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Archie K. Murchie, Agri-Food & Biosciences Institute, UK




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1 Introduction

Monitoring of insect pests is an essential component of integrated pest management. To determine whether pest densities justify an intervention, such as an insecticide application or other control measures, their numbers need to be assessed and compared to economic thresholds (Pedigo and Buntin, 1994; Ramsden et al., 2017; Stern, 1973). Such assessments can be done by physical sampling of the crop and visual inspection or passively using traps. The method used will depend on the biology of the pest species, the ease of assessment, the availability of labour and the relationship between the stage monitored and the damage done. For example, traps may monitor the adults, whilst the damaging stage is the larva. The closer the temporal and spatial association between the monitored stage and the damaging stage, the more accurate the assessment but the less advanced the warning provided.

For traps, cost and ease of use are important practical elements for in-crop sampling. For all trapping systems, the processing times and costs are major considerations. Some flight interception traps such as Malaise traps catch a

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wide variety of flying insects, with several thousand specimens collected over a trapping period (Skvarla et al., 2021). For biodiversity assessment, non-selective traps like this are ideal; however, for monitoring crop pests, selective traps are usually required that produce as targeted a sample as possible with little bycatch. This makes for speedy and accurate processing, as well as minimising impacts on non-target species. In-crop trapping systems therefore aim to be attractive to the target pest species but not to other insects within the crop. The exceptions to this may be where two species interact, such as, two pest species [e.g. cabbage seed weevil, *Ceutorhynchus obstrictus* (Marsham), and brassica pod midge, *Dasineura brassicae* (Winnertz)] (Smart et al., 1992), or where there is an opportunity to monitor a pest and its natural enemy at the same time (Murchie et al., 1997).

The first challenge in successful pest monitoring is to produce a cheap, selective trap that is highly attractive to the pest species. The second challenge is that the trap must be effective enough to provide an accurate population assessment. The third challenge is to reduce the trap sample processing time. Correspondingly, there is increasing interest in automatic methods of sorting and identification of trap catches. The fourth challenge is to reduce the environmental impact of trapping. Indiscriminate traps such as sticky traps are notorious for their large bycatch, even including small birds, bats and lizards, whilst water traps catch beneficial insects such as bees, hoverflies, lacewings and parasitoids. There is also the difficulty of disposal. Sticky traps are awkward to handle and invariably made of plastic, which once contaminated by the sticky glue cannot be easily recycled (Solorzano et al., 2015). The handling and disposal of any chemical preservative involved also needs to be considered.

2 Basic trapping elements

There are four basic components to an insect trap. The first is a method to make the trap attractive to the pest. This is commonly achieved using an attractive colour, light (for nocturnal insects) or more selectively volatile chemicals, in particular pheromone lures. Combinations of colour and chemical attractants are common, and trap design can use both visual and olfactory cues to enhance collection (Blight and Smart, 1999). An exception to this requirement would be where the trap is intended to sample specifically background insect populations, for example with suction traps, pitfall traps or clear flight interception traps. The second component is a mechanism to retain the catch. Insects may be retained on sticky glue, water with a dash of detergent to break the surface tension, or caught into a preservative such as alcohol. Some traps may be run for the purposes of live capture, particularly for biodiversity assessment (e.g. moth trapping) or to collect specimens for further work. In such cases, the design of the trap will allow entry but minimise the likelihood of exit. Often traps will be left for several days or longer before being emptied, so some method of preserving the catch must be used, if identification of specimens is required. Common preservatives are alcohol, if evaporation can be minimised, and ethylene or propylene glycol (anti-freeze), where the trap is more open to the elements. Alternatively, some traps operate dry and use insecticides to rapidly kill the catch. Lastly, the trap must be mounted in such a way as to maximise catch. Often for flying crop pests, this is at the crop canopy height and unobscured by plant foliage, which changes as the crop grows; therefore, height mounts for the trap are adjustable.

3 Making the trap attractive

3.1 Visual cues – colour and light

Colour is the simplest method of increasing trap attractiveness. Insects have diverse and highly developed colour vision, with some species having up to six different spectral receptors and a visual range of <300 nm [ultraviolet (UV)] to >700 nm (Briscoe and Chittka, 2001; Fennell et al., 2019). Specific wavelengths are attractive to certain insect species. Yellow in particular has been used for attracting phytophagous insects and has been recommended for monitoring glasshouse pests since the 1920s (Lloyd, 1922). This may be because the peak colour reflectance of plants is in the yellow band at 500–580 nm (Prokopy and Owens, 1983), due to brightness/intensity effects (Döring and Chittka, 2007) or attraction of phytophagous insects to stressed/diseased plants (Hodge et al., 2011). However, other colours have also proven attractive depending on the specific pest. For example, Kirk (1984) demonstrated that within the thrips species caught in coloured pan traps in an English sports-field, there were strong species-specific colour preferences for yellow, white, or blue, but there were also some grass-feeding thrips that were caught equally in all colours. The author suggested that yellow was in general the most attractive for non-grass foliage feeders, dark colours (black or red) for biting flies and wood borers, and white or blue for predators and parasites not associated with foliage. Responses to colour are therefore specific to the ecology of the pest species and can be subtle. For example, the contrast between the trap colour and background can affect the response of insects to traps, as can the sex and physiological state of the insect (Blackshaw, 1983; Košťál and Finch, 1996; Murchie et al., 2018). Due to the sensitivity of insects to colour, when conducting choice experiments to aid in designing traps, spectral reflectance measurements in visible and UV using spectrophotometers are preferable to human-vision assessment of colour hues.

Light trapping is used extensively for monitoring flying nocturnal insects. Many species have strong attractions towards artificial light sources particularly those emitting UV wavelengths. The reasons for this are not entirely known but seems to do with innate phototactic responses to natural nocturnal light sources such as moonlight and starlight for orientation (Nowinszky, 2003; Spalding, 2019), with the artificial light source acting as a super-stimulant. A wide variety of light-trap designs for catching moths are available, and there are networks of moth recorders in many countries running these for biodiversity assessment (Fox et al., 2011; Woiwod and Harrington, 1994). The other major use of light traps is to monitor biting insects, predominantly mosquitoes but also biting midges and sand flies. Light-trapping techniques for biting insects of medical importance are well established, and standard protocols are often available (European Commission, 2007; Silver, 2007). As with attraction to colour, attraction to light is dependent on the light source wavelength and intensity and is often species and sex specific. Consequently, whilst a general issue with light traps is that they can be non-specific and result in large bycatches, this can be mitigated against by use of very specific wavelengths, such as those available through wavelength-specific light emitting diodes (LEDs), which are highly attractive to the target pest (Kim et al., 2019). However, for many major lepidopteran pests, species-specific pheromone traps have been developed, replacing light traps for monitoring. For crop pests, light trapping has mainly been used for scientific studies or surveillance at the national level, rather than in-crop monitoring. This is due to the cost of the traps and the need for a power source, although solar-powered light traps are becoming more common as technology develops (Mohammed et al., 2018). Light-trap catch data of cotton bollworm, *Helicoverpa armigera* (Hübner), and the native budworm, *Helicoverpa punctigera* (Wallengren), were used to develop weather-based regression models to forecast pest populations in Australia (Maelzer et al., 1996; Maelzer and Zalucki, 1999; Zalucki and Furlong, 2005). Recently, wavelength-specific light traps have been developed for monitoring invasive stink bugs (Pentatomidae), utilising LEDs and pheromone lures, with the specific aims of reducing bycatch and lowering trap power consumption (Endo et al., 2022; Rondoni et al., 2022). Perhaps the most extensive current use of light traps for monitoring pests is to analyse and predict the migrations of the rice pests, the brown planthopper [*Nilaparvata lugens* (Stål)] and white-backed planthopper [*Sogatella furcifera* (Horváth)], into Japan, China and the Korean Peninsula. These migratory pests are monitored using networks of light traps run by plant protection authorities (Hu et al., 2011; Hu et al., 2014; Hu et al., 2017; Lu et al., 2017; Ma et al., 2017; Matsumura, 2001).

3.2 Olfactory cues – semiochemicals

Insects have well-developed olfactory senses and mostly detect volatile chemicals through their antennae. Correspondingly, most insect pests respond

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strongly to olfactory cues derived from their host crop or from conspecifics. The term 'semiochemical' refers to a range of behaviour-altering chemicals. For the purposes of pest trapping, these are usually classed as kairomones and pheromones. Kairomones are chemicals that benefit the receiver but not the sender, for example, plant-volatile chemicals that attract pests, whilst pheromones are chemicals released by organisms that illicit a specific response in the receiving organism of the same species (Dicke and Sabelis, 1988), for example, sex pheromones. Due to their specificity and attractiveness, pheromone-based trapping provides an ideal mechanism for monitoring insect pests on crops, and hundreds of chemicals have been identified for this purpose (Witzgall et al., 2010). However, in cases where pheromones are not suitable, not identified, or synthesised, plant-volatile chemicals are also used as trap lures, although these often lack the specificity of pheromones. The online database, Pherobase, details pheromones and other semiochemicals associated with more than 7500 species (El-Sayed, 2022).

Pheromone trapping commonly utilises either sex pheromones or aggregation pheromones. The former are usually released by the female to attract the male, whilst aggregation pheromones are released by conspecifics often to mass attack a plant in order to overcome its defences as well as initiating courtship and mating. Isolation of sex pheromones usually starts with behavioural observations, such as the mating behaviour of a pest, or the attraction of multiple males to virgin females. Next extracts of the natural pheromone are extracted from the pest. For example, brassica pod midge (*D. brassicae*) males were attracted to live virgin females, crushed females, hexane washes of females and hexane washes of ovipositors but not to washes of female bodies without ovipositors (Williams and Martin, 1986). Natural pheromones can be collected either by solvent washes of females or by air entrainment, with the latter providing a purer sample for subsequent analyses (Hall et al., 2012). Gas chromatography coupled with electroantennography studies are usually conducted to determine the active components of the pheromone. In this process, the raw pheromone source is passed over the target insect's antenna whilst at the same time passing through a gas chromatograph. Electrical signals generated by the antenna in response to the chemicals can be compared with the gas chromatograph trace and the components that elicit most responses identified. There is a great diversity in pheromone chemistry. In the Lepidoptera, which is the most studied group, pheromone components are typically alcohols, acetates or aldehydes derived from fatty acids; however, insect pheromones also include, amongst others, polyenic hydrocarbons, methyl-branched hydrocarbons, epoxides, ketones, terpenes, terpenoids, iridoids and alkaloids (Jacquin-Joly and Groot, 2018; Rizvi et al., 2021). Furthermore, identification and synthesis of pheromones can be complex due to the sensitivity of insects to precise multi-blend formulations and isomeric compounds including chiral molecules. On the other hand, pheromone components can be active across several species, and there is often similarity in pheromone components within taxa, which can aid in identification and synthesis (Xu et al., 2020).

Once synthesised and tested, the synthetic pheromone is embedded in a suitable substrate that allows for a measured and consistent release of the volatile chemicals over weeks or months. This is usually a rubber septum, polyethylene vial or cellulose sponge enclosed in a sealed polyethylene pouch. Fortunately, insects are highly sensitive to pheromones, and therefore small to minute quantities of chemical are required. The optimum release rate of the pheromone lure and its positioning on the trap is derived from field trials. Good examples of the development process for pheromone trapping of apple leaf midge (*Dasineura mali* Kieffer) and strawberry blossom weevil (*Anthonomus rubi* Herbst) from pheromone synthesis through to field use are given in the previous studies (Cross and Hall 2009; Cross et al., 2006a; Cross et al., 2009; Cross et al., 2006b).

4 Common trap types for collecting pest insects

There are a multitude of different insect trap designs reflecting the great diversity of insect pests, the crops on which they are found, the behaviour of the pest in question and the influence of local factors such as the availability of materials, power sources and labour. Trap types and the names given to them are not absolute, and some traps may use a combination of techniques to trap pests, so, for example, what might be termed a funnel trap in one cropping system may be called a cross-vane trap in another. However, there are some basic commonalities, and these are reflected in the following general categories of crop pest trap design. Some traps which are predominantly used for biodiversity studies are not covered in detail here. This includes common designs such as pitfall traps, where a container is dug into the ground to soil level and crawling insects fall into it. Pitfall traps are commonly used in agro-ecology studies to assess the prevalence and activity of ground-dwelling predators (Brown and Matthews, 2016; Hohbein and Conway, 2018; Woodcock, 2005) or occasionally pests (Hanula, 1990) but are not typically used for routine pest assessment.

4.1 Water traps

Water traps are simple generalist traps, also known as Moericke or pan traps. They consist of coloured bowls filled with water and a little detergent, which serves to break the surface tension as the insect lands on the water and then sinks. Water traps are normally mounted at crop canopy height. The most common design is for the trap to be mounted on a central pole that passes through the centre of the bowl, through an appropriate conduit, which in

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commercial traps is a moulded part of the bowl. The trap can then be easily raised and lowered by moving it up and down the pole. The other feature is mesh-covered drain holes to prevent overflow if it rains. The attractiveness of the trap is due to the colour of the bowl. For phytophagous insects, yellow is the most common attractive colour (Kirk, 1984), whilst white and blue are also used widely. The effectiveness of the colour is dependent on the taxa (Disney et al., 1982) and other visual issues such as contrast with the background. In some cases, the attractiveness of the trap will alter as the crop matures, e.g. on oilseed rape a yellow trap becomes more conspicuous and attractive to pests as the yellow flowers senesce and the crop becomes green, whilst *Drosophila suzukii* Matsumura are caught less in baited traps when the surrounding crops ripen and thus become more attractive to the flies. Sodium benzoate, 50/50 ethylene glycol/water or saturated salt solution will aid in preserving the catch, although after a few days in water, specimens will become macerated and start to decay. Trap catches can be sieved and preserved in alcohol before sorting. Due to the lack of specificity, water-trap catches are usually sorted under the microscope.

4.1.1 Case study of a water-trapping regime – aphids on potatoes

Yellow water traps are used to monitor pest aphid species on potato crops in the UK (AHDB, 2021; AHDB Potatoes, 2021) (Fig. 1). Aphids can vector potato viruses, and this monitoring system is concerned with transmission of potato

Figure 1 Yellow water trap mounted on a central pole within a potato crop and used to monitor pest aphid species. Source: AFBI image.

virus Y by peach-potato aphid [*Myzus persicae* (Sulzer)] and other aphid species. A sampling kit consisting of yellow water traps, mounts and sample pots is sent to growers or crop inspectors. Growers maintain the traps on their crops, and weekly trap samples are sieved and posted back into the laboratory for identification. The results consist of aphid species counts and a vector pressure index that is calculated as the number of aphid species present multiplied by their relative transmission efficiency (Northing et al., 2004). Growers receive their individual results which are also incorporated into an online national risk map. Growers and advisers can then use this information for timing of haulm burn down and insecticide application if necessary. For seed potato producers, EEC directive 66/403/EEC states that the virus incidence must be less than 4% in the direct progeny of the crop. The service is funded through a levy board, the Agriculture and Horticulture Development Board (AHDB), on a semi-commercial basis in England, Wales and Scotland, and through Government agricultural advisory bodies in Northern Ireland (AFBI 2022).

4.1.2 How water traps meet the key challenges of pest monitoring (e.g. selectivity, accuracy, processing and cost)

Water traps are widely used for crop pest and biodiversity assessment. They are poor to moderately selective as they rely on colour to attract insects, so trap catch is likely to be diverse. Semiochemical lures can be mounted over water traps to improve catch of target species (Blight and Smart, 1999; Hardie et al., 1996), but without a barrier to incursion of other insects (Conrad et al., 2016) or the use of clear or plant-coloured traps with reduced visual attractiveness, the problem of an indiscriminate catch remains. The accuracy of water traps relies on visual attraction and is good where there is contrast between background foliage and the trap colour (Döring et al., 2004). This is particularly so for migrating insects, such as aphids, which are attracted to conspicuous yellow traps as they fly onto the green crop (Döring, 2014). Absolute accuracy for any pest monitoring trap is difficult to determine, and on-plant counts remain the most accurate assessment. Water-trap catches mostly correlate with on-plant counts. However, this varies with cropping system and pest species (Kisimoto, 1968; Lykouressis et al., 2017; Metspalu et al., 2015; Trumble et al., 1982). Nonetheless, trap catches, at the very least, provide an early indicator of pest species within the crop. Processing is straightforward, and water-trap catches can be sieved and stored in alcohol. However, sorting trap catches to species requires significant microscope time, and only cursory assessment can be done by eye or using a hand lens in the field. Although, possibly some distinctive species such as cabbage stem flea beetle [*Psylliodes chrysocephala* (L.)] may be identified in the field especially if they are sampled in the autumn/winter, when bycatch is low. The great advantages of water traps are that they are cheap and simple, using ready-available containers and water, plus household chemicals. They do catch a wide range of non-target species, but other than that environmental impact is low as they are reusable.

4.2 Sticky traps

Sticky traps are one of the simplest and cheapest methods of monitoring insects. At the basic level, they consist of a thin plastic sheet coated in non-drying, water-resistant glue. The plastic sheet is usually of an attractive colour, with yellow, blue or white being the most common commercial colours. Sticky traps are usually mounted vertically, although horizontal and 45°–mounted traps can be more effective depending on the target pest (Blight and Smart, 1999; Holland et al., 2021; Smart et al., 1997). Some traps utilise a cross-vane design, with vertical panels set perpendicular to each other, e.g. the Rebell® trap (Katsoyannos et al., 2000; Remund and Boller, 1978). Insects alighting on the trap are caught in the sticky glue. The glue used on commercial sticky traps is petroleum derived such as polyisobutylene, but other sticky substances such as petroleum jelly, tree resins, boiled linseed oil, corn syrup or honey have also been used (Murphy, 1985). The type of glue can change the reflectance of the trap colour and alter its attractiveness to pests (van Tol et al., 2021). A coloured sticky trap is non-specific and will catch a wide range of flying insects. If the target species for monitoring is easy to identify or if the trap is operated in a protected environment with a single pest species present, counts can be derived directly from the sticky trap by eye, using a hand lens or dissecting microscope. This can be facilitated by covering the sticky trap with a transparent plastic film (e.g. cling film). In this manner, sticky traps can be easily stored and, provided the trap was dry, kept indefinitely. Otherwise handling sticky traps can be messy due to the glue.

The disadvantages of sticky traps are that specimens are often distorted and can be damaged by bird feeding and features may be obscured due to the glue. Various methods have been tried to dislodge and clean specimens for more accurate identification through microscopy or molecular techniques. Murphy (1985) lists several solvents which can be used to dissolve polyisobutylene, such as toluene, heptane, hexane, xylene, ethyl acetate and others. More recently, for health and safety reasons, solvents such as the commercially available De-Solv-it® (Davidson et al., 2015) and orange oil have been recommended. However, the use of solvents can interfere with polymerase chain reaction procedures for identification of pest species or detection of pathogens within insect vectors (Bextine et al., 2008; Marshall et al., 2010).

Sticky traps are often baited with pheromone lures in a delta trap or prism trap design. The body of the trap typically consists of corrugated plastic or waxed cardboard formed into a triangular prism with a sticky trap insert placed glue-side up on the base of the trap. A pheromone lure is mounted inside the prism, either hanging from a thin wire or directly onto the sticky trap insert. This is a cheap and simple design. The outer delta housing protects the pheromone lure and catch from rain and sunlight and helps minimise bycatch of non-target organisms. Choice of colour and other visual cues can enhance catch (Domingue et al., 2016) or dissuade non-target organisms (Clare et al., 2000).

4.2.1 Case study of sticky traps – pea moths

Pea moth [*Cydia nigricana* (F.)] is an economic pest of peas (*Pisum sativum* L.). The larvae feed inside the pea pod, and, as well as direct feeding damage, contamination can lead to the downgrading or rejection of crops for human consumption. This is particularly so with vining peas (harvested green for freezing or canning) where the moth larvae cannot be removed physically during processing, and the threshold for rejection by processing factories can be very low, e.g. 0.5% damage of the consignment in organic crops in Germany (Schultz and Saucke, 2005; Thöming et al., 2011). Female pea moth lay eggs on the leaves, and the first-instar larvae move to and penetrate the developing pod to feed on the peas. The optimum time for any insecticide treatment is when the larvae are exposed and prior to them entering the pod. Depending on whether the crop type is combining or vining, there is a need to determine the requirement for – and timing of – any control measures (Wall, 1988). This problem was addressed by the development of one of the first commercial examples of a pheromone-based pest monitoring system using economic thresholds (Witzgall et al., 2010).

Pheromone-baited delta traps are used to monitor pea moths within crops (Fig. 2). Originally, two compounds were found to be highly attractive to male moths in the field, (*E*,*E*)-8,10-dodecadien-l-yl acetate and (*E*)-10-dodecen-l-yl acetate, with the former subsequently identified as the female sex pheromone (Greenway, 1984). However, this compound isomerised in sunlight to form a repellent (Witzgall et al., 1993). Therefore, the less attractive pheromone analogue [(*E*)-10-dodecen-l-yl acetate] was the practical choice as a more persistent lure, although the pheromone itself combined with an antioxidant to prevent isomerisation was recommended for the more precise detection of moths required for vining crops (Wall, 1988). This is a good illustration of the unforeseen and subtle responses of insects to semiochemicals and demonstrates the fine-tuning and practical compromises required in developing pheromone lures for field trapping. The pheromone lure is impregnated into a rubber septum and hung inside a delta trap with a sticky, removable basal plate. The current recommendation is that two pheromone traps are set per field and examined every 2 days. For combining crops for human consumption, the

Figure 2 Delta trap with sticky trap insert. The trap is baited with a pheromone analogue to attract male pea moths (*Cydia nigricana*). Source: Image courtesy of the PGRO – Processors & Growers Research Organisation, Peterborough, UK.

threshold is 10 or more moths in either trap during two consecutive occasions, with spray date determined by a computer model of moth egg development. For vining crops, the presence of any male moths suggests that control will be necessary (PGRO, 2022; Ramsden et al., 2017; Wall, 1988).

4.2.2 How sticky traps meet the key challenges of pest monitoring (e.g. selectivity, accuracy, processing and cost)

The advantages of sticky traps are that they are a cheap and flexible method of monitoring pest insect numbers. However, the selectivity, accuracy and processing time of sticky traps depends on whether they are relying on colour or semiochemical lures as an attractant and consequently the visual and olfactory sensory ranges of the target insects. A non-conspicuous delta trap baited with a sex pheromone can be highly specific and allow direct counts by eye for pest monitoring; whereas a yellow sticky trap mounted in an open field situation will catch many insect species. Even within a protected environment dealing with a single pest species, there are many factors influencing the accuracy of sticky traps. Pinto-Zevallos and Vänninen (2013) provide an in-depth review of the use, placement and processing of yellow sticky trap in glasshouses to monitor whiteflies. They comment that there are relatively few studies quantifying yellow sticky trap efficacy but of those that do, yellow sticky traps in glasshouses are comparable in terms of efficiency to other sampling methods, including direct on-plant counts.

4.3 Suction traps

Suction traps are non-attractive and sample flying insects from a defined stratum of air. At their simplest, they consist of an impeller fan that sucks air plus insects through a conical mesh sieve to concentrate the insect sample into a collecting jar. Depending on the design, the fan may be mounted at the air intake or at the bottom of a vertical tube that allows higher strata to be sampled. The advantages of suction traps are that they measure absolute abundance per unit volume of air (Harrington et al., 2007b; Taylor, 1982) and are not reliant on trap attractiveness, which may vary depending on the pest's physiological state or background colours or odours. The main disadvantages of such traps are that they require a mains or generator electric power source (Johnson, 1950), are large, are comparatively expensive pieces of equipment and are therefore less suitable for multiple on-crop use. The first design for a suction trap for use on crops was that of Johnson (1950), with improved designs in subsequent versions (Taylor, 1951) and therefore known as the Johnson–Taylor suction trap. This design of trap is still commercially available through Burkard Manufacturing Co Ltd (Rickmansworth, UK) (www.burkard.co.uk). Due to the expense of the trap and the requirement for a power source, most work on crop pests using the Johnson–Taylor suction traps has been for scientific studies or for national surveillance of pests and not monitoring by commercial growers. A smaller battery-powered suction trap was developed and used by Wainhouse (1980) to sample first-instar larvae of the felted beech scale, *Cryptococcus fagisuga* Lindinger, within a forest environment, and Belding et al. (1991) used a solar-powered suction trap for sampling aphids in arable crops in Illinois.

The most widespread use of suction traps has been the sampling of aphid pests of crops. A network of 12.2-m stationary suction traps throughout the UK (Harrington and Woiwod, 2007; Woiwod and Harrington, 1994) was initiated in 1964 to monitor aphid populations ostensibly but has subsequently been used for much more besides this (see Case study in Section 4.3.1). Developed at Rothamsted Research in Harpenden, UK, the Rothamsted 12.2 m suction trap consists of a 9.2 m plastic pipe (internal diameter 244 mm) sitting on a 3 m tall, sealed box containing an electric fan (Fig. 3). The height of 12.2 m (40 feet) matches the mean logarithmic height for aphid flight and is sufficiently high to minimise the effects of immediate surroundings (Harrington and Woiwod, 2007; Taylor, 1974). Air sucked down the pipe passes into an expansion chamber to reduce its speed, and the catch is then sieved through a stainless steel mesh cone into sample jars containing preservative (Macaulay et al., 1988). Similar networks of aphid-monitoring suction traps have been developed across Europe (Harrington et al., 2007a; Woiwod and Harrington, 1994) and also in the USA since 1983, although they are largely based on the design of Allison and Pike (1988). More recently, suction trap networks have been used as forecasting tools for soybean aphid,

Figure 3 Rothamsted 12.2 m suction trap used to sample aphids. The trap is permanently sited and mains powered with an impeller fan housed in the stainless steel body and a long, vertical sample pipe to collect aphids migrating to and from crops. The door of the trap is open showing the collecting mesh and four sample containers, which rotate every 24 h. Source: AKM image.

Aphis glycines Matsumura, an invasive pest first found in North America in 2000 (Rhainds et al., 2010; Schmidt et al., 2012). There are also suction trap networks in New Zealand (Teulon et al., 2004), South Africa (Krüger and Laubscher, 2013) and China (Qin et al., 2013). Individual traps are operated in Northern Ireland by the Agri-Food and Biosciences Institute and in the Republic of Ireland by Teagasc.

4.3.1 Case study suction traps – Rothamsted trap network

A suction trap network for sampling pest aphid species in Great Britain currently consists of 16 12.2 m Rothamsted traps, 12 in England and 4 in Scotland, managed by the Rothamsted Insect Survey (RIS), England, and Science and Advice for Scottish Agriculture (SASA), Scotland (Bell et al., 2015; Harrington and Woiwod

2007). Trap catches are sent to the RIS or SASA for expert identification. The traps provide daily records for a range of aphid species from April to November and weekly aphid counts in the winter (Rothamsted Research, 2021). The principal reason for the network is to forecast potential damage to crops, mainly cereals, from aphids and the viruses that they transmit, based on the timing and size of aphid migrations (Bell et al., 2015). For example, preceding winter temperatures provide a good predictor for the date of spring aphid migrations (Harrington et al., 2007b; Harrington and Woiwod, 2007), and aphid forecasting models have been developed for cereals as well as hops, beans, potatoes and sugar beet (Woiwod and Harrington, 1994). Any insecticidal applications can then be timed correctly for efficient impact on the pest whilst minimising non-target impacts. Each trap is estimated to be representative of aphid abundance in at least an 80 km radius (Cocu et al., 2005; Rothamsted Research, 2021; Taylor, 1974). Weekly aphid counts are disseminated in electronic bulletins and text alerts to interested parties. These data feed into decision support systems (DSSs) for management of aphid pests. The Northern Irish trap ceased to be part of the RIS network in the late 1980s, but a new trap was erected in 2019. In this case, data are displayed graphically on an aphid-monitoring dashboard that compares current counts with previous years. The chart, summary information and a website link are tweeted out to growers (AFBI, 2022).

In addition to monitoring aphid flight and migration, suction trap catches are also regularly used to assess virus prevalence in aphid vectors and insecticide resistance in aphid populations (Harrington and Woiwod, 2007). Since all samples, including the bycatch, have been stored by Rothamsted since 1974, suction traps have also been used retrospectively to study phenologies and flight activities of other taxa, e.g. biting midges (*Culicoides* spp.) (Fassotte et al., 2008; Sanders et al., 2011), social wasps (Archer, 2001), pollen beetles [*Brassicogethes aeneus* (Fabricius)] (Shortall, 2021) and others (Shortall et al., 2013). However, in recent years, the long-term datasets built up from regular aphid monitoring have attained a unique importance. They have been used to demonstrate the impact of climate change on aphid populations, with aphid spring migrations becoming progressively earlier and increased duration of the flight season, as winters have become warmer (Bell et al., 2015; Harrington et al., 2007a).

4.3.2 How suction traps meet the key challenges of pest monitoring (e.g. selectivity, accuracy, processing and cost)

Suction traps are somewhat different to the other trapping systems in that they are designed to sample background insect populations and are not attractive; therefore, their non-selectivity is an advantage. Nevertheless, the likes of the 12.2 m Rothamsted suction traps are carefully positioned in such a way as to intercept migrating aphids from what has been termed the 'aerial plankton', to give a representative forewarning of the timing and density of aphid migrations. These traps are operated on a regional basis because they are comparatively expensive to build, run and process the aphid samples. However, the outputs from the traps are disseminated to a wide range of growers, so they are cost effective.

4.4 Interception traps (window, Malaise and cross-vane)

Various designs of flight interception trap are used to sample insects. Window traps typically consist of a vertical, clear plastic panel (i.e. acrylic, Perspex or Plexiglass) with a collecting trough or drop tray immediately beneath and a rain-guard above (Leather, 2015). Insects fly against the window and fall or crawl downwards in the drop tray, which may be filled with a suitable preservative. Window traps are particularly good for sampling beetles, as they bounce off the window into the sample tray, and the traps are often used in forestry to sample bark beetles (Ozanne, 2005) and even minute Ptiliidae (Komosiński and Marczak, 2018). They have been used for sampling crop pest beetles during experimentation, e.g. pollen beetles on oilseed rape (Ahmed et al., 2013). As they are double sided, they can be used to assess the direction of insect flight (Riis and Nachman, 2006; Williams et al., 2007). However, for routine sampling, they are bulky and prone to damage in high winds. Alternative versions have been developed using a fine permeable mesh as the window. Masner and Goulet (1981) used a mesh window trap treated with pyrethroid insecticide to sample for small Hymenoptera, which are under-represented in Malaise traps as they show a weaker phototrophic response or have difficulty crawling on the Malaise trap material. A very cheap window trap made with a 2 litre soft-drink bottle has been trialled as a potential citizen science trap for monitoring bark and ambrosia beetles (Steininger et al., 2015).

The Malaise trap is the one of the commonest and most widely used flight interception traps. The trap was originally developed by the Swedish entomologist René Malaise who was inspired by the capture of flying insects inside a tent (Malaise, 1937), and correspondingly the structure of the trap resembles an open-sided ridge tent, constructed from fine mesh material (Fig. 4). The trap is erected like a tent with poles and guy ropes holding the mesh trap in place. A central vertical mesh panel runs the length of the trap. Flying insects are intercepted by this mesh panel and move upwards by positive phototaxis (or negative geotaxis) towards an apex, leading them into a collecting jar containing a preservative, usually ethanol (Sheikh et al., 2016). The roof and end walls of the trap provide some shade and are sloped upwards towards the apex, thus further directing any insects towards the collecting jar. A secondary drop tray may be positioned underneath the central panel in the manner of a window trap, to catch any insects such as beetles which drop down when encountering a barrier. The Malaise trap is a powerful tool for sampling

Figure 4 The Malaise flight interception trap is ideal for biodiversity sampling of flying insects, particularly Diptera and Hymenoptera. As a large, delicate and non-selective trap, its use in crop protection has mainly been for experimental studies. Source: AKM image.

insect biodiversity (Skvarla et al., 2021), and as the catches are retained directly in preservative, the quality of specimens is good. This makes the Malaise trap an ideal technique for collecting Diptera and Hymenoptera (Skvarla et al., 2021), including parasitoids (Darling and Packer, 1988; Fraser et al., 2008), which can have relevance to crop protection. Malaise traps have been often used for experimental purposes and to assess biodiversity in agro-ecosystems (Burgio and Sommaggio, 2007; Chay-Hernández et al., 2006; Letourneau et al., 2012; Macfadyen and Muller, 2013; Volpato et al., 2020). One particular study attracted much attention, with an apparently drastic decline in flying insect biomass over 27 years as derived from Malaise trap samples taken in mixed landscapes in Germany (Hallmann et al., 2017). However, Malaise traps are not generally used for routine pest monitoring as they are large and delicate and produce an extensive and mostly indiscriminate catch.

The cross-vane trap works on a similar principle of flight interception as the window trap (Leather, 2015) but is smaller and usually baited with semiochemical lures or a light source. A basic cross-vane trap consists of two panels (the vanes) perpendicular to each other, with a funnel at the bottom (Hines and Heikkenen, 1977) (Fig. 5). The vanes may be clear plastic or an attractive colour depending on the target species. An upper funnel may serve as a rain-guard or alternative collecting mechanism (Knuff et al., 2019; Wilkening et al., 1981), whilst the

Figure 5 A cross-vane trap used for trapping bark beetles in forestry. The trap is baited with a lure, and beetles impact the vanes and fall into the collecting funnel and jar at the bottom. Source: AKM image.

bottom funnel leads to a collecting jar containing a preservative. Flying insects are intercepted by a clear trap or attracted to the colour, lure or light source whereupon they impact the trap and fall, or crawl, into the collecting funnel. The trap may be suspended from a branch or frame (Hines and Heikkenen, 1977; Wilkening et al., 1981) or mounted on a pole (Murchie et al., 1997).

4.4.1 Case study – experimental cross-vane trap for sampling **Dasineura brassicae** *and its parasitoids*

Murchie et al. (1997) used a cross-vane trap baited with plant kairomones to trap brassica pod midge (*D. brassicae*) and its parasitoids in an oilseed rape crop. The trap design was modified from that of Wilkening et al. (1981) and consisted of yellow painted cross-vanes, above which was a funnel, leading through a clear Perspex column into a collecting jar containing methanol as a preservative (Fig. 6). Isothiocyanates lures were mounted inside the Perspex column. Isothiocyanates are defence chemicals produced by Brassicaceae plants when damaged. The pest species, *D. brassicae*, was caught significantly more in traps

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Figure 6 A cross-vane trap for monitoring brassica pod midge and its parasitoids in oilseed rape. The trap is baited with brassica-derived kairomone lures, with different compounds attracting either pests or their parasitoids. Source: AKM image.

baited with allyl isothiocyanate than the control; whereas *D. brassicae's* main parasitoid, *Platygaster subuliformis* (Kieffer), was caught predominantly in traps baited with 2-phenylethyl isothiocyanate. The advantage of this system is that it can trap both the pest and natural enemy selectively dependent on the plant kairomone lure. Most pest monitoring systems are targeted towards the pest but do not necessarily monitor the presence of natural enemies. Knowledge of the occurrence and prevalence of both would lead to more refined economic threshold parameters and targeted integrated pest management approaches (Zhang and Swinton, 2009).

4.4.2 How interception traps meet the key challenges of pest monitoring (e.g. selectivity, accuracy, processing and cost)

There is a difference in approach between a Malaise or a window trap and a semiochemical-baited cross-vane trap. The former traps are largely non-selective and designed to intercept insects as they move along flight paths. Therefore, these traps are ideal for biodiversity assessment but are not commonly used for in-crop sampling. Whilst cross-vane traps can act as interception traps in their own right, for crop or forestry protection purposes they are usually baited with semiochemical lures. In this set-up, they behave more like a cone or a funnel trap, where the cross-vanes form part of the trap directing insects towards the semiochemical plume rather than acting as the main trapping component. This illustrates how an addition of a lure can dramatically alter the characteristics of the trap design.

4.5 Funnel and cone traps

Funnel and cone traps typically rely on semiochemicals to attract pest species. They form the basis of many commercially available trap designs for pheromone-based on-crop monitoring. They have the advantages of being reusable and more durable than sticky traps as they are not affected by dust and detritus and they do not lose effectiveness as catch numbers build up.

Funnel traps are designed for use with pheromone lures. The term 'funnel trap' is used for two main designs. A common design for capturing pest moth species consists of a pheromone dispenser mounted above a funnel structure leading into a collecting jar (Fig. 7). This design may also be commonly referred to as a 'bucket trap'. The funnel acts as a catch-retaining mechanism, as it is easier to enter the collecting jar through the funnel than to exit. They are normally constructed of three plastic sections that clip together. The upper section is moulded to form a rain-guard and pheromone dispenser receptacle. The middle section is the funnel unit and lower section is the collecting jar. Flying insects that get attracted to the pheromone flutter around the dispenser, fall into the funnel and are captured. Some designs may have cross-vanes to intercept insect flight and aid in capture (Fountain et al., 2017). These traps are used in field crop situations to monitor moth pests such as diamond back moth [*Plutella xylostella* (L.)] and silver Y moth [*Autographa gamma* (L.)].

The Lindgren funnel trap consists of a stack of several wide black funnels, convex side down, loosely hung together to form a column that mimics a tree trunk (Lindgren, 1983) (Fig. 8). The funnels can be compressed into each other to minimise space for transport and storage. Lindgren funnel traps are used predominantly to capture bark beetles in forests, rather than crop pest monitoring. Beetles attracted to the pheromone lures attached to the trap collide with it and fall through the series of funnels to a collection jar at the bottom containing a preservative such as ethylene glycol. A rain-guard sits above the uppermost funnel to prevent flooding.

Cone traps are the inversion of funnel traps and work by insects flying or crawling upwards inside a cone (or funnel), towards an attractant. The apex of the cone or spout of the funnel protrudes upwards into the collecting jar, and insects are retained on the lobster-pot principle that it is easier to enter the trap, being channelled by the cone, than it is to escape back through the small entrance hole. For many insects, a positive phototropic response will retain them in the collecting jar as well, especially if the trap bottom is opaque and

Figure 7 A moulded plastic funnel trap for trapping moths. Source: AKM image.

Figure 8 A Lindgren funnel trap used for trapping beetle pests in forest. The trap is baited with semiochemical lures, and the elongated shape mimics a tree trunk. Beetles that impact the trap fall into the funnels and ultimately the collecting jar at the bottom. Source: Image from Pavuk and Wadsworth (2013) under the Creative Commons Attribution (CC BY) licence. No changes were made to the picture.

the collecting jar transparent. For some cone traps, liquid preservatives, such as ethanol, can be held in the collecting jar or insecticidal kill strips can be used to further retain insects and aid in processing (Hardee et al., 1996). Cone traps can be mounted on crop for flying insects or placed on the ground for crawling pests. Depending on the design, the latter may be referred to as pyramid traps or Leggett traps and are used for trapping pest beetles which may crawl as well

Figure 9 A McPhail trap for monitoring fruit flies (Tephritidae). Flies enter the trap through an inverted funnel entrance in the bowl base attracted by pheromone lures or liquid food baits. Source: Image by Peggy Greb, USDA Agricultural Research Service.

as fly to their host crops (Mulder et al., 2012; Nielsen and Jensen, 1993; Prokopy and Wright, 1998; St Onge et al., 2018).

The McPhail trap is a common trap design that works on the cone trap principle and is used mainly for sampling pest flies or social wasps. In this design, the cone is built into the base of the main receptacle, with insects entering through an invaginated, inwardly tapering entrance in the bottom, attracted either by a pheromone lure or by liquid food baits (Hernández-Ortiz et al., 2021) (Fig. 9). Commercial traps are made of moulded plastic in a similar way as the funnel/bucket traps. They comprise two detachable parts: the receptacle bowl with the inverted funnel entrance, typically coloured yellow; and a transparent domed-cylinder upper section forming a cover to the bowl and inside which a pheromone lure can be suspended. A liquid bait is poured into the base of the trap and can contain a preservative to preserve the insect catch; alternatively, if a pheromone lure is used, liquid preservative (e.g. water, propylene glycol and sodium borate) can also be held in the base of the trap (Hernández-Ortiz et al., 2021; Thomas, 2008). In a cropping situation, McPhail traps are used for monitoring pest fruit flies (Tephritidae). The trap is named after M. McPhail who used the traps to study the Mexican fruit fly [*Anastrepha ludens* (Loew)] in the 1930s and 1940s, but the basic concept is older (Steyskal, 1977).

4.5.1 Case study suction traps – boll weevil monitoring using cone traps

One of the most extensive uses of cone traps is for monitoring cotton boll weevil (*Anthonomus grandis* Boheman) using the boll weevil 'grandlure'

Figure 10 A boll weevil cone trap. Weevils are attracted to the colour and pheromone plume from the baited traps. Upon landing, they move upwards into the cone and are retained within the collecting jar at the top. Source: Image by Dale Spurgeon, USDA Agricultural Research Service.

synthetic pheromone (Hardee et al., 1972; Tumlinson et al., 1969). The basic boll weevil trap consists of three parts: a light green or yellow upwardly tapered plastic cylinder, with a mesh funnel above leading to a transparent collecting jar (Fig. 10). Boll weevils are attracted to the colour of the trap and the pheromone plume. On landing on the tapered cylinder, they move upwards to the pheromone, through the overlapping funnel into the collecting jar. The specifics of the trap have been under constant refinement since the early 1970s, but most current traps follow the basic design of Mitchell and Hardee (1974) (Dickerson, 1986; Hardee et al., 1996). Boll weevil cone traps are used extensively in the Cotton Boll Weevil Eradication Program in the USA, a collaborative venture involving the USDA, state governments, researchers and cotton producers, which has seen boll weevil eradicated from most of the USA (Raszick and Bernaola, 2021).

4.5.2 How funnel and cone traps meet the key challenges of pest monitoring (e.g. selectivity, accuracy, processing and cost)

Funnel and cone traps mostly rely on semiochemical attraction. They are therefore highly selective and, depending on the attractant source, have a low bycatch. Accuracy is dependent on the range of the semiochemical plume and, as with all lure-based traps, dependent on weather conditions, the age of the lure and the response of the target species. The trap design of the moulded plastic funnel and McPhail-type traps can be used for a wide variety of pest species, which reduces manufacturing costs, and the traps are robust and reusable.

5 Automated traps

5.1 Time-sorting traps

The earliest form of trap automation was to time-sort trap catches. A light trap fitted with a clockwork time-sorting mechanism was in use as early as 1933 to determine the flight activity of nocturnal Lepidoptera (Williams, 1935). Timing automation serves to provide more information on the time and date of trap catches whilst avoiding the extra effort in changing sampling receptacles. The mechanisms for time-sorting are rotating turntables (Bolliger et al., 2020; Taylor et al., 1982; Williams, 1935; Williams, 1958), rotating sticky surfaces (Riedl and Croft, 1981) or mechanical separation of the catch. For the mechanical separation of the catch, the Johnson–Taylor suction trap used a disc-dropping mechanism to time-sort catches (Johnson, 1950; Taylor, 1951). An electrical time switch connected to a solenoid releases metal discs, fringed with cotton, to drop down the sample tube, stacking the suction trap catch into successive hourly samples. In an extensive study, Lewis and Taylor (1965) used the disc-dropping timing mechanism to derive the diurnal flight periodicity of 400 taxa of insects at multiple sites in the UK. Others have used clockwork mechanisms to time-sort catches. Murchie et al. (2001) used a 24 h quartz clock motor to rotate a funnel mechanism that directed Malaise trap catches into 2-hourly samples. This had the advantage of being cheap to run, as it relied on a single 1.5 V AA battery, which could power the motor for at least 3 months. They used the mechanism to derive the diel flight periodicity of *D. brassicae* on oilseed rape. Rohitha and Stevenson (1987) used a clockwork mechanism to drop and expose sticky trap drums in sequence to provide information on daily aphid catches. For the practical purposes of avoiding staff working during the weekends, the 12.2 m Rothamsted suction traps rotate sample jars using a stepping motor to provide 24 h trap counts over a 4-day period (Bouchery, 1979; Macaulay et al., 1988).

5.2 Automated counting and identification

The next development was traps that could automatically count and identify trapped insects. This is a rapidly evolving area as electronic sensors become cheaper and more sensitive, whilst traps can now be monitored remotely using mobile phone, wireless and cloud technology (Lima et al., 2020). Initially, most designs to count insects entering traps have used some form of optical detection (Sciarretta and Calabrese, 2019). For example, Hobbs and Hodges (1993) illuminated insects passing down a suction trap against a dark background, with sensors to detect the scattered light. Similarly, the passage of insects through infrared beams has also been used for counting insects entering traps, e.g. tobacco budworms and cabbage loopers (Hendricks, 1985), boll weevil (Beerwinkle, 2001), stored product pests (Epsky and Shuman, 2001) and orchard moth pests (Holguin et al., 2010). More recently a sophisticated system using backscattered infrared coupled with a high-speed photodetector has been used to detect insects within targeted volume of air (Rydhmer et al., 2022). Jiang et al. (2008) counted oriental fruit fly [*Bactrocera dorsalis* (Hendel)] entering methyl eugenol-baited traps using interruption of infrared beams as the counting trigger. Fruit fly counts along with temperature, humidity and wind speed logging were done remotely at the trapping unit and periodically sent to a server PC via the mobile phone (Global System for Mobile communication) network. The eventual idea is that real-time trapping data from a network of traps could be disseminated to regional farmers via the internet.

Other approaches have used digital images to count insect samples. Selby et al. (2014) used baited pyramid (cone) traps modified to house an infrared motion-sensing camera set-up, originally designed for wildlife recording, to count plum curculio [*Conotrachelus nenuphar* (Herbst)] in Michigan. The system took photographs of each weevil entering the trap, so the number of photographs acted as a count for trap entry, whilst the photographs could be retained to confirm identity. Photographs were stored on the camera's memory card and collected physically. Others have collected photographs remotely. Doitsidis et al. (2017) monitored olive fruit fly, *Bactrocera oleae* (Gmelin), in Crete using ammonium sulphate-baited McPhail traps fitted with GSM/GPRS-linked digital cameras that uploaded images to the internet using the mobile phone network. Counting of fruit flies was then accomplished using an automated multi-stage image analysis approach, whereby the image is enhanced and filtered and the fruit flies are counted as the percentage black within the processed image. This process relied on the specificity of the trap and was not able to distinguish fruit flies from other insects, although separation on the basis of size would seem to be possible using this approach.

5.3 Automated visual identification

For non-specific traps, identification of the target species is required. There are two main methods by which this can be achieved: direct visual assessment of the target or identification via wingbeat frequency. Visual assessment relies on a digital image of the catch being captured (reviewed by Barbedo, 2020; Preti et al., 2021). Similar to the examples above, Guarnieri et al. (2011) used a modified mobile phone integrated into a commercial pheromone sticky trap to monitor codling moths [*Cydia pomonella* (L.)] in apple orchards. Daily photographs of the sticky plate were sent from the trap to a remote server where they could be identified and counted on-screen, without disturbing the trap. Similarly, Rassati et al. (2016) used modified security cameras that automatically collected and sent images to an internet repository. The cameras were strapped to Lindgren funnel traps and used to monitor longhorn and bark beetles, with the intention to use them at seaport sites where there was a plant health risk. Such approaches rely on human identification of the photographs.

Increasingly, image analysis algorithms and machine learning are being used to automatically identify and count insect catches (Barbedo, 2020; Preti et al., 2021; Sciarretta and Calabrese, 2019; Xie et al., 2015); this is a rapidly developing area. In a review paper, Hoye et al. (2021) argue that computer vision and deep learning, a subset of machine learning, have tremendous potential to automate high-throughput systems for the identification of insects. In another review paper, Gaston and O'Neill (2004) discuss some of the limitations to automation of insect identification, both technical and cultural. Ding and Taylor (2016) provide a useful overview of the image analyses techniques used to identify insects, including analyses of wing venation and structures, colour histogram features and morphometric measurements. However, they point out that whilst some systems will work in a laboratory set-up, automatic identification of field trap catches has significant challenges related to poor-quality images, trap movement, contamination with dirt and leaves, decay and distortion of the specimens. Nevertheless, by using a sliding window-based detection pipeline with a convolutional neural network approach, they were able to identify codling moths from pheromone sticky traps. Similar deep learning approaches have been used for the identification of red turpentine beetle (*Dendroctonus valens* LeConte) from pheromone traps in pine forests in China (Sun et al., 2018), whereby the detector and processor are embedded in the pheromone trap, with the ultimate aim to develop traps as part of an Internet of Things monitoring network. Sun et al. (2018) also give a useful review of the use of image-based pest detection methods, which have been practically applied.

5.4 Automated identification via wingbeat harmonics

Flying insects can be identified by measurement of wingbeat harmonics (Moore et al., 1986). Wingbeat frequency is usually inversely proportional to the body mass and particularly the size of the insect wing. This, plus other characteristics such as wing shape, stroke and number, can result in a unique wingbeat harmonic signal that is phylogenetically clustered (Tercel et al., 2018). Whilst differences in wingbeat frequency can be detected acoustically (for example, compare the different sounds of a mosquito and bumblebee), for trapping purposes, detection is usually by optoacoustic means. Light scattered off beating wings can give a unique signal. Moore and Miller (2002) demonstrated that an optical sensor and data acquisition system could identify alate aphids passing through a halogen lamp light source. They specified that wingbeat waveform derived from a trained artificial neural network approach rather than wingbeat frequency alone provided the most discriminatory signal, as, for instance, wingbeat frequency altered with ambient temperature.

Potamitis et al. (2014) developed a prototype electronic McPhail trap to monitor olive fruit fly *Bactrocera oleae* (Rossi). The key component was an optoelectronic sensor comprising infrared-emitting LED coupled with an array of five phototransistors (Fig. 11). As olive fruit flies flew upwards into the McPhail trap through the ventral entrance, they passed through the infrared beam generating an analogue signal that was sent to a microcontroller, which enabled counting. Trigger events (insects breaking the beam) were stored and sent by text message from a GSM expansion board at predefined intervals to a remote operator. Potamitis et al. (2015) further developed this trap to include GPS, temperature and humidity monitoring. More pertinently, they were able to identify olive fruit fly by their wingbeat harmonic signature, with all processing conducted by the on-board electronics in the trap. The fruit flies only take 30–50 ms to cross the infrared beam. The photodiodes (replacing the earlier phototransistors) detect the minute fluctuations in the infrared beam, and the

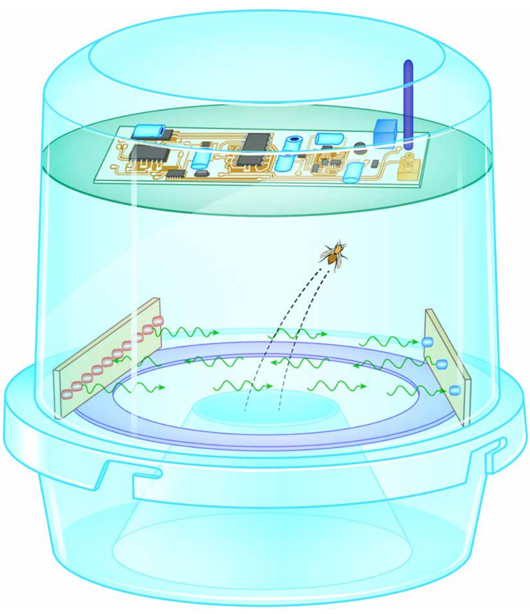

Figure 11 Schematic for an electronic McPhail trap that counts and identifies olive fruit fly. As the flies enter the trap, they pass through an infrared beam, and fluctuations in the beam caused by the flies' wingbeats can generate a unique signal that can be used to identify them. Source: Image from Potamitis et al. (2015) under the Creative Commons Attribution (CC BY) licence. No changes were made to the figure.

signal is processed by Fourier transform, which decomposes the waveform into its constituent frequencies. The wingbeat frequency alone is insufficient to discriminate between species, but the associated wingbeat harmonics can. Potamitis et al. (2015) provide the analogy of musical instruments playing the same note (e.g. frequency), but the human ear can distinguish between say a flute and a violin by other harmonics produced by the instruments. Wingbeat frequency spectra in the trap are compared with those collected in the laboratory, using embedded software. In laboratory trials, the trap was able to discriminate well between *B. oleae* and dissimilar insects such as mosquitoes, non-biting midges and honeybees (>90% accurate) but was less accurate with flies whose wingbeat spectra overlapped.

6 Conclusion

The term precision agriculture is used to describe enhanced temporal and spatial targeting by which crop inputs are applied. To incorporate integrated pest management principles within a precision agriculture framework requires an accurate assessment of pest numbers, preferably before any damage to the crop. Monitoring of pest species occurrence, activity and density is an essential part of integrated pest management. This allows crop protection decisions to be made using economic thresholds, which are a cost/benefit analyses of whether to apply a control measure. In theory, economic thresholds should be flexible and reflect market value of the crop and the cost of any control measure, including machinery costs, labour and fuel. They should also take cognisance of any indirect effects of application, in particular, any effects on natural enemies within the crop that are already exerting a level of pest control. In practice, economic thresholds are often fixed and dated and tend not to be based on published evidence. Furthermore, the method of pest density assessment in relation to the economic threshold must be practicable (Ramsden et al., 2017).

Pest monitoring via trapping falls into three broad categories. Regional-scale monitoring, often for forecasting purposes, utilises specialised traps and experts to process trap catches. The Rothamsted suction trap network is an exemplar of this approach, with aphid survey bulletins freely available to farmers. Another approach involves in-crop monitoring of selected crops as regional sentinels, also with expert trap processing. An overview of pest threat within a region can then be disseminated to growers. Finally, there is on-farm, in-crop monitoring. This is the most accurate method of pest assessment as it relates to the individual crop and therefore can be directly linked to economic spray thresholds. However, to be successful, in-crop monitoring relies on traps being robust, easy to use and easy to process. The trap should be selective, and whilst colour and light act as attractants and are somewhat discriminatory, selectivity has mainly been achieved using semiochemical lures. Since 1959, when the first pheromone was identified, knowledge of insect chemical ecology has expanded to the point where synthetic lures, either pheromones or kairomones, are available for many if not most major pest species. Use of semiochemical lures can reduce bycatch, meaning that most specimens are of the target species, reducing the need for expert identification.

Clean trap catches also make automatic counting more accurate. Whilst trap automation for time-sorting has been used since the 1930s, recent advances in the development and costs of digital cameras and microelectronics mean that automatic counting is now a practical possibility. There are two main ways to count insects in a trap: by image capture of trap catches or by incoming insects disrupting an infrared beam. More ambitious is to both count and identify trapped specimens. Again, the two main approaches are image analyses or identification via wingbeat harmonics derived from reflected infrared beams. Most of the developments in this area have been experimental, and few are adopted commercially, as yet. This is because the technology is still developing but also because the cost of 'smart traps', both purchasing and consumables (e.g. batteries), is considerably more per unit than the basic trap, which is likely to be 10–100 times less expensive, although substantial savings can be made in labour costs. However, a number of entrepreneurial companies are developing traps with an aim to full commercialisation (e.g. FaunaPhotonics www.faunaphotonics.com, DIOPSIS www.diopsis.eu).

The research that is ongoing in trapping technology seeks to improve and refine trap catches whilst crucially minimising processing time. Automated 'smart traps' that count and identify pest species have the potential to substantially increase data collection from crops. This will enable more informed decision-making about insecticide applications. Instead of all-crop spraying or prophylactic applications, control measures will only be targeted at crop areas that need them and only when justified by economic spray thresholds. Such an approach has clear environmental benefits in terms of reduced impact on non-target organisms and potential pollution of waterways and prolongs the life of active ingredients by reducing the conditions for insecticide resistance to develop.

7 Future trends in research

There are many trap designs for monitoring insect pests, and their improvement is a continuous process. Identification and refinement of semiochemical lures will continue as new pheromones and kairomones are identified and synthetic blends improved. New technologies are being utilised to dispense pheromones at a more natural rate, prolonging the life of the lure (Heuskin et al., 2011; Leskey et al., 2021; Muñoz-Pallares et al., 2001). There is also a need to determine the range and accuracy of trapping systems utilising both olfactory and visual responses and relate these to damage on the crop. Whilst this is a basic requirement of an economic threshold approach, it is difficult to quantify trap efficacy in the field, and mathematical approaches coupled with field validation are required.

Trap automation holds great potential. Technologies that are used to count and identify pests on-trap or through remote connections have been developed and will become more refined as technology advances. The use of image and wingbeat analyses using machine learning is at the forefront of these techniques, and there is great scope to advance these methods further. One possible scenario is that traps will connect through an autonomous network, proving real-time data on pest migration to a region or crop. It is also hoped that such monitoring technologies could provide data on the presence and densities of pest natural enemies within the crop, which would provide a holistic monitoring system to feed into DSSs.

Some of the technological developments for insect trapping of human–pathogen vectors could also be utilised in crop protection. Vector biologists have been exploring the use of unmanned aerial vehicles (or drones) to operate traps in field (Faraji et al., 2021; Vacek et al., 2017). For example, traps may be positioned by drones into marshy, inhospitable or dangerous areas. Drones could be used to collect trap contents and deliver physical samples to a collection point for identification or further analyses; or the drone itself is developed into a flying trap that can move easily from location to location (Ye et al., 2020). The Microsoft Premonition project seeks to develop a network of robotic smart traps to monitor mosquitoes to aid in forecasting disease outbreaks. Using wingbeat identification, the traps capture only pathogen–vector species and retain them for detection of any possible pathogens. In addition, these traps are continually monitoring the environmental conditions related to individual mosquito capture, with the potential to build up an immense dataset for modelling mosquito populations (Choney, 2020; Microsoft, 2022). Some approaches looking at disease prevalence do not require capture of the insects per se, but rather their saliva or faeces are collected from traps, which subsequently can be processed for pathogen identification (Hall-Mendelin et al., 2010; Meyer et al., 2019). Such techniques may be of value in monitoring pathogens vectored by phytophagous insects, particularly in cases where insect-feeding damage is not economically important, or surveillance for an invasive plant health pathogen.

If we look to the future, perhaps we have the potential to produce a network of solar-powered, autonomous, interlinked traps sited in individual crops but providing data for a regional dataset. Such traps will be baited with pest and natural-enemy-specific semiochemicals, with automated counting and identification. Real-time data on pests, natural enemies and environmental conditions will be supplied through the mobile phone network to a central server where DSSs linked to market value will calculate the risk to the crop with respect to the economic threshold.

8 Where to look for further information

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