains extensive deserts and extensive rain forests. Madagascar, Mauritius and smaller island nations have palaeogeological links to Africa. Eastop, early in his career, wrote about the aphids of East Africa and of West Africa but unfortunately these works are hard to come by. Theobald (1914) reported only 35 species.

I have not been able to get any overall idea of African aphid endemicity. Known endemic aphids include two species of *Protaphis*, a genus with strong diversity in Eastern Europe/ Western Asia, and Gondwanan genera *Paoliella* (Lizeriinae), and *Eonaphis* (Greenideinae) with four species. It was suggested (von Dohlen, pers. comm.) that *Sitobion* might be a guide to endemicity, with 24 species known so far only from Africa (B&E). Another group of *Sitobion* species seems to be indigenous to India and there are others in Southeast Asia and South America. There are probably other undescribed species in Australia and New Zealand, as well. It seems likely to be a Gondwanan genus. Nieto Nafría *et al.* (2024) with their deep knowledge of South American species reached the same conclusion.

ENDEMIC AND NATIVE APHIDS OF INDIA

The *Aphis* as a proxy approach was less successful with the Indian fauna. Only two species fitted the criteria for endemicity (not known from other countries). *Casimira bhutanensis* Ghosh, Basu & Raychaudhuri was described from India in 1971, with the only other described member of the genus being endemic to Australia. Ghosh and Ghosh (2006) reported on the Aphidini of India (not seen). As in South America, there are many apparently endemic members of the genus *Sitobion* in India. Ghosh and Singh (2000) reported on distribution and endemism of insects across India. Later, Singh and Singh (2019) stated that "In India, 794 species of aphids under 208 genera are reported out of which about 385 are endemic". They did not give a list, and did not clearly cite the sources of the figures in their tables.

Indian aphid taxonomists have been very active and if this count is even approximately correct, there is a very high degree of endemism, possibly related to speciation of a Gondwanan fauna during the millions of years between India's departure from Gondwana to the Northern Hemisphere, and its collision with the Asian plate, the south side of the Himalayas being geologically of earlier Gondwanan origin (see Chatterjee *et al.* 2006, for a palaeoclimatic and geological description).

APHID PARASITOIDS AND HYPERPARASITOIDS

Two taxa of small wasps, Braconidae: Aphidiinae and Aphelinidae, contain obligate parasitoids of aphids. The wasp's egg is laid into the aphid and the larva develops and eventually pupates within the host, whose cuticle becomes swollen and hardened. This is known as a "mummy", typically a beige colour if the wasp is an aphidiine and black if it is an aphelinid. Ferrar-Suay *et al*. (2025) recently reviewed the biology and diversity of aphid parasitoids.

Wasps of both groups have a degree of specificity and have been imported for biological control, e.g. *Aphidius ervi* for control of the (introduced) blue-green lucerne aphid *Acyrthosiphon kondoi* in Australia (Milne 1986). Importation for biological control has a risk to the native aphid fauna. Bulman *et al.* (2021) found evidence that aphidiine wasps introduced for biological control did not parasitise native aphids in NZ, but that endemic aphids were parasitised by a suite of endemic aphidiines. In Chile, Tomanović *et al.* (2023) recently described four new endemic South American species of the aphidiine *Pseudephedrus* parasitising *Neuquenaphis* species on *Nothofagus*. And in Australia, a new genus *Parephedrus* was erected containing a species of aphidiine parasitising *Taiwanaphis* (*Sensoriaphis*) on *Nothofagus* in New South Wales (Starý and Carver 1971).

Belshaw *et al*. (2000) conducted phylogenetic molecular studies and concluded that a southern origin was likely for the aphidiines. This agrees with conclusions by others including Stary and Carver (1971), and Schlinger (1974), but has been questioned more recently by Ortego-Blanca *et al.* (2009) on the basis of the morphology of a fossil in amber, found in Spain. Petrović (2022) wrote about the taxonomic impediments to aphidiine studies, which included lack of taxonomists and their uneven geographic spread, and he decried the tendency to rush to molecular methods including bar-coding: "(studying) *Aphidius colemani* group in Eastern Africa and the native Aphidiinae of New Zealand used destructive DNA extraction protocols and lost potentially very valuable information, while those from New Zealand probably also lost several as yet undescribed species".

Less common internal parasitoids are midges in the family Cecidomyiidae (Diptera). The aphid *Schoutedenia ralumensis*[5] is parasitised in NSW by an undescribed gall midge in the genus *Pseudendaphis*[6] (identified by K.M. Harris, pers.comm.) The midge lays its eggs near an aphid colony and the hatching larva enters the young aphid via unsclerotised parts of the cuticle. The larva grows within the body of the aphid and exits via the anus, pupating in the soil (Hales and Carver, 1976). We found that the earliest stage parasitised was the third instar, and that the parasite avoided major organs but destroyed the fat body and embryos (Lardner and Hales (1990). In contrast, Kirkpatrick (2009) found that all instars of *Toxoptera aurantii* were parasitised by *P. maculans* in Trinidad. *Endaphis* is widespread in northern continents.

The wasp larvae and midge larvae can in turn be parasitised by other wasps ("hyperparasitoids"). The gall midge in *Schoutedenia* is parasitised by a polyembryonic wasp, *Tricacis* (Platygastrinae, identification by E. Riek, pers. comm.) that lays its egg in the brain of the developing midge embryo before hatching! In mounted late instar or adult aphids, 2-4 cyclopoid larvae of the hyperparasite can sometimes be seen in the brain of the parasite (Hales and Carver, 1976, Hales unpublished).

Sullivan (1987) mentioned cases where aphids had tertiary parasites, generally hyperparasites attacking each other. Sullivan and Völkl (1999) reviewed the hyperparasitoids of aphids, listing ten different genera in three different superfamilies, but do not give biogeographic information. They indicated that the majority are host specific, either to the aphid species or the primary parasite species. The major hyperparasite of aphids in Australia is *Alloxysta* (Cynipidae: Charipinae). Carver (1992) listed five species occurring here. Ward *et al.* (2021) gave an account of the hymenopterous parasites and hyperparasites in aphids on grain crops in Australia. *Alloxysta* is widespread in northern continents. It is not clear whether there is a Gondwanan history, but Ferrer-Suay *et al*. (2012) listed all the Charipinae by current distribution. By far the majority were northern species, but twelve species were considered Australasian, with another 28 species in the Neotropics, Afrotropics and Oriental regions. These all have more or less clear Gondwanan connections and the host specificity suggests an ancient link with southern aphids and their aphidiine parasites and their hyperparasites. Just as aphids showed rapid radiation in the northern Tertiary, it is likely that the wasps did too.

This story is not about southern aphids but is too good to leave out. The parasitoid wasp *Protaphidius nawaii* parasitises the lachnine *Stomaphis japonica*. The aphid colonies are attended by two subgenera of ants in the genus *Lasius*. The ants of the two subgenera attend different colonies of the aphid. Molecular analysis showed that genetically different strains of the wasp parasite attacked aphids depending on which ant sub-genus was attending them! The ants had different behaviours around the aphids, and this may have caused selective pressure resulting in divergence within the parasite species (Yamamoto *et al*., 2020).

Schlinger (1974) reviewed biological evidence for "continental drift" using the multi-trophic relationship of plant (*Nothofagus*), aphid (*Neuquenaphis* and *Sensoriaphis*[7]) parasitoid and hyperparasitoid (a species of *Alloxysta* that appeared to be host specific) and considered that the associations were too intricate and strong to have arisen in South America by any method other than continental drift. More recent evidence supports the view that Gondwanan plants host Gondwanan aphids, and the major primary parasitoids have a Gondwanan origin. Most likely the host-specific hyperparasites do too.

UNANSWERED QUESTIONS

The sections above have indicated many holes in our understanding of these trophically related groups. A few, just related to possible hosts for Australian and New Zealand aphids, are listed below.

1. Are there aphids on mosses and horsetails in southern continents?

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[5] *S. lutea* synonymised under *S. ralumensis*, (Remaudière 1988)

[6] Since noted as a junior synonym of *Endaphis* (Tang *et al*. 1994)

[7] A species of *Sensoriaphis* (now as *Taiwanaphis*) occurs in South America. Schlinger postulated that *Neuquenaphis* would probably occur in Australasia. It might, but it hasn't been found yet.

2. Are there aphids on *Nothofagus gunnii* (Tasmania)?
3. Are there aphids on the Wollemi pine (living fossil related to *Araucaria*, NSW)?
4. Are there aphids on celery-top pine or Huon pine in Tasmania (*Phyllocladus* and *Lagarostrobus*)?
5. Are there aphids on *Agathis* (kauri) in Australia or New Zealand? (*Neophyllaphis rappardi* has been collected from *Agathis labilliardieri* in Papua (B&E)
6. Are there indigenous/endemic aphids on *Dacrydium*, another member of the Podocarpaceae?
7. Are there endemic aphids in Australia on endemic grasses?

Why are the answers to these simple questions (and many others) not already known? The first answer is the elusive nature of aphids - their small size, their rarity, their seasonality, and a lack of perceived charisma for the general entomologist. The second is a lack of search effort. Search effort is limited by availability of informed hunters - many people with knowledge of aphids are tied to research and management relating to agricultural pests, or are working on entirely different aspects of aphid biology. The few aphid taxonomists are often committed to the care and management of large general collections. The second problem is that native aphids are hard to find even if you know they exist, with small, seasonal and transient populations, and often far removed from the cities where the jobs are. Travel is expensive in time and money and unlikely to receive funding. Retired entomologists with an interest in aphids have the time, but there are various hurdles to jump: access to laboratory alcohol becomes difficult, scientific collecting licences need prior planning and may be difficult to obtain, most scientific literature is commercialised and often not available without substantial payment for people without institutional support (my thanks to authors who have provided detailed abstracts). Opportunistic observations, often adjuncts to road trips, have provided a lot of our endemic aphids, but in national parks or state forests collecting is generally illegal without a licence. If you are lucky enough to see what may be a new aphid in a national park, are you going back home to apply for a licence, wait x weeks for it and come back? Not usually feasible and something will probably have eaten the aphids in the meantime. In Australia the licences are different from state to state. Queensland requires separate permission for each national park, or did when I asked. For non-aphidologists, recognising an aphid can be a challenge, let alone deciding on whether it may be new. And most likely they still can't legally collect it to find out. Aphids are complicated and not spectacular, need careful slide mounting for examination (need good stereomicroscope) and identification (need good compound microscope). Identification needs a lot of background knowledge and if you suspect you have a new species, you definitely need a specialist's opinion. Aphid taxonomists are hard to find. This has been an impediment to the descriptions of the new endemic aphids in New Zealand, for example. But there are plenty of opportunities for the courageous!

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SCIENTIFIC NOTE

INSECTS AS SENTINELS OF CLIMATE CHANGE: IMPLICATIONS FOR CONSERVATION, AGRICULTURE AND PUBLIC HEALTH

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Summary

Global warming is reshaping insect populations and the ecosystem functions they support worldwide. Insects have persisted for ~480 million years, closely tracking regional climates and seasonal cycles, but rapid anthropogenic warming is now disrupting these long-standing relationships. Rising temperatures, more frequent extremes and altered rainfall patterns are shifting life cycles, distributions and species interactions, driving declines in habitat specialists while favouring generalist and invasive pests, including vectors of plant diseases. Climate-driven changes in pollinator phenology and mosquito populations further threaten crop pollination and increase public health risks from mosquito-borne diseases. Using Australian case studies of threatened native species and expanding pest and vector populations, this note argues that insect responses to climate change should be explicitly integrated into conservation, biosecurity and public health strategies worldwide.

Keywords: insect biodiversity; climate change; phenological mismatch; endangered species; pollinator decline; pest outbreaks; disease vectors.

GLOBAL WARMING: AN ANCIENT CHALLENGE FOR INSECTS, A NEW CRISIS FOR HUMANS

Global warming may seem like a recent challenge from a human perspective, but for insects it is part of a long evolutionary history of responding to climatic variation. Insects have been thriving on Earth for roughly 480 million years, long before *Homo habilis* appeared about 2.8 million years ago. By the time Australia's Aboriginal peoples arrived around 65,000 years ago, many Australian insect lineages were already well established. Groups such as the Bogong Moth and the Giant Burrowing Cockroach had spent tens of millions of years adapting to local climates and seasonal patterns.

For much of this time, temperature and seasonality provided reliable cues. The warmth of spring signalled development and emergence, while cooler autumn conditions triggered diapause, migration or dormancy. However, recent anthropogenic climate change is altering the baseline on which these evolutionary relationships were built. Since the beginning of national instrumental records in 1910, Australia's mean temperature has increased by about 1.5–1.6 °C, with marked increases in the frequency and intensity of heat extremes (Grose et al. 2023; CSIRO & Bureau of Meteorology 2024). At the same time, historical accounts such as Diego de Prado's 1606 report of "great quantities of flies" in the Torres Strait provide some of the earliest European observations of abundant insect life in Australia. These descriptions were recorded under cooler Little Ice Age conditions. Seen against today's warming trends, this early account highlights that those insects that once flourished in a cooler, more stable climate now have to cope with environments that are significantly hotter and more variable.

For insects, even modest shifts in average temperature can substantially alter developmental rates, reproduction, survival and behaviour. Many species already experience summer temperatures close to their upper thermal limits, so further warming and more frequent heatwaves reduce their thermal safety margins and increase the likelihood of local population collapse. Experimental and theoretical work shows that short, intense heat events can be more damaging than equivalent changes in mean conditions. These brief extremes can push ectothermic insects beyond critical physiological thresholds and alter their interactions with host plants and natural enemies (Harvey et al. 2020; Ma et al. 2015).

In addition to gradual warming, recent years have brought record-breaking anomalies. For example, typical winter maximum temperatures in many parts of Australia have historically been around 18 °C. However, in August 2024, some regions recorded winter maxima of up to 29 °C, roughly 11 °C above the long-term average (The Guardian 2024). In addition, the 2024–25 financial year was Australia's warmest on record, being with the nationally averaged mean temperature 1.68 °C above the 1961–1990 baseline (Bureau of Meteorology 2025). In winter 2025, national mean temperatures remained about 0.5 °C above the long-term average, even though it was the second coolest winter of the decade. Such anomalies disrupt seasonal cues that insects use to time emergence, mating and migration, leading to mismatches with host plants, predators and mutualists. As climate variability increases, insects must cope with both a shifted mean climate and more frequent extremes (Figure 1).

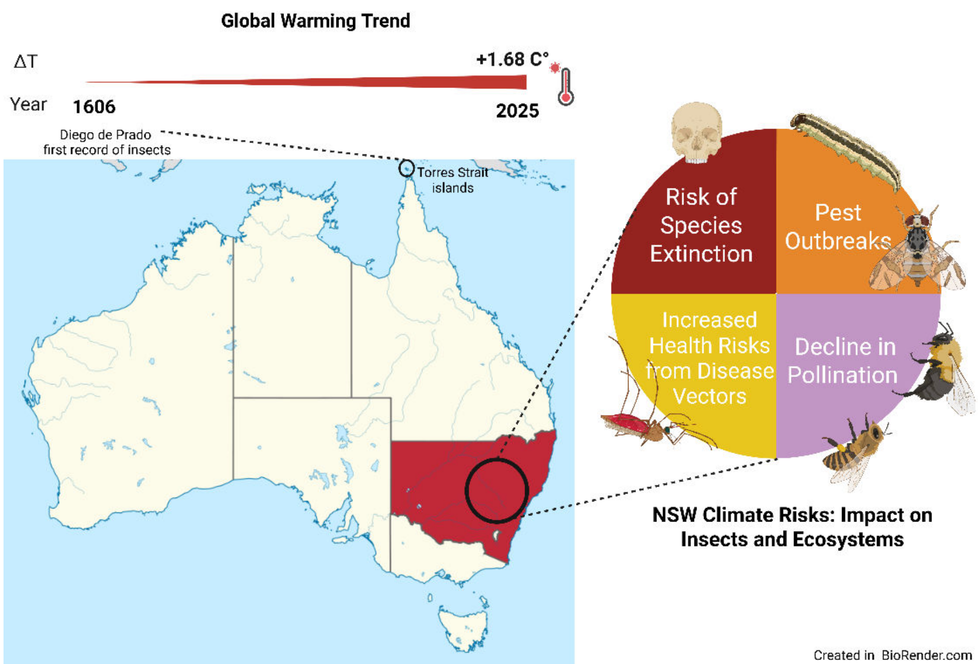

Figure 1. Global warming trends and their impacts on insect populations and ecosystems in New South Wales, Australia. Created in BioRender.com

FALLING OUT OF SYNC: CLIMATE CHANGE AND INSECT SURVIVAL

Extinction risk for plants and animals is escalating in many parts of the world as climate change and habitat loss intensify. In New South Wales (NSW), 1,043 plant and animal species and 115 ecological communities are listed as threatened, and 78 species are already presumed extinct (NSW Environment Protection Authority 2021). Among these, insects are both under-described and under-monitored, yet they are integral to the functioning of virtually all NSW ecosystems. Similar patterns are emerging globally, where climate change and land-use intensification interact to drive changes in insect abundance, richness and community composition across multiple biomes (Outhwaite et al. 2022; Harvey et al. 2023).

The Bogong Moth (*Agrotis infusa*) provides a well-known example of a climate-sensitive migratory insect. This temperate noctuid undertakes long-distance migrations exceeding 1,000 km to aestivate in cool caves and boulderfields of the Australian Alps during summer (Green et al. 2021). Historically, these migrations were so abundant that Aboriginal communities held large gatherings in the alpine region to harvest Bogong Moths as a seasonal resource. This practice is now documented archaeologically at Cloggs Cave in Victoria (Stephenson et al. 2020). Bogong Moth populations have declined markedly since the 1980s, with severe collapses recorded in 2017–2018 and 2018–2019 (Green et al. 2021). These declines are likely driven by a combination of drought and warming in lowland breeding areas and rising temperatures in alpine aestivation sites (Green et al. 2021). In addition, the 2019–2020 'Black Summer' megafires burned large areas of south-eastern Australia, including parts of the Bogong Moth's range and the distributions of many other invertebrates (Hyman et al. 2020; Marsh et al. 2023). These fires added another pressure to populations already stressed by climate-driven changes in temperature and moisture regimes (Marsh et al. 2023; Dole et al. 2023). The resulting loss of moths has cascading consequences for alpine food webs (Green et al. 2021). The Mountain Pygmy Possum relies on Bogong Moths as a key spring food source after hibernation, and suffers reduced condition and litter survival when moth numbers are low (NSW DCCEEW 2025; Parrott 2024). In some caves once packed with aestivating moths, surveys now record few or no individuals, illustrating the vulnerability of cold-adapted migratory species to rapid warming (Green et al. 2021). As Bogong Moth numbers dwindle, the stability of the alpine food web is compromised, highlighting the fragile existence of cold-adapted species in a warming Australia.

The Giant Dragonfly (*Petalura gigantea*), one of the world's largest dragonflies with wingspans up to 12.5 cm, is another emblematic NSW species under pressure (NSW DCCEEW 2020; NSW EPA 2021). This species occurs in peat swamps, bogs and seepages along the coast and ranges of NSW and south-eastern Queensland (NSW DCCEEW 2020). It is listed as endangered under the NSW Biodiversity Conservation Act 2016 (NSW DCCEEW 2020; NSW TSSC 1998). Its larvae spend six to ten years in fossorial burrows that extend below the water table, feeding on small invertebrates and depending on stable groundwater levels and intact peatland hydrology (Baird 2012; Baird 2014). Urban development, longwall coal mining, altered fire regimes and climate-driven drying and warming threaten these upland swamps, compromising both larval habitat and adult emergence (NSW DCCEEW 2020; Baird 2014; Ware et al. 2014). Because *P. gigantea* belongs to the ancient dragonfly family Petaluridae, whose fossil record extends back to the Jurassic, it represents a "living relic" among modern dragonflies (Ware et al. 2014). Therefore, its decline signals the degradation of specialised wetland habitats that support this lineage (Ware et al. 2014).

The Golden Sun Moth (*Synemon plana*), once more widespread in south-eastern Australia, is now critically endangered (Kutt et al. 2015). This small diurnal moth is largely confined to native grasslands and grassy woodlands and depends on wallaby grasses (*Austrodanthonia* spp.) as larval hosts (Kutt et al. 2015). Over 99.5% of temperate native grasslands in south-eastern Australia have been destroyed or heavily modified since European settlement, leaving *S. plana* populations highly fragmented (Kirkpatrick et al. 1995; Williams et al. 2015; NSW TSSC 2016). Adults are short-lived, with non-feeding females that rarely fly and rely on stored reserves and local dispersal (Kutt et al. 2015). This limited mobility, combined with habitat loss, invasive grasses, altered fire regimes and climate extremes, restricts recolonisation potential and reduces genetic diversity, further increasing extinction risk (Kutt et al. 2015; NSW TSSC 2016).

Together, these case studies, from alpine caves to upland peat swamps and lowland grasslands, illustrate how climate change compounds existing pressures such as habitat loss, hydrological alteration and invasive species. The result is a systematic erosion of insect diversity across NSW's major biomes.

CLIMATE CHANGE AND THE EXPANSION OF PEST INSECTS

While many specialist insects are declining, some generalist and invasive species are likely to benefit from warming climates and altered land use. This has direct implications for agriculture and horticulture in NSW. For key pests, species distribution models that combine physiological thresholds with climate projections are increasingly being used to map future risk, and these generally predict range expansions and increased voltinism in warmer southern and inland regions of Australia (Sultana et al. 2017, 2020; Simpson et al. 2020).

The fall armyworm (*Spodoptera frugiperda*) is a highly polyphagous lepidopteran pest native to the Americas that has spread across Africa, Asia and, more recently, Australia (CABI 2025; Maino et al. 2021). It was first detected in northern Australia in early 2020 and is now established in multiple states, including parts of NSW (Plant Health Australia 2020; Maino et al. 2021). Warmer conditions allow *S. frugiperda* to complete multiple generations per year and to overwinter in areas that were previously too cold, transforming a seasonal invader into a chronic, year-round threat in some regions (Maino et al. 2021). The species attacks a wide range of crops, including maize, sorghum and rice, and has the potential to cause substantial economic losses to Australian grain and horticultural industries (CABI 2025; Plant Health Australia 2020). National and international planning efforts, including Australia's Fall Armyworm Industry Contingency Plan for grains and the FAO Global Action for Fall Armyworm Control, emphasise the importance of climate-aware surveillance and integrated pest management in limiting these impacts (Plant Health Australia 2020; FAO 2020).

The Queensland fruit fly (*Bactrocera tryoni*) provides another example of a pest whose distribution and impact are closely linked to climate. Historically, cooler winters limited *B. tryoni* survival and reproduction in temperate regions (Sultana et al. 2017). Rising temperatures and milder winters now enable higher overwintering survival and longer breeding seasons, increasing pressure on fruits such as peaches, plums and cherries (Sultana et al. 2017; Sultana et al. 2020). Infestations already cost Australian horticulture an estimated \$300 million annually (Plant Health Australia 2018). Modelling studies project that suitable habitat, and the number of generations per year for *B. tryoni*, will increase in many southern and inland horticultural regions under future climate scenarios, including parts of NSW (Sultana et al. 2017, 2020; Simpson et al. 2020). In addition, recent changes in distribution and quarantine boundaries show that climate-related shifts in *B. tryoni* range have influenced market access and management zones in eastern Australia (Dominiak & Mapson 2017).

An additional, increasingly important issue for crop production is insect-vectored plant disease. In New South Wales (NSW), Tomato spotted wilt virus (TSWV) is one of the most widespread and damaging viruses of vegetable and ornamental crops and is endemic in the region (Persley et al. 2006; Plant Health Australia 2011; Agriculture Victoria 2021). It is transmitted by a complex of thrips species, including western flower thrips *Frankliniella occidentalis* and onion thrips *Thrips tabaci*, which acquire the virus as larvae and can transmit it throughout their adult lives (Persley et al. 2006; Agriculture Victoria 2021). Because TSWV replicates within its thrips vectors and persists in a wide range of crop and weed hosts, effective management requires simultaneous suppression of vector populations, removal of infected plants and control of alternative hosts; insecticides alone are seldom sufficient once virus–vector cycles are established (Plant Health Australia 2011; Agriculture Victoria 2021). Warmer conditions that favour rapid thrips population growth, together with documented insecticide resistance in some thrips populations and resistance-breaking TSWV strains in capsicum and other crops, are likely to increase the frequency and severity of outbreaks under climate change (Sharman & Persley 2006; Chen et al. 2021).

These trends highlight a key duality of climate change for insects: while many native specialists are pushed towards the margins of their climatic niches, mobile, generalist and invasive species can exploit new opportunities, often at significant economic cost.

POLLINATION UNDER PRESSURE

Beyond their roles as pests or threatened species, insects underpin agricultural production through pollination services. Bees, including managed European honey bees and diverse native bee species, are critical pollinators of crops such as almonds, apples, blueberries and many other horticultural species in Australia (Cunningham et al. 2013; Rader et al. 2016). Native stingless bees such as *Tetragonula carbonaria* are increasingly managed as pollinators of macadamia, mango and other fruit crops in eastern Australia, highlighting the importance of native insects for crop production (Heard 2016; Cunningham et al. 2013). A substantial proportion of Australia's agricultural production is dependent, to varying degrees, on insect pollination (Aizen et al. 2009; Cunningham et al. 2013). Reviews of plant–pollinator interactions indicate that global warming is already generating spatial and temporal mismatches between flowering and pollinator activity, with likely consequences for seed set and crop yields (Gérard et al. 2020; Stoddard 2017).

Climate change can disrupt these services via phenological mismatches. Warmer springs may cause bees and other pollinators to emerge earlier, while flowering in crops and native plants may advance at a different rate. When pollinators and flowers are no longer synchronised, the effective window for pollination is shortened, reducing fruit set and yield. Heatwaves can directly impair pollinator performance and flower viability, further reducing pollination efficiency.

In addition, habitat loss and fragmentation, pesticide use and competition from managed honey bees interact with climatic stressors to reduce pollinator abundance and diversity. In Tasmania, the introduced large earth bumblebee *Bombus terrestris* now contributes to pollination of berries and greenhouse crops. However, it invades native vegetation and has the potential to displace native pollinators and promote invasive weeds, illustrating the trade-offs involved when novel pollinators establish under changing climates (Hingston & McQuillan 1999; NSW Scientific Committee 2010). For Australian agriculture, maintaining resilient pollination networks under climate change will require integrated approaches that conserve nesting and floral resources, reduce chemical pressures and incorporate climate adaptation into orchard and crop management.

MOSQUITO-BORNE DISEASE IN A WARMING WORLD

Insects affect human wellbeing worldwide through their roles as vectors of disease. Across Australia, mosquitoes transmit pathogens such as Ross River virus (RRV) and Barmah Forest virus, which cause febrile illness with rash and often debilitating polyarthritis (Webb 2020; Hime et al. 2022). In New South Wales (NSW), these viruses are major causes of locally acquired arboviral disease, with roughly 700 notified cases of RRV each year and larger outbreaks in some years linked to climatic and environmental conditions (Webb 2020; Qian et al. 2020; Hime et al. 2022). Temperature strongly shapes RRV transmission potential, with risk peaking at intermediate temperatures typical of many temperate and subtropical regions (Shocket et al. 2018). In coastal and inland NSW, RRV and Barmah Forest virus are transmitted mainly by the saltmarsh mosquito *Aedes (Ochlerotatus) vigilax* and the common banded mosquito *Culex annulirostris*, with *Aedes (Ochlerotatus) camptorhynchus* contributing in southern estuarine habitats (Webb 2020; Shocket et al. 2018; Hime et al. 2022).

Warming temperatures and altered rainfall patterns are likely to extend the seasonal window of mosquito activity and increase the frequency of favourable breeding conditions, particularly in peri-urban wetlands, floodplains and container habitats. Warmer conditions can shorten mosquito development times and increase biting rates, while changes in rainfall and extreme events such as floods can create or expand larval habitats. Recent analyses of NSW and neighbouring jurisdictions show that RRV and Barmah Forest virus notifications often increase following periods of heavy rainfall and flooding, underscoring the importance of weather extremes for arboviral risk (Hime et al. 2022; Qian et al. 2020).

Extreme events interact with these dynamics. Flooding can generate extensive breeding sites, whereas bushfires can alter wildlife distributions and bring reservoir hosts and humans into closer contact. Following the 2019–2020 bushfires, for example, NSW experienced an unusual combination of fire, smoke, COVID-19 restrictions and increased RRV activity (Webb 2020). Such compound events complicate disease prediction and control, placing additional demands on public health systems.

As climates continue to warm, medical entomology will become increasingly important for surveillance, early warning and integrated mosquito management. Understanding how temperature, rainfall and land-use interact to shape mosquito populations will be critical for anticipating future disease risk in NSW.

A SHARED FUTURE IN A CHANGING WORLD

Insects perform a wide spectrum of functions—pollinating native plants and crops, decomposing organic matter, regulating pest populations and supporting the diets of birds, mammals and reptiles (Prather et al. 2013). Their responses to climate change—whether decline, range shift or population explosion—will reverberate across ecosystems, economies and public health.

From an entomological perspective, global warming is not only an environmental issue but also a complex biological and societal challenge. Conservation of threatened insect species, management of emerging and expanding pests, and surveillance of vector-borne diseases all depend on a clear understanding of how insects experience and respond to a rapidly changing climate.

Practical actions such as preserving and restoring native vegetation, reducing chemical inputs, managing water resources, and mitigating greenhouse gas emissions can help buffer insect communities and the services they provide. For entomologists, documenting insect responses, informing policy and fostering collaboration between conservation, agriculture and public health sectors will be essential to safeguard both insect biodiversity and human wellbeing in a warming world.

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SCIENTIFIC NOTE

IN TIMES OF *VARROA* – LET'S NOT FORGET THE INS AND OUTS OF THE SMALL HIVE BEETLE, *AETHINA TUMIDA*

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Summary

The small hive beetle, *Aethina tumida*, one of the most devastating pests of the honey bee *Apis mellifera* has the capability to destroy an entire colony within weeks including by causing slime-out of honey bee colonies due to the fermentation of honey by its symbiotic yeast.

Keywords: Small hive beetle, *Aethina tumida*, wandering larvae, honey bees, Varroa.

INTRODUCTION

The European honey bee, *Apis mellifera* L. (Hymenoptera: Apidae) is one of the most important beneficial insects, providing hive products like honey, beeswax and propolis to humans along with the all-important crop pollination services. It is globalization that has made it possible for *A. mellifera* to reach every continent on Earth (except Antarctica), which proved to be a boon to humanity. But globalization has also shown that it can be a curse, and one of those is the spread of parasites and pests of honey bee. To mitigate this, strict biosecurity regulations and measures are in place in many countries, including Australia. Owing to these, Australia has been successful in intercepting and eradicating several incursions of the most destructive and significant honey bee parasites, *Varroa* mites.

However, when *Varroa destructor* was detected in sentinel and other hives near Newcastle, New South Wales (NSW), in June 2022, the large eradication effort that followed suit unfortunately remained unsuccessful because the mite had already reached areas outside the established eradication zones. Therefore, in September 2023 the focus shifted from eradication to management (NSW DPI, 2024). *Varroa* is now established in many parts of NSW and has also been detected in Victoria. As honey bee-specific ectoparasite, *Varroa* is capable of drastically weakening and reducing honey bee colonies, through direct parasitism but also by vectoring viruses that affect bee health. Importantly, *Varroa* can also make honey bee more vulnerable to attack by other honey bee pests already present in Australia. Among these, the small hive beetle (SHB), *Aethina tumida* Murray (Coleoptera: Nitidulidae) is the most significant.

SMALL HIVE BEETLE

Currently, SHB occurs in many parts of the globe including Africa, parts of America, Europe, as well as Australia. It is native to sub-Saharan Africa, where it is considered a minor pest because of the effective hygienic behaviour of the more aggressive African bees. But in response to their aggressive behaviour, SHB has also developed defence strategies like running away, hiding in cracks, and dropping from frames. However, domesticated honey bees, reared for tameness outside their native range are far more susceptible to SHB attack. In Australia, SHB was first reported in Richmond (NSW) in July 2002. Because it had already spread across other regions by November 2002, and, because it can fly and use alternate resources than the ones found in hives, an eradication was not deemed feasible (Spooner-Hart et al., 2017).

The SHB may, by its name, give the impression of being just a small pest, but it causes a substantial problem for apiculture and has substantially affected honey bees in Australia, especially in the coastal regions of NSW and Queensland. In search of a suitable colony with full honey and pollen reserves, adult SHB may fly long distances (up to 15 km), and, thereby, can quickly spread over a large area. Initially their movement is random, but they can recognize the odour and volatiles from a bee colony when present nearby. In between, they may attack feral colonies or might also survive on alternative resources like ripe fruits (e.g. rockmelon, mango, peach, banana and grapes), or on decaying meat.

Once, the adults reach a hive, they may enter through cracks and crevices, or through the main hive entrance, bypassing guard bees, the first line of a colony's defence. They are able to do so because of their cuticular hydrocarbon profile that is similar to that of honey bees, but at 10-fold lower concentration (Papach et al., 2021). When inside the hive, the adult SHBs are chased down by the adult worker honey bees that stop them from roaming freely inside the hive. Worker bees achieve this by corralling or confining SHB to specific parts of the hives known as confinement sites, or by simply preventing SHB from ovipositing by encapsulation in propolis. Although, adult SHB have the capacity to live without food for about two weeks, while being corralled by honey bee workers, SHB have developed an unusual strategy of tickling adult honey bees using their antennae. In this way they obtain food from the honey bees, via trophallactic feeding. For these and other reasons, the SHB is a very difficult insect to manage. Both males and females may mate multiple times, either inside or outside of the honey bee colony and the females lay eggs in clusters of 10-30 eggs within capped brood cells, cracks and crevices, or on the outer frames due to the patrolling (second line of defence) of honey bees around the brood area. Oviposition of SHB can be stimulated when SHB females have access to protein (pollen and yeast). A SHB female may oviposit around 1000 eggs during her lifetime, perhaps even up to 2000 eggs. The SHB larvae hatch from the eggs within one to three days, depending on humidity and temperature, and, for 6-14 days, the larvae feed on almost everything which crosses their way inside the hive, from honey and pollen reserves by penetrating the wax cells to honey bee brood (egg, grub and pupa). Apart from this, the adults (and possibly the larvae) have also been reported to show cannibalism by feeding on SHB eggs, as well as weaker and dead conspecifics (Neumann et al., 2016). Thereby, the SHB has the ability of taking down an entire honey bee colony in just a couple of weeks, in the case of a severe infestation.

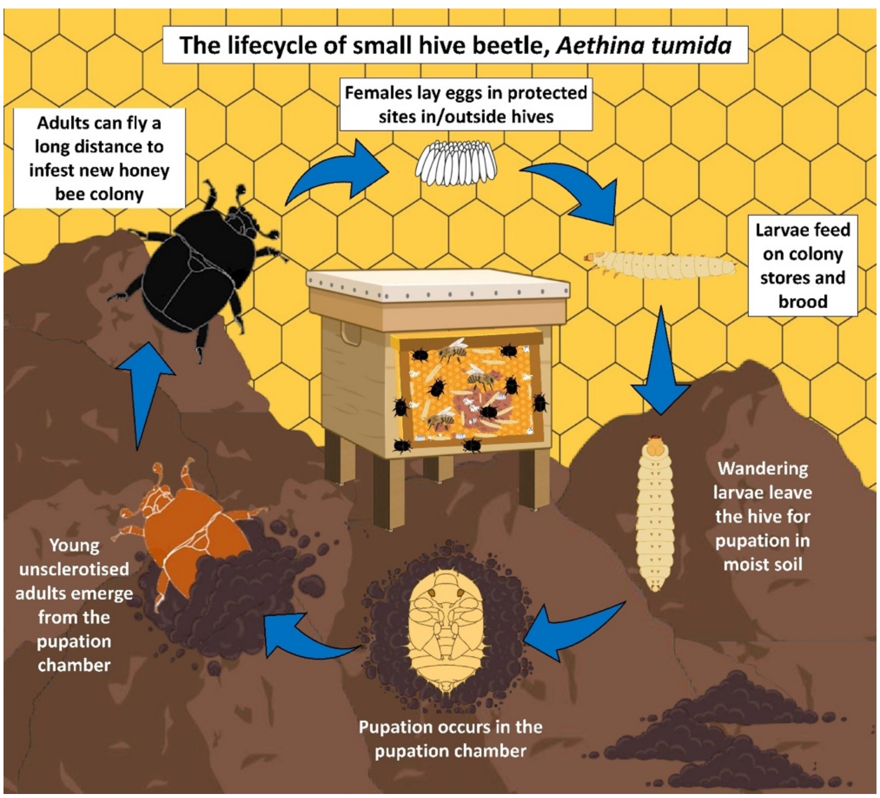

Figure 1. Life cycle of small hive beetle

Larvae have three pairs of thoracic legs, a characteristic row of paired dorsal spines on each segment and two larger paired spines on the rear end of the dorsum, which are helpful in distinguishing them from wax moth larvae that are also common honey bee pests. Most troublesome is though that SHB infestation can lead to the characteristic, smelly and damaging manifestation of the slime-out of infested hives; this occurs due to the fermentation of honey stores by the yeast *Kodamaea ohmeri* which is symbiotically associated with SHB (Amos et al., 2018).

The SHB can have multiple generations per year and can keep feeding on the hive contents even after the colony has died or absconded. The fully developed last stage larvae, known as wandering larvae, are positively phototactic and leave the hive in search of suitable soil for pupation which occurs within 3-4 days. The wandering larva and the pupa are the most vulnerable stages of *A. tumida*, and hence a good target for the application of management strategies. These two SHB developmental stages are outside the hive. Being outside the hive is an adaptive advantage for SHB to be removed from honey bee defence during this inactive and longest period of SHB life cycle as pupa (21-28 days, depending on temperature). Honey bee workers are capable of eating SHB eggs and deposing SHB larvae and eggs outside the hive (third line of defence), but only in strong colonies (Neumann et al., 2016). The wandering larva can move up to 200 m in search of suitable pupation sites in soil with optimum temperature, soil moisture and soil type to create a pupation chamber. After 3-4 weeks, the unsclerotised brown teneral SHB adults emerge from the soil and become sclerotized to turn black and fly in search of new hives. Even before adult emergence, subterranean mating has also been reported in SHB. The sex ratio of adult populations appears to be biased towards females and the adults may live up to 6 months, depending on temperature and food availability. Hence, SHB may be considered as a successful invasive ecological generalist because it can act as a scavenger and herbivore, parasite, host and vector for a yeast symbiont that damages colonies, vector of bee pathogens such as viruses and predator. This makes SHB a challenging issue for beekeepers in Australia, in particular in the context of the recent establishment of *Varroa* as the two pests have the potential to interactively damage honey bee colonies therefore requiring integrated pest management strategies that target both pests.

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A REVIEW OF THE BIOLOGY OF BRAULA FLY (*BRAULA COECA* NITZSCH 1818) (DIPTERA: BRAULIDAE) AND ITS SIGNIFICANCE FOR EUROPEAN HONEYBEES IN AUSTRALIA.

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Summary

Braula fly (*Braula coeca*) has worldwide distribution but is usually considered a minor pest of honey bees (*Apis mellifera*) in most countries. In Australia, it has been present in Tasmania for at least 90 years but was found only recently on the mainland. The wingless fly has strong capabilities to grasp the hairs on honey bees and can move quickly on the host. Braula fly is a honey bee inquiline, inhabiting the honey bee colony within which it undertakes all stages of its life cycle. They use chemical camouflage to avoid detection by the colony. The honey bee queen is the preferred host and the fly steals food from the bees as they are fed. Heavily infested queens may have decreased egg laying, and braula fly larvae damage the caps of honeycomb. Usually, braula fly populations are controlled by treatments for the ectoparasitic mite *Varroa destructor*.

Key words; ectoparasite, attachment, bee, pheromone, *Apis mellifera*

INTRODUCTION

The European honey bee (*Apis mellifera* L.) (EHB) were first imported into Australia in 1822 (Schlipalius 2023) and the industry continues to grow. Australian exports of agricultural commodities in 2019/2020 were valued at more than A$25 billion with many industries relying on pollination by EHB (Plant Health Australia 2021). In 2021, the Australian beekeeping industry including approximately 25,000 registered beekeepers managing about 672,200 colonies (Plant Health Australia 2021). The Australian beekeeping industry faces many endemic pests, diseases, and extremes of climate (Roberts *et al.* 2015; Bourke 2020a,b; Frost 2020; Bernasconi and Honan 2022; Schlipalius 2023).

Several exotic insect pests have established in Australia and adversely impact honey bee activities including European wasp (*Vespula germanica* F.), small hive beetle *Aethina tumida* (Murray), braula fly (*Braula coeca* Nitzsch 1818), wax moths, (*Galleria mellonella* (Linnaeus 1758) and *Achroia grisella* (Fabricius, 1794)), and giant willow aphid *Tuberolachnus salignus* (Gmelin) (Brown 1979; Gillespie *et al.* 2003; Dominiak and Worsley 2018). The ectoparasitic mite, *Varroa destructor*, has recently established on the east coast (Bourke *et al.* 2024). Many of these insect pests rapidly became widely distributed in Australia (Horwood *et al.* 1993; Neumann *et al.* 2010; Kwadha *et al.* 2017). Conversely, braula fly has been slow to expand its range beyond Tasmania.

BRAULA FLY BIOLOGY

Braula fly (sometimes called *bee lice* or *bee louse*) is a small red-brown, flattened, wingless parasite that lives on the bodies of bees. Much of the life history and knowledge of braula fly was summarised by Knutson (1978) and Morse (1987) and will not be covered in detail here. In brief, braula fly eggs are 0.84 mm by 0.42 mm. Eggs may be deposited in many locations however only eggs deposited in capped honeycomb will hatch (Frost 2024). Larvae hatch and tunnel under the beeswax cappings, leaving 1 mm tracks across the surface of the honeycomb. The larval tunnelling results in a fractured appearance and this is a key indicator for the presence of braula fly (Frost 2024). Larvae progress through three instars (7 – 11 days) and subsequently pupate. The pupae are 1.4 – 1.7 mm long and 0.5 – 0.75 mm wide, and the pupal period lasts about three days. The development from egg to adult takes 10 – 23 days, depending primarily on temperature. Adult braula fly survive on adult bees and are not known to survive in their absence (Frost 2024).

Braula fly moves close to the mouth parts of bees and steals small amounts of the food fed to it by other bees (Frost 2024). Braula fly feed on honey and pollen directly from the mouth of its host during trophallaxis where food is shared between bees (Martin and Bayfield 2014). Queen honey bees receive a much higher frequency of feeding compared to workers and drones (Barton-Smith and Caron 1984; Martin and Bayfield 2014). In *Braula pretoriensis,* which also infests *Apis mellifera*, Esnault *et al.* (2019) reported that flies could induce regurgitation by striking the upper end of the bee's labium until the bee extended its tongue.

ATTACHMENT TO BEES

Braula fly has six legs with feet that have several comb-like teeth or claws which enable braula fly to grip to the hairs of the bee's body (Buscher *et al.* 2021). Despite this, adults can move freely, quickly and efficiently in any direction on the host (Buscher *et al.* 2021). Adults stay resting on the host thorax for most of the time (Buscher *et al.* 2021). However, braula fly often move their legs back and forth to capture hairs in close proximity and may collect bunches of hairs to secure attachment (Buscher *et al.* 2021). This grip allows the braula fly to avoid detachment during bee grooming (Buscher *et al.* 2021). These attachment forces have a safety factor of >1000 (Buscher *et al.* 2021). Parasitised bees, particularly queens, are restless, nervous, weakened, and occasionally shake their legs or rub their bodies with their wings to remove the parasites, usually without success (Marchiori 2023).

DAMAGE AND IMPORTANCE

Some authors consider that braula fly is not a serious threat to commercial beekeeping, as it does not damage or parasitize any stage of the honey bee life cycle (Honan 2022; Biosecurity New Zealand 2022; Marchiori 2023). Braula fly is of very little economic importance and there are no international trade implications (Anonymous 2022). The main economic effect of braula fly is that the larval stage burrows under the cappings of the honeycomb and the damage can detract from the appearance of honeycomb for sale making it unsaleable or of reduced value (Frost 2024).

However, dense braula fly populations in hives can be significant. For instance, Benton (1896) found 75 braula fly on one queen although numbers of less than a dozen were more common. Bailey (1963) suggested that high levels of braula fly on a single queen could decrease her egg-laying capacity. In severe cases, an infested queen can carry up to 100 adult braula fly which caused reduced egg laying (Bailey and Ball 1991). Heavily infested queens may be seen as inferior or old and replaced by a younger queen (supersedure) (Tarpy 2024). Heavy larval infestations especially in weak colonies causes paralysis of larvae and decreases the queens egg laying efficiency (Marcangeli *et al.* 1993; Yusuf *et al.* 2024). Also, death of developing bees could occur (Yusuf *et al.* 2024). Its potential as a pathogen vector has not been fully explored (Marchiori 2023). Colwell *et al.* (2023) were the first to establish the presence of black queen cell virus (BQCV) and two strains of deformed wing virus (DWV-A and DWV-B) in braula fly in Canada, but they did not establish if braula fly could serve as a viral vector.

BRAULA PREFERENCE FOR DRONES, WORKERS OR QUEENS

The fact that queen bees are fed more intensively than other EHB castes is likely the reason that queens may be preferred over workers and drones as braula fly hosts. However, the published data on host preference within EHB hives is quite equivocal.

Worker bees

In America, braula fly was found usually on worker bees (Phillips 1925). Barton Smith and Caron (1984) found that 98.6% of workers had a single braula fly.

Drones

Yusuf *et al.* (2024) found that braula fly rarely attaches to drones (males). Young drones were more likely to be infested versus old drones (Barton Smith and Caron 1984).

Queens and nurse bees

Braula fly preferred to attach to the queen, rather than the drone or worker bees (Phillips 1925; Honan 2022). Often, braula fly were found on the queen and drones (Biosecurity New Zealand 2022). Virgin and mated queens were more likely to be infested than young drones (Barton Smith and Caron 1984). Mated queens were more likely to be infested than virgin queens (Barton Smith and Caron 1984; Buscher *et al*. 2021). Braula fly has a preference to infest the queen (Martin and Bayfield 2014) or the queen and nurse bees (Yusuf *et al.* 2024) with 62% of examined queens harboured lice (Barton Smith and Caron (1984). Queens and nurse bees are preferred hosts because they smell like a queen, and queens receive more food than workers (Korst and Velthius 1982; Martin and Bayfield 2014; Yusuf *et al.* 2024).

Normally, alien species in an EHB colony are attacked but braula fly have evolved a survival mechanism. The queen is the most attended and longest living individual in the colony, but braula fly remains undetected on queens by the worker bees. Braula fly possesses a cuticular hydrocarbon profile that mirrors that of their host honey bee colony (Martin and Bayfield 2014). This chemical camouflage is most likely due to odour acquisition from the honey bee host: even small colony-specific differences in the alkene isomer patterns of the honey bees also were detected in the associated braula fly profiles (Martin and Bayfield 2014).

For braula fly, this odour acquisition may have other advantages. Braula fly successfully re-mounted worker bees that previously carried them but not worker bees that had not carried them, with those carrying them having more 9-oxo-2(E)-decenoic acid (9-ODA; queen pheromone) and 10-hydroyx-2(E)-decenoic acid (10-HDA; worker pheromone) (Yusuf *et al.* 2024). By eavesdropping on their host's pheromones, braula fly makes choices regarding which bee to use as a host, and this technique should result in their higher prevalence and survival (Yusuf *et al.* 2024).

DETECTION AND TREATMENT

Braula fly is not considered a significant pest (Honan 2022; Marchiori 2023) however apiarists should check the health of colonies. Examination of adult worker and queen bees may reveal the presence of braula fly. When viewed with the naked eye, braula fly can be confused with *Varroa* spp., *Tropilaelaps* spp. (ectoparasitic mite), and *Mellitiphis alvearios* (pollen mite) that are found sometimes in colonies (Buscher *et al*. 2021; Biosecurity New Zealand 2022). Additionally, detection methods used for *Varroa* spp. are likely to be suitable for braula fly (Frost 2024). The methods include alcohol wash, soapy water wash, or sugar shake of 300 bees (Frost 2024; Taylor *et al.* 2025).

Usually, treatment is minimal (Frost 2024). The standard practise of uncapping honeycomb during extraction is an effective means of larval control (Frost 2024). Freezing for at least 24 hours will kill all stages of braula fly (Biosecurity New Zealand 2022; Frost 2024; Honan and Webster 2024).

For adult braula fly, early remedies included a bag of naphthalene on the bottom of the hive which caused the fly to drop from the bees to the bottom of the hive, where they could be captured (Phillips 1925, Marchiori 2023). However, naphthalene can drive the bees from the hive, perhaps depending on the dosage (Phillips 1925). Nicotine smoke numbed braula fly and they fell from their host to the floor of the hive (Marchiori 2023).

Fumigation with 0.02 g of amitraz or aerosol treatment with 0.006 g of amitraz killed very few braula fly (Kulincevic *et al.* 1991). Fumigation with 0.0025 g of fluvalinate killed many braula fly but the aerosol application using 0.0012 g of fluvalinate killed significantly more braula fly (Kulincevic *et al.* 1991). Treatment timing may be important; for *Braula orientalis* Orosi, treatment in early summer was likely to keep pest populations low for the remaining seasons (Ghzawi *et al.* 2009). This timing was based on the hypothesis that braula fly has one generation per year (Marchiori 2023).

Following the recent establishment of *V. destructor* on June 2022 in New South Wales (NSW), Australia (Bourke *et al*. 2024), the use of miticides for *Varroa* management also should provide control of braula fly populations (Frost 2024) in areas where both pests are present. This is because amitraz and fluvalinate, used to control *V. destructor,* are effective also on braula fly. In August 2024 and March 2025, *V. destructor* was detected in Victoria and Queensland respectively (Agriculture Victoria 2024; Queensland Government 2025).

Braula fly prefers clean and pristine conditions and dies in response to exposure to many chemicals, even nicotine smoke (Honan and Webster 2024). There are no specific pesticide treatments registered in Australia for braula fly (J. Kidston (NSW DPI) 2024, pers. comm.). A new fungal species, *Dimeromyces braulae* (Ascomycota, Laboulbeniales) was described on braula fly in the Czech Republic with an infection rate of 12.6% (Rossi *et al.* 2016) and may be a biological control in the future.

DISPERSAL AND DISTRIBUTION

Dispersal happens because braula fly attaches itself to adult honey bees. Therefore, braula fly is spread by swarms, drifting bees (bees returning accidentally to the wrong hive), packaged bees (a package of many workers and a single queen used to initiate new colonies) and queen bees (Buscher *et al.* 2021; Frost 2024). Drifting drones, infested with braula fly, are accepted readily in other colonies within an apiary (Buscher *et al.* 2021). Additionally, robbing behaviours (removal of resources from weak hives by strong hives) and absconding bees can spread braula fly (Biosecurity New Zealand 2022). Internationally, braula fly adults can be introduced on imported infested queens (Phillips 1925). Additionally, braula fly larvae are spread by the removal and transport of infested honeycomb (Frost 2024).

Braula fly was first reported from Europe, Russia, and South Africa in the early 1900's, and there were frequent introductions in the USA (Phillips 1925). Apart from Antarctia, braula fly is currently widespread overseas and was reported from all other continents except mainland Australia until recently (Honan 2022; Marchiori 2023).

In Australia, braula fly was not present in the early 1900's (Gale 1905). By 1932, braula fly was found at many areas in Tasmania (Anonymous 2024) and now is considered endemic in Tasmania (Honan 2022; Frost 2024).

Nearly a century later, braula fly was detected in multiple apiaries in Victoria in August 2022 (Anonymous 2022). The Consultative Committee on Emergency Plant Pests (CCEPP – see Anderson *et al*. 2017 for details) advised all other states of the extension of range (Anonymous 2022). Braula fly remains a notifiable pest in Victoria under the Livestock Disease Control Act 1994, and beekeepers must report the suspected presence of braula fly. The pest had been eradicated several times at different locations in Victoria (Honan 2022).

In September 2022, braula fly was detected in NSW for the first time in colonies originating from Victoria, following an illegal movement of colonies from the Sunraysia region to NSW (Honan 2022). The infested colonies were returned to Victoria to help keep NSW free from braula fly (Honan 2022). Movement restrictions were implemented to control the movement of potentially infested colonies to delay or prevent establishment in NSW. In January 2024, braula fly was detected in colonies near Tamworth NSW (Honan and Webster 2024). There were no interstate links with the Tamworth detection (Honan and Webster 2024). No attempt was made to eradicate the infestation as there was no economic or scientific justification (Honan and Webster 2024). Braula fly remains a notifiable pest in NSW (Honan 2022; Frost 2024). NSW was mindful that the cost of regulations should not exceed the economic importance (Anonymous 2022). Currently, braula fly is considered established in Tasmania, Victoria and New South Wales (Frost 2024). However, any suspect detections should be reported to the free call Emergency Plant Pest Hotline on 1800 084 881 (Frost 2024).

Braula fly has not been detected in Queensland (Schlipalius 2023) despite Tamworth being relatively close to the Queensland boarder. Braula fly was not found in a survey of Norfolk Island (Malfroy *et al.* 2016). Given that honey bee imports into Norfolk Island ceased in 1992, it is hoped that Norfolk Island will remain free from braula fly.

CLOSING COMMENTS

Braula fly is likely to remain part of beekeeping in south eastern Australia as it is well established. Braula fly remains exotic to New Zealand and is a notifiable organism there (Biosecurity New Zealand 2022), and has not been reported in other Pacific nations. Pacific nations may restrict or regulate imports from Australia so that they can remain free from braula fly. The need to actively manage braula fly is likely to be supplanted by management procedures required to control populations of *V. destructor* as it likely spreads throughout south eastern Australia. Other Australian states without *V. destructor* will need to check colonies for braula fly and may need to introduce treatments if pest populations increase. Early summer may be the best time for treatments.

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A CHECKLIST OF ARTHROPODS IN CAMBODIAN AGRICULTURAL SYSTEMS

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Summary

Cambodian farmers rely almost exclusively on chemical insecticides for controlling insect pests in agricultural production systems. Farmers indiscriminately apply a calendar-based cocktail of chemical insecticides aimed at controlling insects in general, regardless of whether they are pests or whether they are present in the crop or not. Farmers are left to rely on themselves and pesticide input sellers for 88% of the information they receive on insect pest management and are "locked-in" to pesticide use through the supply chain. Farmers are generally not aware that insect pests have natural enemies which are being decimated by indiscriminate use of broad-spectrum insecticides. The preservation of natural enemies of insect pests is a fundamental objective of Integrated Pest Management (IPM). It is a priority that farmers should be taught how to recognise natural enemies and then they need to learn how to preserve them. The purpose of this article was to compile a checklist of arthropods found in Cambodian agricultural systems that can be used as a basis for the development of training resources for IPM. The checklist contains hyperlinks to entries of the specimens uploaded to the iNaturalist App. The checklist currently includes over 300 entries for arthropods. Fifty-three percent of the taxa are considered to be pests and 36% as beneficial in that they are predators or parasitoids of insect pests. Eleven percent of taxa are considered to be neutral in that they do not directly affect pests. Ninety percent of specimens are identified to genus level and 62% to species level. Using the checklist to help with provision of IPM training to farmers is the first step but there is a need to expand training and provision of resources to include other actors along the value chain and especially retailers of insecticides. Lack of access to IPM inputs, market constraints, and access to training are among the key barriers preventing farmers adopting IPM strategies. Retailers should be encouraged to stock relevant IPM tools such as pheromone traps, sticky traps and bio-pesticides. In addition, this Arthropod Database can be used as a resource for the preparation of banners, posters and leaflets for display at retail outlets. Pesticide retailers should be encouraged to stock relevant IPM tools such as pheromone traps, sticky traps and bio-pesticides.

Key words: integrated pest management, insecticide, predator, parasitoid, economic threshold

INTRODUCTION

Cambodian farmers rely almost exclusively on chemical insecticides and spray up to 20 times per season, especially in vegetable crops (Srinivasan et. al, 2019). Growers of mungbean (*Vigna radiata* L. Wilczek) in Battambang province mostly apply non-selective broad-spectrum insecticides at least 10 times per crop cycle (Martin et. al., 2020). Such indiscriminate use of insecticides risks the evolution of insecticide resistance and consequent failure to kill the target pest, which might increase its numbers or result in a secondary pest outbreak of a formerly minor pest (Srinivasan et. al, 2019). For instance, the main insect pest problem in mungbean crops in Preaek Trab Village Battambang Province, has become the bean flower thrip (*Megalurothrips usitatus* Bagnall), which appears to be a secondary pest outbreak as it was previously a minor pest (Martin et al. 2021). Bean flower thrip was first observed in 2019 at Preaek Trab village (Martin et al. 2020) but was not recorded by Hinchcliffe (2018) in the same area in 2018. Mungbean growers in Preak Trab village indiscriminately apply a calendar-based cocktail of broad-spectrum chemical insecticides aimed at controlling insects in general, regardless of whether they are pests or whether they are present in the crop or not. Spraying commences at crop emergence and is repeated every six days during the crop cycle.

Cambodian farmers usually do not intentionally protect natural enemies of insect pests (Schreinemachers et al., 2017) and use of registered bio-pesticides is rare and negligible. Hence, farmers almost exclusively rely on chemical pesticides. In general, the value of pesticides imported into Cambodia has increased by 69-fold during the period 1995–2015 (FAO, 2015). Flor et al. (2018) concluded that the over-reliance on pesticides in Cambodia has resulted from a lack of alignment of policy, lack of extension programs, lack of access to and market supply of alternative technologies. For example, mungbean farmers in Preaek Trab village are left to rely on themselves and pesticide input sellers for 88% of the information they receive on insect pest management (Hinchcliffe, 2018). Donors account for 9% of information but government extension services were not mentioned by farmers (Figure 1). According to Flor et al. (2018), the private sector, especially village retailers, should be the focus for information delivery on insect management and be encouraged to expand their product base to include Integrated Pest Management (IPM) products, especially the retail of already registered biological insecticides. Hinchcliffe (2018) found that mungbean farmers in Preaek Trab village were mostly not aware that insect pests have natural enemies.

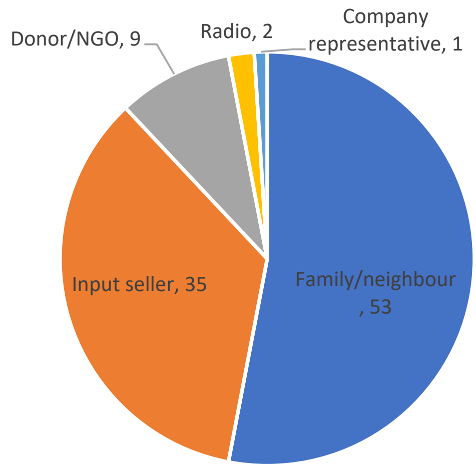

Figure 1. Farmers' sources of information on the control of insect pests of mungbean in Preaek Trab village Battambang province (Hinchcliffe, 2018)

Farmers were presented with photographs of five insect species commonly found in their mungbean fields, three being pests and two being natural enemies. Seventy-one percent of interviewees identified the beneficial ladybird (*Cheilomenes sexmaculata* Fabricius) as a crop pest, while the remaining interviewees classed natural enemies as either having 'no effect' or 'don't recognise'.

No respondent acknowledged the natural enemies as having a beneficial impact on the crop. The majority of respondents identified all five insects as being bad for the crop. Eighty-six percent of interviewees stated they would like to receive more information on insect control, with 82% wanting further information specifically on insecticides. However, only 32% expressed a desire to receive more information on biological control.

Farmers need to become more aware that not all insects found in their crops are pests and some arthropods (insects and spiders) play a significant role in keeping insect pests from causing economic damage to crops. Natural enemies of insect pests include predatory arthropods such as spiders (Araneae), ground beetles (Coleoptera), ladybird beetles (Coleoptera), robber flies (Diptera), true bugs (Hemiptera), ants (Hymenoptera), wasps (Hymenoptera), mantids (Mantidae), dragonflies (Odonata) and lacewings (Neuroptera). Ladybird beetle adults and larvae are important predators of small insects such as aphids and insect eggs. *Cheilomenes sexmaculata* Figure 2 is a common predator of insect pests of both upland crops (Hinchcliffe, 2018) and lowland rice (Dunn, 2022) in the region.

Parasitoids are insects whose larvae feed and develop within or on the bodies or within eggs of other arthropods. Approximately 80% of all parasitoids belong to the order Hymenoptera (mostly wasps) and 20% to the order Diptera (flies) (Heraty, 2017). The Tachinid fly, *Exorista xanthaspis* Wiedemann, indigenous in Cambodia, was found to parasitise larvae of the recently introduced Fall Armyworm (*Spodoptera frugiperda* J. E. Smith) (FAW) in maize crops in Samlout district Battambang province in 2024 (Figure 3). FAW larvae were collected from maize plants in the field and raised through to pupation and emergence of adult moths. In one case, the FAW larva had been parasitised by *E. xanthaspis.* The adult fly laid an egg, circled in red, on the FAW larva (a). The parasitoid larva developed within the FAW larva and the parasitoid pupa (c) exited from the FAW pupa (b). The adult parasitoid fly (d) emerged about two weeks later.

The preservation of natural enemies of insect pests is a fundamental objective of IPM. Firstly, farmers should be taught how to recognise the natural enemies and then they need to learn how to preserve them. The conservation of natural enemies has been greatly assisted by pest-threshold research that has identified pest–crop scenarios where spraying is not warranted or spraying for major primary pests can be delayed until the crop is at lesser risk from secondary pests (Brier et al., 2008).

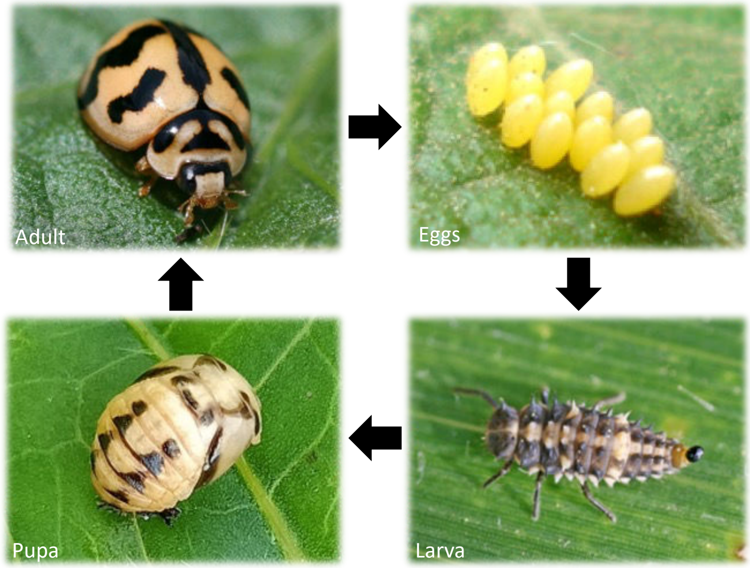

Figure 2. Life cycle of the six-spotted ladybird beetle (*Cheilomenes sexmaculata* Fabricius)

Dunn (2022) studied the social and ecological constraints affecting insect pest management in rice farming systems in Banteay Meanchey and Battambang provinces between 2017 and 2021. The most important Orders for pests of rice were found to be Hemiptera (10 genera), Orthoptera (7), Coleoptera (5) and Lepidoptera (3) Figure 4. In a study of management of bean flower thrips in mungbean, a total of 36 insect pest taxa were found compared with 32 taxa classed as insect predators (Martin et al., 2021). The number of taxa classed as neutral was 14. The most important Orders for pests of mungbean were Hemiptera (15 genera), Lepidoptera (10), Coleoptera (5) and Orthoptera (4) (Figure 4). For rice, the most important Order for pests was Hemiptera. However, the number of Lepidopteran pests was greater for mungbean compared with rice where the number of Orthopteran taxa was greater compared with mungbean crops Figure 4. In mungbean, the most important Orders for insect predators were Coleoptera (12 genera), Hymenoptera (5), Diptera (5), Odonata (4) and Hemiptera (2) Figure 5.

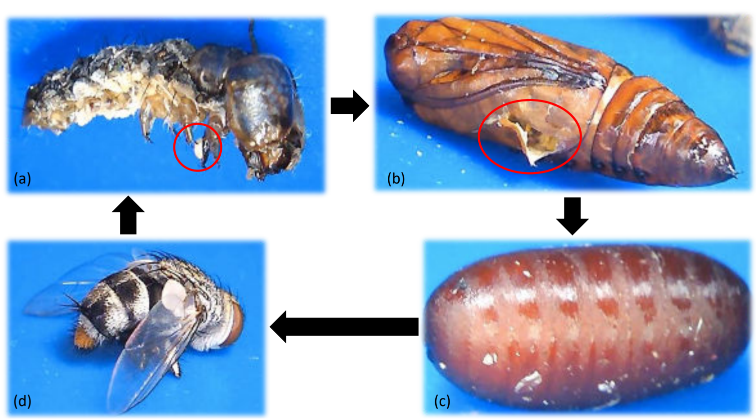

Figure 3. The Tachinid fly, *Exorista xanthaspis*, is a parasitoid of Lepidopteran crop pests including *Spodoptera frugiperda*

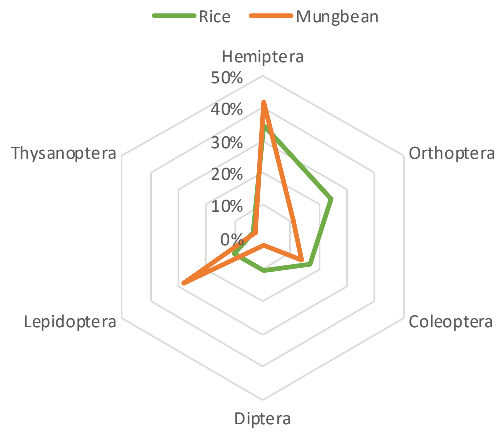

Figure 4. Order of importance of insect orders representing pests in rice compared with mungbean

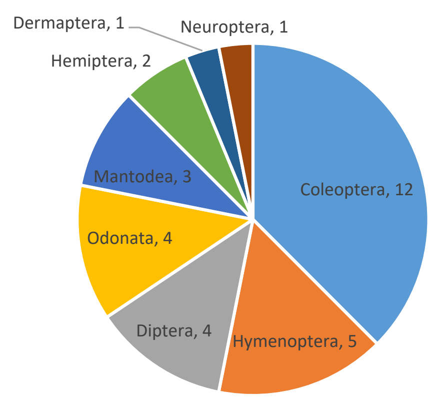

Figure 5. Order of importance of insect orders representing insect predators in mungbean

The purpose of this article was to compile a checklist of arthropods found in Cambodian agricultural systems as a resource for the development of education and training materials for IPM.

METHODS

The first arthropod specimens in this collection were collected in upland crops between 2003 and 2007 in Battambang, Kampong Cham and Tboung Khmum provinces with support from the Australian Centre for International Agricultural Research (ACIAR) project ASEM/2000/109 Table 1. This work resulted in the publication of the ACIAR monograph "Insects of upland crops in Cambodia" (Pol et al., 2010). Subsequent projects were based in Banteay Meanchey, Battambang and Pailin Provinces in North-West Cambodia Figure 6.

The field guide to insects of upland crops in Cambodia (Pol et al., 2010) produced by ACIAR project ASEM/2000/109 was used to identify the bulk of insect species associated with upland crops. The collections extended to lowland rice systems in the ACIAR project CSE/2015/044 which focused on Banteay Meanchey and Battambang Provinces and ran from 2016 to 2021.

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Table 1. Projects and provinces where arthropod specimens were collected

Project	Duration	Provinces
ASEM/2000/049[1]	2003-2007	Battambang, Kampong Cham, Tboung Khmum
ASEM/2006/130[2]	2007-2011	Battambang, Pailin
ASEM/2010/049[3]	2011-2016	Battambang, Pailin
CSE/2015/044[4]	2016-2021	Banteay Meanchey, Battambang

Figure 6. Provinces where the majority of arthropod specimens were collected

The field guide to insects of upland crops in Cambodia (Pol et al., 2010) produced by ACIAR project ASEM/2000/109 was used to identify the bulk of insect species associated with upland crops. The collections extended to lowland rice systems in the ACIAR project CSE/2015/044 which focused on Banteay Meanchey and Battambang Provinces and ran from 2016 to 2021.

Photos of arthropods collected were uploaded for identification to an iNaturalist site (https://www.inaturalist.org/projects/camsid-ipm) set up for the project (https://aciarcambodiasidproject.wordpress.com/). The checklist database also contains hyperlinks to show the current status for each entry (https://www.inaturalist.org/observations/16511510, https://www.inaturalist.org/observations/71353090). Although most of the arthropods of economic importance were identified, a number of insects on the sticky traps could not be identified. The full list of arthropod specimens identified is given in Appendix 1.

RESULTS

The database currently includes 305 entries for arthropods: Arachnida (17) and Insecta (288). Twelve insect Orders are represented with the largest number of insect specimens belonging to Families Lepidoptera (67), Coleoptera (64), Hemiptera (51), Hymenoptera (35) and Diptera (31) Figure . Fifty-three percent of the taxa are considered to be pests and 35% beneficial in that they are predators or parasitoids of insect pests.

[1] https://www.aciar.gov.au/sites/default/files/2022-03/Final%20Report%20for%20ASEM-2000-109.pdf
[2] https://www.aciar.gov.au/sites/default/files/2022-03/Final%20Report%20for%20ASEM-2006-130.pdf
[3] https://www.aciar.gov.au/project/asem-2010-049
[4] https://www.aciar.gov.au/sites/default/files/2023-01/CSE-2015-044-final-report.pdf

Eleven percent of species are considered to be neutral in that they do not directly affect pests. Although bees are considered neutral, they benefit crops as pollinators. Likewise, "neutral" species include species such as dung beetles which have ecosystem functions such as nutrient cycling and parasite suppression. Ninety percent of specimens are identified to genus level and 62% to species level.

Agricultural pests predominantly belong to the insect orders Lepidoptera (67 species), Coleoptera (34), Hemiptera (34), Orthoptera (13) and Diptera (7) Figure 7 whereas natural enemies predominantly belong to the orders Hymenoptera (23 species), Coleoptera (22), Araneae (spiders, 16), Hemiptera (14), Odonata (12), Diptera (10), Mantodea (5) and Neuroptera (2) Figure 8.

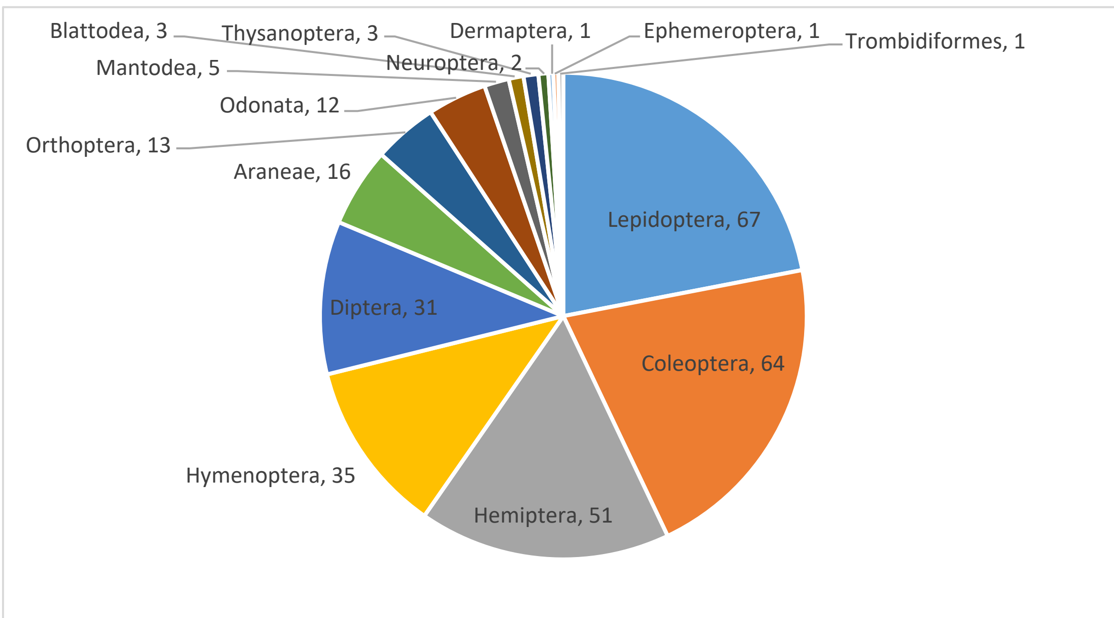

Figure 7. Order of importance of insect orders in agricultural systems in Cambodia

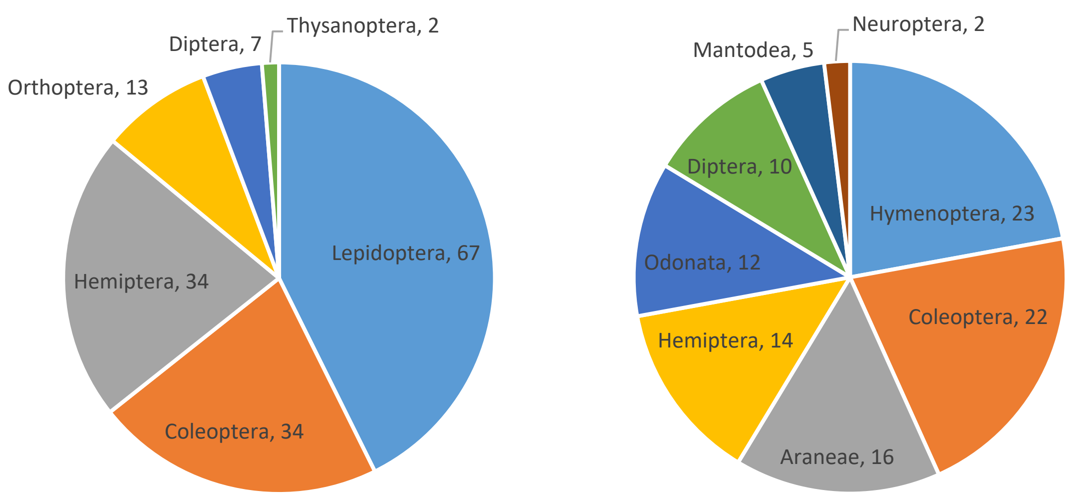

Figure 1 Order of importance of insect orders representing pests (left) and their natural enemies (right)

DISCUSSION

Martin et al. (2020) proposed an IPM strategy for managing insect pests in mungbean where the potential for ecomomic losses is relatively low during the vegetative stage but becomes critical as the crop reaches the reproductive stage, so it is a two-stage strategy:

Vegetative stage

- Avoid using chemical insecticides during the vegetative stage unless leaf loss is greater than 30% or there are 8–9 folded leaves/plant (*Omiodes* spp.).
- Use biological insecticides such as Metabe® (*Metarhizium anisopliae* plus *Beauveria bassiana*) at 15 and again at 25 days after sowing (DAS) to control caterpillars as well as flower thrips.
- Deploy blue sticky traps at 15 DAS and again at 25 DAS to monitor for bean flower thrips and pod-borers.

Reproductive stage

- At 35 DAS, inspect buds and flowers for podborer larvae and pod-sucking insects.
- If economic thresholds are exceeded, apply selective insecticides that do not harm beneficial arthropods such as ladybird beetles.

Implementation of this strategy requires careful checking or "scouting" for insect pest damage in the field as well as for insect pests and their natural enemies present. The determination of economic thresholds help with these management decisions by providing guidance as to whether insect control will have an economic benefit. An economic threshold is the insect pest population level or extent of crop damage at which the value of the crop destroyed exceeds the cost of controlling the pest The purpose of the economic threshold is to prevent a pest population from reaching the point where its damage causes monetary losses that are equal to the cost of control. This "break-even" point is referred to as an economic injury level (EIL) (Seiter, 2018). The economic threshold (ETL) is always set lower than the EIL to provide a lead time before the break-even point is reached and an economic loss occurs. The ETL is the point when an insect control measure is justified. Approximate ETLs for important insect pests of mungbean are given in Figure 9 and this requires the field to be carefully checked for specific pests. The ETL is a useful guide but other considerations should be taken into account such as the risk of crop loss from other factors not related to insect damage, for example, risk of drought or flood. It is also important to record numbers of natural enemies in the field. If populations of natural enemies are high, then the decision to implement control measures could be delayed for a few days before making the final decision. Common natural enemies to look for in mungbean fields are shown in Figure 10. An alternative to field counts, blue and yellow sticky traps, can be used to monitor populations of insect pests and their natural enemies (Martin et al., 2021).

The challenge is to find a way to have the IPM strategy adopted by mungbean growers. Zhang et al. (2020) found that "top-down" methods such as mass media and extension through the local Agricultural Cooperative (AC) were not effective in the transfer of technical information in Angsangsak village which shares the AC with Preaek Trab village. Transfer of technical information in this community is primarily through the village social network. An important element of an effective intervention strategy is to identify who are the key influencers on insect pest management in the community and engage them to facilitate adoption of IPM in crops such as mungbean (Zhang et al., 2020).

Public policy in developing countries has failed to invest in educating farmers on how to deal with variable agro-ecosystems and a changing world (Van Den Berg and Higgins, 2007) who assessed a participatory training approach in changing crop protection by farmers from dependence on chemicals to more sustainable practices in line with IPM and they found the substantial benefits of participation in Farmer Field Schools (FFS). The FFS approach was developed by FAO in Southeast Asia as an alternative to the prevailing top-down extension method of the Green Revolution, which failed to work where more complex and counter-intuitive problems existed with insect pest management. A typical FFS comprises 20-25 farm households who meet in local field settings and under the guidance of a trained facilitator. The FFS observe and compare demonstration plots over the course of an entire cropping season. One plot typically follows local conventional methods while the other is used to demonstrate with what could be considered "best practice". FFS enable the integration of knowledge which requires solving problems by combining farmer knowledge with scientific knowledge to achieve realistic livelihood outcomes.

It is important to ensure that key influencers on insect pest management in the village are invited to participate in FFS. If key influencers happen to be input sellers, it is likely they cannot participate in the FFS because they cannot leave their shop. In such cases, IPM training and resources could be delivered

Order Hemiptera, Family Aphidae (*Aphis crassivora* C. L. Koch)
Damage caused: Aphids amass on growing points where they suck sap from flowers, pods and stems.
ETL: 3 infested growing points per m^2 or 1 infested plant per metre of row.

Order Hemiptera, Family Pentatomidae (*Piezodorus hybneri* Gmelin)
Damage caused: Adults and nymphs pierce and suck developing seeds and pods, which are then lost, deformed or develop dark marks.
ETL: 1 adult equivalent per m^2.

Order Lepidoptera, Family Crambidae (*Omiodes indicata* Fabricius)
Damage caused: Larvae live between two leaves spun together. In later stages they may spin several leaves together, forming a mass of partially eaten leaves.
ETL: 8-9 folded leaves per plant.

Order Lepidoptera, Family Crambidae (*Maruca vitrata* Fabricius)
Damage caused: Flowers may be damaged and discoloured; flower-bud shedding may occur and pod production may be reduced. Pods have small, darkened entry holes on the surface.
ETL: 3 larvae per m^2 or 1 per metre of row.

Order Lepidoptera, Family Noctuidae (*Helicoverpa armigera* Hübner)
Damage caused: Most damage is from feeding on tips, buds, flowers and pods. Larvae will also feed on leaves, but this is not usually significant.
ETL: 3 larvae per m^2 or 1 per metre of row.

Order Thysanoptera, Family Thripidae (*Megalurothrips usitatus* Bagnall)
Damage caused: Infestations begin in buds and flowers and cause abortion of flowers. Large infestations cause poor pod set, stunted plants, and leaves and flowers to wilt.
ETL: 4–6 thrips per flower at flowering and pod setting.

Figure 9. Economic Threshold Levels (ETLs) for important insect pests of mungbean using images extracted from the checklist database

Figure 10. Examples of natural enemies that should be checked for during field inspection, their numbers should be taken into account in the decision to apply insecticide

to input sellers individually. Provision of IPM training to farmers is the first step but there is a need to expand training and and provision of resources to include other actors along the value chain and especially retailers of insecticides. Lack of access to IPM inputs, market constraints, and access to training are among the key barriers to and motivations for adopting IPM strategies (Muriithi et al., 2024). Firstly, retailers need to be encouraged to stock relevant IPM tools such as pheromone traps, sticky traps and bio-pesticides. In addition, this Arthropod Database can be used as a resource for the preparation of banners, posters and leaflets for display at retail outlets. The challenge is to find retail outlets that are interested to market IPM products to gain a commercial advantage.

The majority of the arthropod photos were taken as part of projects funded by the Australian Centre for International Agricultural Research (ACIAR) between 2003 and 2021. Photographs taken for the ACIAR insect guide (Pol et al. 2010) were taken by Kelly Baker, Rowena Eastick, Robin Gunning, Wes Leedham, Robert Martin, Stephanie Montgomery, Adrian Nicholas, Chanthy Pol, Fiona Scott and Tanya Smith. Photographs taken for the mungbean study (Martin et al., 2021) were taken by Chariya Korn, Robert Martin, Samnang Pheng, Ratha Rien, Sopha Yous and Sophea Yous. Photographs of arthropods in rice systems were taken by Lucinda Dunn (Dunn, 2022).

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Appendix 1. Checklist of arthropods found in cropping systems of Cambodia

Class	Order	Family	*Genus*	*Species*	Common name
Arachnida	Araneae	Araneidae	*Argiope*	*aemula*	St Andrew's cross spider
Arachnida	Araneae	Araneidae	*Argiope*	*catenulata*	Grass cross spider
Arachnida	Araneae	Araneidae	*Neoscona*	*theisi*	Spotted orb-weavers
Arachnida	Araneae	Clubionidae	*Clubiona*		Leafcurling sac spiders
Arachnida	Araneae	Lycosidae	*Hippasa*		Funnel web wolf spiders
Arachnida	Araneae	Lycosidae	*Pardosa*	*pseudoannulata*	Wolf spiders
Arachnida	Araneae	Oxyopidae	*Oxyopes*	*javanus*	Lynx spiders
Arachnida	Araneae	Oxyopidae	*Oxyopes*	*macilentus*	Lynx spiders
Arachnida	Araneae	Oxyopidae	*Peucetia*		Lynx spiders
Arachnida	Araneae	Pholcidae	*Crossopriza*	*lyoni*	Tailed cellar spider
Arachnida	Araneae	Salticidae	*Plexippus*	*petersi*	Common housefly catcher
Arachnida	Araneae	Sparassidae	*Heteropoda*		Giant huntsman spiders
Arachnida	Araneae	Tetragnathidae	*Tetragnatha*		Stretch spiders
Arachnida	Araneae	Tetragnathidae	*Tetragnatha*		Stretch spiders
Arachnida	Araneae	Thomisidae	*Runcinia*		Grass crab spiders
Arachnida	Araneae	Thomisidae	*Runcinia*		Grass crab spiders
Arachnida	Trombidiformes	Tetranychidae			Spider mites
Insecta	Blattodea	Ectobiidae	*Blattella*	*asahinai*	Asian cockroach
Insecta	Blattodea	Ectobiidae	*Hemithyrsocera*	*vittata*	Wood cockroaches
Insecta	Blattodea	Temitidae	*Macrotermes*	*gilvus*	Higher termites
Insecta	Coleoptera	Anthribidae	*Araecerus*	*fasciculatus*	Coffee bean weevil
Insecta	Coleoptera	Bostrichidae	*Rhyzopertha*	*dominica*	Lesser grain borer
Insecta	Coleoptera	Brentidae	*Cylas*		Sweet potato weevils
Insecta	Coleoptera	Carabidae	*Pheropsophus*		Ground beetle

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Coleoptera	Carabidae	*Zuphium*	*olens*	Ground beetle
Insecta	Coleoptera	Carabidae	*Chlaenius*	*circumdatus*	Vivid metalic ground beetles
Insecta	Coleoptera	Carabidae	*Stenolophus*		Seedcorn beetles
Insecta	Coleoptera	Carabidae	*Chlaenius*	*thieleni*	Vivid metalic ground beetles
Insecta	Coleoptera	Carabidae	*Lebia*	*circumdata*	Colorful Foliage Ground Beetles
Insecta	Coleoptera	Carabidae			Pedunculate Ground beetle
Insecta	Coleoptera	Carabidae	*Scarites*		Ground beetle
Insecta	Coleoptera	Carabidae			Tiger beetles
Insecta	Coleoptera	Carabidae	*Ophionea*	*nigrofasciata*	Long-neck ground beetle
Insecta	Coleoptera	Cerambycidae			Flat-faced Longhorn Beetles
Insecta	Coleoptera	Cerambycidae	*Perissus*		Longhorn beetle
Insecta	Coleoptera	Cerambycidae	*Aristobia*	*approximator*	Longhorn beetle
Insecta	Coleoptera	Cerambycidae			Flat-faced Longhorn Beetles
Insecta	Coleoptera	Cerambycidae	*Coptops*		Longhorn beetle
Insecta	Coleoptera	Cerambycidae	*Euryphagus*	*lundii*	Longhorn beetle
Insecta	Coleoptera	Cerambycidae	*Batocera*	*rufomaculata*	Mango stem borer
Insecta	Coleoptera	Chrysomelidae	*Cassida*	*circumdata*	Tortoise beetle
Insecta	Coleoptera	Chrysomelidae	*Dicladispa*	*armigera*	
Insecta	Coleoptera	Chrysomelidae	*Aspidimorpha*	*furcata*	Furcated tortoise beetle
Insecta	Coleoptera	Chrysomelidae	*Aulacophora*	*flavomarginata*	Black-back cucumber beetle
Insecta	Coleoptera	Chrysomelidae	*Aulacophora*	*indica*	Cucumber beetle
Insecta	Coleoptera	Chrysomelidae	*Monolepta*	*signata*	Monolepta beetle
Insecta	Coleoptera	Chrysomelidae	*Phyllotreta*		Striped flea beetle
Insecta	Coleoptera	Chrysomelidae			Leaf beetle
Insecta	Coleoptera	Chrysomelidae	*Podontia*	*affinis*	Flea beetle
Insecta	Coleoptera	Chrysomelidae	*Callosobruchus*	*maculatus*	Cowpea bruchid

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Coleoptera	Chrysomelidae	*Medythia*	*nigrobilineata*	Two-striped leaf beetle
Insecta	Coleoptera	Coccinellidae	*Cheilomenes*	*sexmaculata*	Six-spotted zigzag ladybird
Insecta	Coleoptera	Coccinellidae	*Coccinella*	*transversalis*	Small transverse ladybird
Insecta	Coleoptera	Coccinellidae	*Harmonia*	*octomaculata*	Maculate ladybird
Insecta	Coleoptera	Coccinellidae	*Micraspis*	*discolor*	Discolored lady beetle
Insecta	Coleoptera	Coccinellidae	*Henosepilachna*	*vigintioctopunctata*	Hadda beetle
Insecta	Coleoptera	Coccinellidae	*Synonycha*	*grandis*	Giant bamboo ladybird
Insecta	Coleoptera	Curculionidae	*Sitophilus*	*oryzae*	Rice weevil
Insecta	Coleoptera	Curculionidae	*Sitophilus*	*zeamais*	Maize weevil
Insecta	Coleoptera	Curculionidae	*Hypomeces*	*pulviger*	Gold-dust weevil
Insecta	Coleoptera	Curculionidae	*Xanthochelus*	*faunus*	Putrea weevil
Insecta	Coleoptera	Curculionidae	*Atactogaster*	*zebra*	Root-feeding weevils
Insecta	Coleoptera	Curculionidae	*Euplatypus*	*parallelus*	Ambrosia beetle
Insecta	Coleoptera	Dytiscidae	*Copelatus*		Diving beetles
Insecta	Coleoptera	Elateridae			Click beetles
Insecta	Coleoptera	Erotylidae			Pleasing fungus beetles
Insecta	Coleoptera	Heteroceridae	*Augyles*		Variegated mud-loving beetles
Insecta	Coleoptera	Hydrophilidae	*Berosus*		Water scavenger beetle
Insecta	Coleoptera	Hydrophilidae	*Sternolophus*	*rufipes*	Water scavenger beetles
Insecta	Coleoptera	Hydrophilidae	*Hydrophilus*		Giant water scavenger beetle
Insecta	Coleoptera	Laemophloeidae	*Cryptolestes*		Lined flat bark beetles
Insecta	Coleoptera	Laemophloeidae			Lined flat bark beetles
Insecta	Coleoptera	Lampyridae	*Asymmetricata*		Firefles
Insecta	Coleoptera	Melyridae	*Intybia*		Soft-winged flower beetles
Insecta	Coleoptera	Scarabaeidae	*Protaetia*	*acuminata*	Mango flower beetle
Insecta	Coleoptera	Scarabaeidae	*Oryctes*	*rhinoceros*	Coconut rhinoceros beetle

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Coleoptera	Scarabaeidae	*Anomala*	*antiqua*	Groundnut chafer
Insecta	Coleoptera	Scarabaeidae	*Onitus*		Dung beetle
Insecta	Coleoptera	Scarabaeidae	*Adoretus*	*sinicus*	Chinese rose beetle
Insecta	Coleoptera	Scarabaeidae	*Xylotrupes*	*socrates*	Siamese rhinoceros beetle
Insecta	Coleoptera	Scirtidae	*Exochomoscirtes*		Marsh beetle
Insecta	Coleoptera	Scirtidae	*Ora*		Flea marsh beetles
Insecta	Coleoptera	Staphylinidae	*Paederus*		Whiplash beetles
Insecta	Coleoptera	Staphylinidae	*Anotylus*		Spiny-legged rove beetles
Insecta	Dermaptera	Forficulidae	*Proreus*	*simulans*	Black earwigs
Insecta	Diptera	Agromyzidae	*Liriomyza*	*sativae*	Vegetable leaf miner
Insecta	Diptera	Agromyzidae	*Ophiomyia*	*phaseoli*	Bean fly
Insecta	Diptera	Asilidae			Robber flies
Insecta	Diptera	Calliphoridae	*Chrysomya*	*megacephala*	Oriental latrine fly
Insecta	Diptera	Chironomidae			Non-biting midges
Insecta	Diptera	Culicidae			Culicine mosquitos
Insecta	Diptera	Culicidae	*Aedes*		Mosquito
Insecta	Diptera	Dolichopodidae	*Chrysosoma*		Long-legged flies
Insecta	Diptera	Drosophilidae	*Drosophila*		Small fruit fly
Insecta	Diptera	Ephydridae	*Hydrellia*	*philippina*	Rice whorl maggot
Insecta	Diptera	Ephydridae			Shore flies
Insecta	Diptera	Fanniidae	*Fannia*		Lesser house flies and allies
Insecta	Diptera	Lauxaniidae	*Sapromyza*		Lauxaniid flies
Insecta	Diptera	Milichiidae			Freeloader flies
Insecta	Diptera	Muscidae	*Lispe*		True flies
Insecta	Diptera	Muscidae	*Musca*	*domestica*	Common house fly
Insecta	Diptera	Mycetophilidae			Fungus gnats

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Diptera	Phoridae			Humpbacked Flies
Insecta	Diptera	Sarcophagidae			Flesh flies
Insecta	Diptera	Stratiomyidae	*Microchrysa*		Soldier flies
Insecta	Diptera	Syrphidae	*Eristalinus*		Lagoon Flies
Insecta	Diptera	Syrphidae	*Mesembrius*		Rat-tail Maggot Flies
Insecta	Diptera	Syrphidae	*Episyrphus*		Hoverflies
Insecta	Diptera	Syrphidae	*Allograpta*		Streak flies
Insecta	Diptera	Tabanidae	*Chrysops*	*dispar*	Deer flies
Insecta	Diptera	Tachinidae	*Trichopoda*		Feather-legged flies
Insecta	Diptera	Tachinidae	*Exorista*	*xanthaspis*	Tachinid flies
Insecta	Diptera	Tephritidae	*Carpomya*	*vesuviana*	Putrea fruit fly
Insecta	Diptera	Tephritidae	*Bactrocera*	*cucurbitae*	Melon fly
Insecta	Diptera	Tephritidae	*Bactrocera*	*dorsalis*	Oriental fruit fly
Insecta	Diptera	Tipuloidea	*Trentepohlia*		Limoniid crane flies
Insecta	Ephemeroptera	Caenidae	*Caenis*		Mayfly
Insecta	Hemiptera	Delphacidae	*Nilaparvata*	*lugens*	Brown planthopper
Insecta	Hemiptera	Delphacidae	*Sogatella*	*furcifera*	White-backed planthopper
Insecta	Hemiptera	Aleyrodidae	*Bemisia*	*tabaci*	Silverleaf whitefly
Insecta	Hemiptera	Alydidae	*Riptortus*	*linearis*	Brown bean bug
Insecta	Hemiptera	Alydidae	*Leptocorisa*		Rice bug
Insecta	Hemiptera	Aphididae	*Aphis*	*craccivora*	Cowpea aphid
Insecta	Hemiptera	Aphididae	*Aphis*	*glycines*	Soybean aphid
Insecta	Hemiptera	Aphididae	*Aphis*	*gossypii*	Cotton aphid
Insecta	Hemiptera	Aphididae	*Rhopalosiphum*	*maidis*	Corn leaf aphid
Insecta	Hemiptera	Blissidae	*Dimorphopterus*	*rondoni*	Chinch bugs
Insecta	Hemiptera	Cicadellidae	*Bothrogonia*	*ferruginea*	Black-tipped leafhopper

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Hemiptera	Cicadellidae	*Maiestas*	*dorsalis*	Zig-zag winged leafhopper
Insecta	Hemiptera	Cicadellidae	*Nephotettix*	*nigropictus*	Green rice leafhopper
Insecta	Hemiptera	Cicadellidae	*Nephotettix*	*virescens*	Green paddy leafhopper
Insecta	Hemiptera	Cicadellidae			Leafhopper
Insecta	Hemiptera	Cicadellidae			Leafhopper
Insecta	Hemiptera	Cicadellidae	*Cofana*	*spectra*	White rice leaf hopper
Insecta	Hemiptera	Cimicoidea			Cimicomorph bugs
Insecta	Hemiptera	Coreidae	*Anoplocnemis*	*phasiana*	Leaf-footed bugs
Insecta	Hemiptera	Coreidae	*Cletus*	*bipunctatus*	Spined legume bug
Insecta	Hemiptera	Crabronidae	*liris*	*subtessellatus*	Predatory wasp
Insecta	Hemiptera	Cydnidae			Burrowing bugs
Insecta	Hemiptera	Derbidae	*Proutista*	*moesta*	Palm derbid
Insecta	Hemiptera	Flatidae	*Salurnis*		Fulgoroid planthoppers
Insecta	Hemiptera	Geocoridae	*Geocoris*		Big-eyed bugs
Insecta	Hemiptera	Gerridae	*Gerris*		Water striders
Insecta	Hemiptera	Lygaeidae	*Kleidocerys*		Seed bugs
Insecta	Hemiptera	Lygaeidae	*Graptostethus*	*servus*	Seed bugs
Insecta	Hemiptera	Lygaeidae	*Melanotelus*		Seed bugs
Insecta	Hemiptera	Micronectidae	*Micronecta*		Pygmy Water Boatmen
Insecta	Hemiptera	Miridae	*Nesiodiocoris*		Mirid
Insecta	Hemiptera	Nabidae	*Prostemma*	*fasciatum*	Damsel bugs
Insecta	Hemiptera	Pentatomidae	*Eysarcoris*	*guttigerus*	Two-spotted Sesame Bug
Insecta	Hemiptera	Pentatomidae	*Megymenum*		Stink bug
Insecta	Hemiptera	Pentatomidae	*Nezara*	*viridula*	Green vegetable bug
Insecta	Hemiptera	Pentatomidae	*Piezodorus*	*hybneri*	Red-banded shield bug
Insecta	Hemiptera	Pentatomidae	*Plautia*		Green stink bug

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Hemiptera	Pentatomidae	*Eocanthecona*	*furcellata*	Predatory shield bug
Insecta	Hemiptera	Plataspidae	*Brachyplatys*		Black bean bug
Insecta	Hemiptera	Pseudococcidae	*Paracoccus*	*marginatus*	Papaya mealybug
Insecta	Hemiptera	Pyrrhocoridae	*Dysdercus*	*cingulatus*	Indian cotton stainer
Insecta	Hemiptera	Pyrrhocoridae	*Physopelta*	*slanbuschii*	Red and Bordered Plant Bugs
Insecta	Hemiptera	Reduviidae	*Coranus*		Assassin bugs
Insecta	Hemiptera	Reduviidae	*Rhynocoris*	*fuscipes*	Assassin bugs
Insecta	Hemiptera	Reduviidae	*Peirates*	*atromaculatus*	Corsair bugs
Insecta	Hemiptera	Reduviidae	*Sirthenea*	*flavipes*	Corsair bugs
Insecta	Hemiptera	Reduviidae	*Polytoxus*		Assassin bugs
Insecta	Hemiptera	Reduviidae			Assassin bugs
Insecta	Hemiptera	Reduviidae	*Ectomocoris*	*biguttulus*	Corsair bugs
Insecta	Hemiptera	Reduviidae	*Ectrychotes*	*andreae*	Millipede assassin bugs
Insecta	Hemiptera	Rhyparochromidae	*Horridipamera*	*nietneri*	Dirt-coloured seed bugs
Insecta	Hymenoptera	Ichneumonidae			Ichneumonid Wasps
Insecta	Hymenoptera	Ichneumonidae	*Xanthopimpla*	*flavolineata*	Ichneumonid Wasps
Insecta	Hymenoptera	Apidae	*Apis*	*cerana*	Asian honey bee
Insecta	Hymenoptera	Apidae	*Apis*	*dorsata*	Giant honey bee
Insecta	Hymenoptera	Apidae	*Apis*	*florea*	Dwarf honey bee
Insecta	Hymenoptera	Apidae	*Ceratinidia*		Striped small carpenter bees
Insecta	Hymenoptera	Apidae	*Pithitis*		Small carpenter bee
Insecta	Hymenoptera	Apidae	*Xylocopa*	*minor*	Yellow sided carpenter bee
Insecta	Hymenoptera	Braconidae	*Apanteles*	*cypris*	
Insecta	Hymenoptera	Braconidae	*Biosteres*	*arisanus*	Braconid wasp
Insecta	Hymenoptera	Braconidae	*Cotesia*	*yakutatensis*	Braconid wasp
Insecta	Hymenoptera	Crabronidae	*Astata*		Predatory wasp

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Hymenoptera	Crabronidae	*Liris*		Predatory wasp
Insecta	Hymenoptera	Crabronidae	*Trypoxylon*		Predatory wasp
Insecta	Hymenoptera	Cydnidae	*Macroscytus*		Burrower bugs
Insecta	Hymenoptera	Eulophidae	*Elasmus*		Chalcidoid Wasps
Insecta	Hymenoptera	Formicidae	*Aenictus*		Aenictus Army Ants
Insecta	Hymenoptera	Formicidae			Lasiinine ants
Insecta	Hymenoptera	Formicidae	*Oecophylla*	*smaragdina*	Asian weaver ant
Insecta	Hymenoptera	Formicidae	*Solenopsis*	*geminata*	Tropical fire ant
Insecta	Hymenoptera	Formicidae	*Pheidole*		Big-headed ants
Insecta	Hymenoptera	Formicidae	*Odontoponera*	*denticulata*	Ant
Insecta	Hymenoptera	Halictidae	*Lipotriches*	*ceratina*	Red-waisted Grass-Nomia
Insecta	Hymenoptera	Halictidae	*Nomia*		Nomiline bees
Insecta	Hymenoptera	Megachilidae	*Megachile*		Sculptured resin bee
Insecta	Hymenoptera	Pompilidae			Spider wasps
Insecta	Hymenoptera	Pteromalidae	*Pteromalus*	*cerealellae*	Pteromalid Wasps
Insecta	Hymenoptera	Pteromalidae			Pteromalid Wasps
Insecta	Hymenoptera	Pteromalidae	*Trichomalopsis*	*apanteloctena*	Ectoparasitoid
Insecta	Hymenoptera	Scelionidae	*Trissolcus*	*basalis*	Pentatomid egg parasitoid
Insecta	Hymenoptera	Vespidae	*Polistes*	*olivaceus*	Yellow oriental paper wasp
Insecta	Hymenoptera	Vespidae	*Ropalidia*	*marginata*	Old world paper wasps
Insecta	Hymenoptera	Vespidae	*Polistes*	*brunus*	Predatory wasps
Insecta	Hymenoptera	Vespidae	*Ropalidia*	*stigma*	Tropical paper wasp
Insecta	Hymenoptera	Vespidae	*Vespa*	*tropica*	Greater banded hornet
Insecta	Lepidoptera	Bombycidae	*Trilocha*	*varians*	Greenish silk-moth
Insecta	Lepidoptera	Crambidae	*Scirpophaga*	*incertulas*	Rice yellow stem borer
Insecta	Lepidoptera	Crambidae	*Antigastra*	*catalaunalis*	Sesame leaf roller

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Lepidoptera	Crambidae	*Chilo*	*suppressalis*	Striped rice stem borer
Insecta	Lepidoptera	Crambidae	*Cnaphalocrocis*	*medinalis*	Rice leaf roller
Insecta	Lepidoptera	Crambidae	*Conogethes*	*punctiferalis*	Durian fruit borer
Insecta	Lepidoptera	Crambidae	*Diaphania*	*indica*	Cucumber moth
Insecta	Lepidoptera	Crambidae	*Herpetogramma*		Moth
Insecta	Lepidoptera	Crambidae	*Omiodes*	*diemenalis*	Bean leafroller
Insecta	Lepidoptera	Crambidae	*Omiodes*	*indicata*	Bean-leaf webworm
Insecta	Lepidoptera	Crambidae	*Ostrinia*	*furnacalis*	Asian cornborer
Insecta	Lepidoptera	Crambidae	*Parapoynx*	*stagnalis*	Rice case moth
Insecta	Lepidoptera	Crambidae	*Spoladea*	*recurvalis*	Beet webworm
Insecta	Lepidoptera	Erebidae	*Asota*	*producta*	Snouted Tiger Moths
Insecta	Lepidoptera	Erebidae	*Amata*	*sperbius*	Wasp Moths
Insecta	Lepidoptera	Erebidae	*Creatonotos*	*gangis*	Baphomet moth
Insecta	Lepidoptera	Erebidae	*Psichotoe*	*duvaucelii*	Wasp Moths
Insecta	Lepidoptera	Erebidae	*Utetheisa*	*lotrix*	Crotalaria moth
Insecta	Lepidoptera	Erebidae	*Hypocala*		Moth
Insecta	Lepidoptera	Erebidae	*Dasychira*		Tussock moths
Insecta	Lepidoptera	Erebidae	*Anomis*		Semilooper
Insecta	Lepidoptera	Erebidae	*Achaea*	*janata*	Castor semilooper
Insecta	Lepidoptera	Erebidae	*Buzara*	*onelia*	Moth
Insecta	Lepidoptera	Erebidae	*Euproctis*		Tussock moths
Insecta	Lepidoptera	Erebidae	*Miltochrista*		Moth
Insecta	Lepidoptera	Erebidae	*Platyja*		Moth
Insecta	Lepidoptera	Erebidae	*Trigonodes*	*hyppasia*	Triangles Semilooper
Insecta	Lepidoptera	Eupterotidae	*Eupterote*	*lineosa*	Monkey moth
Insecta	Lepidoptera	Gelechiidae	*Aproaerema*	*modicella*	Peanut leaf miner

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Lepidoptera	Gelechiidae	*Sitotroga*	*cerealella*	Angoumois grain moth
Insecta	Lepidoptera	Gelechiidae	*Stomopteryx*	*subsecivella*	Soybean leaf miner
Insecta	Lepidoptera	Geometridae	*Scopula*		Moth
Insecta	Lepidoptera	Hesperiidae			Grass skippers
Insecta	Lepidoptera	Hesperiidae	*Erionata*	*thrax*	Banana skipper
Insecta	Lepidoptera	Hesperiidae	*Pelopidas*	*mathias*	Rice skipper
Insecta	Lepidoptera	Hesperiidae	*Potanthus*	*ganda*	Ganda dart
Insecta	Lepidoptera	Hesperiidae	*Taractrocera*	*archias*	Yellow Grass Dart
Insecta	Lepidoptera	Hesperiidae	*Telicota*	*colon*	Pale palm dart
Insecta	Lepidoptera	Lasiocampidae	*Streblote*		Lappet moths
Insecta	Lepidoptera	Lycaenidae	*Cigaritis*	*lohita*	Long-banded silverline
Insecta	Lepidoptera	Lycaenidae	*Castalius*	*rosimon*	Common perriot
Insecta	Lepidoptera	Lycaenidae	*Euchrysops*	*cnejus*	Gram blue butterfly
Insecta	Lepidoptera	Lycaenidae	*Lampides*	*boeticus*	Pea blue butterfly
Insecta	Lepidoptera	Lycaenidae	*Zizina*	*otis*	Lesser grass blue
Insecta	Lepidoptera	Noctuidae	*Agrotis*	*ipsilon*	Black cutworm
Insecta	Lepidoptera	Noctuidae	*Bastilla*	*arcuata*	Arcuate Passenger Moth
Insecta	Lepidoptera	Noctuidae	*Chrysodeixis*	*eriosoma*	Green garden looper
Insecta	Lepidoptera	Noctuidae	*Helicoverpa*	*armigera*	Cotton bollworm
Insecta	Lepidoptera	Noctuidae	*Pindara*	*illibata*	Moth
Insecta	Lepidoptera	Noctuidae	*Spodoptera*	*litura*	Cluster caterpillar
Insecta	Lepidoptera	Noctuidae	*Spodoptera*	*exigua*	Beet armyworm
Insecta	Lepidoptera	Noctuidae	*Spodoptera*	*frugiperda*	Fall armyworm
Insecta	Lepidoptera	Nymphalidae	*Danaus*	*genutia*	Common tiger
Insecta	Lepidoptera	Nymphalidae	*Melanitis*	*leda*	Rice butterfly
Insecta	Lepidoptera	Papilionidae	*Pachliopta*	*aristolochiae*	Common rose

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Lepidoptera	Papilionidae	*Papilio*	*demoleus*	Swallowtail butterfly
Insecta	Lepidoptera	Papilionidae	*Triodes*		Common birdwing
Insecta	Lepidoptera	Pieridae	*Pareronia*	*hippia*	Indian wanderer
Insecta	Lepidoptera	Pyralidae	*Etiella*	*zinckenella*	Pulse podborer
Insecta	Lepidoptera	Pyralidae	*Maruca*	*vitrata*	Maruca podborer
Insecta	Lepidoptera	Sphingidae	*Acherontia*	*styx*	Death's head hawk-moth
Insecta	Lepidoptera	Sphingidae	*Agrius*	*convolvuli*	Convolvulus hawk-moth
Insecta	Lepidoptera	Sphingidae	*Theretra*	*clotho*	Common hunter hawk moth
Insecta	Lepidoptera	Sphingidae	*Theretra*	*nessus*	Yam hawk moth
Insecta	Lepidoptera	Tortricidae	*Cochylis*		Moth
Insecta	Lepidoptera	Tortricidae	*Cryptophlebia*	*peltastica*	Pod moth
Insecta	Lepidoptera	Zygaenidae	*Thyrassia*		Forester moths
Insecta	Mantodea	Hymenopodidae	*Creobroter*	*gemmatus*	Flower mantis
Insecta	Mantodea	Mantidae	*Mantis*	*religiosa*	European mantis
Insecta	Mantodea	Mantidae	*Tenodera*		Mantis
Insecta	Mantodea	Mantidae	*Hierodula*	*patellifera*	Giant Asian mantis
Insecta	Mantodea	Toxoderidae	*Heterochaetula*		Toxoderid mantises
Insecta	Neuroptera	Chrysopidae	*Chrysoperla*		Green lacewings
Insecta	Neuroptera	Hemerobiidae			Brown lacewings
Insecta	Odonata	Coenagrionidae	*Aciagrion*		Slims
Insecta	Odonata	Coenagrionidae	*Agriocnemis*	*pygmaea*	Wandering midget
Insecta	Odonata	Coenagrionidae	*Ceriagrion*	*auranticum*	Orange-tailed sprite
Insecta	Odonata	Coenagrionidae	*Ischnura*	*senegalensis*	Common bluetail
Insecta	Odonata	Libellulidae	*Brachythemis*	*contaminata*	Ditch Jewel
Insecta	Odonata	Libellulidae	*Crocothemis*	*servilia*	Scarlet skimmer
Insecta	Odonata	Libellulidae	*Diplacodes*	*nebulosa*	Black-tipped percher

Class	Order	Family	*Genus*	*Species*	Common name
Insecta	Odonata	Libellulidae	*Diplacodes*	*trivialis*	Chalky percher
Insecta	Odonata	Libellulidae	*Neurothemis*	*tullia*	Pied paddy skimmer
Insecta	Odonata	Libellulidae	*Trithemis*	*pallidinervis*	Dancing dropwing
Insecta	Odonata	Platycnemididae	*Onychargia*	*atrocyana*	Marsh dancer
Insecta	Odonata	Zygoptera	*Ischnura*	*rubilio*	Western golden dartlet
Insecta	Orthoptera	Tettigoniidae			Coneheads
Insecta	Orthoptera	Acrididae	*Aiolopus*	*thalassinus*	Slender Green-winged Grasshopper
Insecta	Orthoptera	Acrididae	*Locusta*	*migratoria*	Migratory locust
Insecta	Orthoptera	Acrididae	*Gesonula*	*mundata*	Common Gesonula
Insecta	Orthoptera	Acrididae	*Oxya*	*hyla intricata*	Rice grasshoppers
Insecta	Orthoptera	Acrididae	*Acrida*		Slantface grasshoppers
Insecta	Orthoptera	Acrididae	*Cyrtacanthacris*	*tatarica*	Brown-spotted locust
Insecta	Orthoptera	Gryllidae	*Acheta*	*domesticus*	House cricket
Insecta	Orthoptera	Gryllidae	*Anaxipha*		Katydids
Insecta	Orthoptera	Gryllotalpidae	*Gryllotalpa*		Mole crickets
Insecta	Orthoptera	Pyrgomorphidae	*Atractomorpha*		Gaudy grasshoppers
Insecta	Orthoptera	Tettigoniidae	*Onomarchus*	*uninotatus*	True katydids
Insecta	Orthoptera	Tettigoniidae	*Conocephalus*	*maculatus*	Spotted Meadow Katydid
Insecta	Thysanoptera	Phlaeothripidae			Tube-tailed thrips
Insecta	Thysanoptera	Thripidae	*Megalurothrips*	*usitatus*	Bean flower thrip
Insecta	Thysanoptera	Thripidae	*Thrips*	*tabaci*	Onion thrip

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References should be quoted as follows:-
Carter and Zeck (1937) or (Carter and Zeck 1937) and listed:-
Carter, H.J. and Zeck, E.H. (1937). A monograph of Australian Colydiidae.
Proceedings of the Linnean Society of New South Wales. **62**: 181-208. Periodical titles should not be abbreviated.

For general information on the writing of scientific papers, reference may be made to the guides on the subject published by the CSIRO Melbourne.

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