

Ways to monitor arthropod activity on native insectary plants

Winegrape growers can assess the performance of native insectary plants on their properties

Mary Retallack, from The University of Adelaide and Retallack Viticulture, describes methods for assessing arthropods that may be found in association with native plants and vineyards. This is the fifth in a series of articles that provides practical insights for growers.

Introduction

In last issue's article ('The functional diversity of predator arthropods in vineyards', January 2019), I discussed some of the groups of natural enemies commonly found in association with selected native insectary plants. They have the capacity to nourish and support the presence of predatory arthropods throughout the year. Native plants are preferred as supplementary flora as they are naturally adapted to Australia's often hot and dry climatic conditions (Danne *et al.* 2010, Pandey *et al.* 2018). They are regularly reported as having a low occurrence of pests (Parry *et al.* 2015) and a high occurrence of natural enemies (Gurr *et al.* 2017, Gagic *et al.* 2018). It is recommended that selected plant species are trialled to assess their capacity to provide insectary benefits on each site. One of the ways to do this is to monitor the arthropods found in association with them. Thomson *et al.* (2007) suggest that monitoring the presence of predatory arthropods, which have a direct impact on pest abundance, can also be used as a way to assess the benefits of enhancing biodiversity.

In this article, I will highlight common ways that winegrape growers can sample arthropods, and considerations when using each approach. Visual monitoring

is a useful way to look for arthropod activity, but it can be difficult to see well-camouflaged insects and spiders in dense foliage, so they may be overlooked as a result. It is important to use sampling techniques that are suited for each plant type to ensure a representative sample is found. I discuss a selection of sampling techniques here.

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Sampling techniques

A representative number of samples should be collected from plants with any selected technique. For all techniques, the captured arthropods can be viewed non-destructively by looking through the clear plastic container (Figure 1a).

Key messages

- It is difficult visually observe arthropods in dense canopies and important species may be missed when scouting.
- There is growing interest about the use of native insectary plants to support populations of natural enemies, which contribute to biological control of pests.
- Sampling techniques tailored to each plant species can be used to determine which arthropods are found in association with them.
- Monitoring arthropods gives wine grape growers the confidence to assess the performance of native insectary plants on their own properties.

Alternatively, they can be transferred to a killing jar (Figure 1b), or freezer so they can then be identified under a microscope (Figure 1c).



Figure 1. Captured arthropods in a plastic container (a), killing jar (b), arthropod samples are sorted from plant debris and then identified (c).

A killing jar is commonly used to immobilise arthropods (Schauff 1986). It consists of a glass jar sealed with a metal screw cap. A layer of plaster of Paris poured in the base of the jar provides a substrate to absorb the killing agent, ethyl acetate. The vapour immobilises the arthropods. Crumpled paper is used to soak up any excess moisture and inhibit damage to the specimens inside the jar. Reference specimens can be photographed, mounted and preserved for future reference.

Modified beat net

Arthropod samples can be collected from grapevines by firmly striking the cordon with a rubber mallet (Figure 2a) over a beat net (Irvin 2010) fashioned around a card table frame (Figure 2b), which holds a funnel and plastic collection container (Figure 2c).

Modified sweep net

Arthropods found in association with native insectary shrubs can be collected by firmly shaking the foliage inside an insect sweep net (Figure 3a), which has been modified to hold a funnel and a collection container (Figure 3b), and then immobilised in the field (Figure 3c).

Pitfall trap

Pitfall traps charged with a killing and preserving agent are commonly used in the studies of ground-dwelling arthropods (Greenslade and Greenslade 1971). Spiders and beetles are often collected, as well as parasitic wasps (Thomson and Hoffmann 2007). The traps can be fashioned from a range of materials including a narrow vial or wider container (Figure 4a). Containers that fit inside a sleeve, flush with the

soil surface, are often used (Majer 1978). Ensure containers have a mesh cover to limit bycatch (Schauff 1986). A cover should be placed above each trap to exclude rainfall. Charge the traps with propylene glycol, or a saline solution, to a height of approximately 30mm (Figure 4b). Propylene glycol is preferred instead of ethylene glycol (also used in antifreeze) (Thomas 2008), which can prove harmful to animals if the solution is accidentally ingested. Pitfall traps can be left in the field for up to two weeks. Then the contents can be screened to extract the arthropods, and decanted into 70-80% ethanol for storage prior to identification (Figure 4c).

Other sampling techniques

Mechanical vacuums and sticky traps are two other sampling techniques to consider. ▶



Figure 2. A rubber mallet is used to strike the cordon to dislodge the arthropods (a), the net is placed below the cordon (b), and insects are collected in a plastic container positioned below the funnel (c).



Figure 3. Foliage is placed inside the modified sweep net (a), the captured arthropods fall to the bottom of the net (b), and can then be immobilised in the field.



Figure 4. Pitfall trap covered with a plastic cover (a), recharging the trap with preservative (b), and an example of a field processing kit (c).



Figure 5. A mechanical vacuum is used to sample *Goodenia* sp. (a), a yellow sticky trap placed below a grapevine canopy (b), and stored inside a plastic sleeve to facilitate handling once it is used (c).

A mechanical vacuum (Figure 5a) can be used to sample vegetation by placing a tube into the air intake slot of a leaf blower and then placing a sock over the opening of the tube. Care needs to be taken not to suck up excessive quantities of plant trash along with the arthropod sample. This method can be used to sample vegetation located near the ground. A mechanical vacuum has the capacity to sample everything that is within the sampling area. However, this technique performs poorly if large leaves are present as they may block the intake, and also some insects may remain attached to the foliage. Therefore, there can be a large variation of suction sampling efficiency (Sanders and Entling 2011).

Flying insects such as moths, flies and parasitoids are often sampled using yellow sticky traps placed in the canopy (Thomson and Hoffmann 2007) (Figure 5b). They may be messy to use due to the sticky glue incorporated on the card, which is used to capture the arthropods. The used sticky traps can be placed inside a clear plastic bag to facilitate the storage and identification the contents (Figure 5c).

Sampling bias

Each sampling method has advantages and disadvantages, depending on the species of arthropod to be studied and the local habitat (Standen 2000, Thomson *et al.* 2004, King *et al.* 2012). Bias will occur regardless of the sampling technique employed. For example, spiders are less likely to be collected on yellow sticky traps and airborne insects are less likely to be collected in pitfall traps. Likewise, ground based arthropods such as wolf spiders (Lycosidae) may be collected in higher abundance in pitfall traps than canopy dwelling species due to the sampling bias associated with these traps.

Bias can also occur depending on the time of day and/or season that samples are collected and the height of the vegetation sampled. One way to reduce the bias is to collect samples at the same location and time each day or night. The use of sweep and beat nets during the day may not take into account specific arthropod activity, which is predominantly nocturnal, such as green lacewing adults, and this must be considered when reviewing the results. Growers may wish to focus on collecting a representative sample that can be processed in a timely manner to provide useful insights.

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