MONITORING INSECT POPULATIONS

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INTRODUCTION

Monitoring a population of insects, whether beneficial or pestiferous, is a prerequisite for making a management decision, and thus monitoring serves as the fundamental tool for insect pest management. The purposes for monitoring insect populations are diverse, from detecting the spread of an insect into a quarantine area to estimating the number of insects per plant and relating this to an economic or action threshold. Because of this diversity, it is imperative that the monitoring technique be appropriate for the specific purpose. Monitoring the insect population may be done during a specific time of year, for example, to detect the emergence of an overwintering generation so that an appropriate control strategy may be invoked. At the other extreme, monitoring may be required on a weekly basis, such as with food crops for which an economic or action threshold with an appropriate sampling method has been developed. The development and use of monitoring techniques are also essential in order to study phenomena of insect ecology such as colonization processes, epizootiology of insect pathogens, and biological control by natural enemies. Whatever we wish to know about a population, whether it be to conduct a survey of its occurrence, monitor for insecticide resistance over a wide area, or study its host range, we can only learn about the insect population by collecting samples form it and making inferences from our samples. In all cases, the purpose of monitoring is to gather information on the status of the insect population within a given period of time and space.

The complexity of insect behavior often allows entomologists the opportunity to employ a diverse set of monitoring tactics, such as the use of kairomones, pheromones, or visual traps. However, even with these tools, we often are left with only a glimpse of the insect population we wish to monitor. This occurs because all of our insect monitoring techniques fail, to a greater or lesser degree, to provide us with information on the true population we wish to study. While we may be able to obtain an exact or absolute insect count on a discrete sample unit, such as a single plant or volume of soil, the labor involved in such an effort will limit the number of samples which can be taken and, hence, the inferences that can be made about insect densities on nearby plants or adjacent soils. Additionally, even in these intensive sampling efforts, we must rely on the accuracy of our procedures, such as extraction of insects from the soil. Thus, although we wish to have information on the true population, we are forced to take smaller collection (samples) and use these to make inferences about the population.[1] Fortunately, the discipline of statistics provides us with guidelines which help direct our inferences.

In this chapter we hope to give an overview of techniques and applications of monitoring in insect pest management. A complete treatment of insect monitoring here is not possible, and the reader is referred to Southwood[2] for additional details and references. This chapter is divided into two main topics: the first outlines how monitoring is being utilized in insect pest management, and the second provides an overview of statistical considerations involved in developing sampling plans.

In considering how monitoring is utilized, it may be helpful to divide the first part of this chapter into the somewhat arbitrary classifications of regional and localized monitoring. We consider it helpful to make this distinction because often different questions are being examined on the various scales. For example, on a regional basis one might try to determine what environmental or habitat factors are influencing insects to migrate within a relatively large area; while on a more localized basis, one might examine how insects are colonizing

plants in a particular field or how insects are responding to specific phytochemicals in certain parts of plants and changing their within-plant distribution accordingly. However, regardless of the scale on which we are monitoring insects, the tools which are available for this purpose are often similar.

SAMPLING TOOLS

The tools that we can use to monitor insect populations are diverse, and the reader is again referred to Southwood[2] for more details. Aerial sampling can utilize suction traps, sticky traps with or without an attractant, light traps, intercept traps, or a diversity of other active or passive traps. Passive traps, such as suction traps or rotary traps, allow unbiased estimates of flying populations because insects are neither attracted nor repelled by the traps.[3] Such estimates can be used to document movement of insects into an area, such as the Rothamstead trapping system in England which uses suction traps to monitor aphid flights from the continent.[4] Counts from these traps are then used to warn growers of impending flights. Traps designed to attract insects with a light source or pheromone are better detection tools although they give biased counts of insect density per unit area. Pheromone traps or traps baited with plant volatiles (e.g., the apple maggot trap which utilizes synthetic apple volatiles as well as a red-colored sphere for visual attraction[5,6]) are examples of active aerial traps. Active and passive traps may be modified to automatically sample during specific time intervals to obtain information on temporal movement patterns.

The most direct measure of potential injury to an agricultural crop will arise from an assessment of the population on the crop rather than aerial samples. However, crop sampling also has its limitations and inherent difficulties. Variation in the population within the field, between plants, and even within the plant makes precise population estimates difficult. Additionally, the cryptic nature of some insects or stages of insects makes them difficult to detect. Direct counting of the insects can be done on a plant basis or area/habitat basis. In the latter case, a quadrant can be used to delineate a certain area or habitat, and then all the insects within that area are counted. Counting can be done by enumerating the insects *in situ*, using a suction apparatus[7,8] dislodging the insects onto some type of counting cloth or tray by beating the foliage or spraying it with a chemical, or removing the foliage and then extracting the insects. Extraction of insects from foliage can be done through using heat in a Burlese unit, using vapors of several chemicals as is commonly done with thrips,[9] brushing foliage with automated devices such as mite brushing machines,[10] or washing the foliage with various solutions or solvents depending on the particular insect. In the case of extracting insects from foliage by washing, the insects may then be floated to the surface or filtered through screens for the actual counts.

REGIONAL MONITORING

QUARANTINE

A primary use of regional monitoring is to detect exotic insects which may pose a threat to agriculture. In the U.S. the Plant Quarantine Act of 1912 was enacted to control the artificial introduction of exotic pests into the country. The reasons for adoption of the quarantine strategy are obvious when one considers that many of the major agricultural pests in the U.S. originated from outside its borders. For example, the boll weevil, *Anthonomus grandis* Boheman entered the U.S. in the late 1800s and has caused billions of dollars in losses to the cotton crop.[11] Subsequent to this Act, additional quarantine measures have been enacted on the federal and state levels to prevent the introduction and interstate and intrastate movement of insects. Monitoring pests for quarantine purposes usually involves the inspection of plants or plant products at airports, seaports, or roads. In some cases

specific plant species are not allowed to be imported because the country of origin has a particular pest problem and the probability of insects entering on plants from that area is high. In the remaining cases the plants may be inspected using statistically designed sampling surveys such as for the light brown apple moth on New Zealand apples.[12] The California Department of Food and Agriculture (CDFA) operates a major quarantine program because of the importance of agriculture in the state. Plants as well as vehicles are inspected for insects, and the results are published quarterly in the CDFA bulletin.

The use of semiochemicals (pheromones and kairomones) has provided quarantine personnel with an efficient method of monitoring for pests. The gypsy moth, *Lymantria dispar* (L.), which was introduced into the U.S. at Medford, MA in 1869, still only occupies a relatively small portion of its potential host range and since 1912 has been subjected to quarantine measures. With the synthesis of disparlure, the male sex pheromone of the gypsy moth, it has been possible to monitor gypsy moth populations in infested areas and track their movement into new areas. The scale of the gypsy moth monitoring program has grown so that during 1979, nearly 95,000 pheromone traps were set in 38 different states to detect the presence of the gypsy moth.[11] Additional programs for monitoring quarantine insects using pheromones include the Japanese beetle, *Popillia japonica* (Newman), and the pink bollworm, *Pectinophora gossypiella* (Saunders). One of the most extensive quarantine efforts in recent years is the work in California on the Mediterranean fruit fly, *Ceratitus capitata*. Because of the potential loss to agriculture, a statewide program was initiated to detect and trap adults using a Jackson/trimedlure in thousands of trees throughout California.[13] Catches of flies in these traps has initiated area-wide aerial spraying and sterile release programs. CDFA currently budgets $7 million for its fruit fly trapping which includes sex lure traps as well as sticky and bait traps.[14] In all these cases the purpose of the quarantine and monitoring program is to prevent the movement of these pests from one area to another.

PEST SURVEYS

Pest surveys, systematic and usually repeated surveys over a wide area, may or may not be conducted in connection with quarantine programs. These large-scale monitoring projects are funded by the state and federal governments, and the survey work is performed to detect new species or to document distribution and population trends of indigenous species. Examples of these surveys would be the Animal Plant Health Inspection Service (APHIS) funded projects and state surveys. In 1982, the APHIS-Plant Protection and Quarantine (PPQ) established the Cooperative National Plant Pest Survey and Detection Program (CNPPSDP) under the coordination of the U.S. Department of Agriculture-APHIS-PPQ. The program, now known as the Cooperative Agricultural Pest Survey (CAPS) was designed primarily to monitor exotic and endemic economic pests. The program has four types of surveys: exotic pest detection, beneficial survey, quarantine/regulatory surveys, and endemic pest survey. A major component of the CAPS program is the electronic data base (NAPIS) which is used for collecting and storing data in a standard form. In turn, NAPIS interfaces with all pest survey activities in each state.[15]

MOVEMENT SURVEYS

Large-scale surveys have been used for a number of different purposes other than quarantine or historical data bases. Such purposes include developing a better understanding of ecological, climatological, and biological factors which influence insect movement, as well as using this information to develop predictive models which can be used to warn growers of impending flights. Working groups, such as regional committees on the migration and dispersal of insects, have been formed in the U.S. to foster cooperation on understanding insect movement and monitoring pests. These working groups not only include entomologists, but also meteorologists and researchers from other disciplines who can lend insight

into developing methods which will help monitor insect movement. Studies on movement of *Spodoptera* spp.,[16] *Trichoplusia ni* and related looper species,[17] velvet bean caterpillar,[18] and *Heliothis* spp.,[19-20] have provided insight into the long-range movement of Lepidoptera. The classical studies on the influence of weather patterns on locust movement by Rainey[21] and his co-workers have predictive value in determining when and where locust outbreaks will occur in the Middle East and Africa.

EMERGENCE PATTERNS OR GENERATION PEAKS

Monitoring insect populations may be done for the purpose of determining emergence patterns or detecting generational peaks. Information derived from these studies can be most useful for helping to time further sampling schemes or to initiate treatment strategies. Sticky traps, water traps, and inverted cone traps are used most commonly for catching insects in the field.[22] A good example of the use of traps to monitor emergence patterns is provided by studies with the cabbage root fly. Adults lay their eggs at the base of cruciferous plants, and it is difficult and labor intensive to monitor populations by examining the soil around the plants. Trapping of adult flies in water pan traps or sticky traps provides information which can be used to predict oviposition, and control measures can be taken accordingly. Trap effectiveness can be augmented by adding various chemical and visual stimuli. Catches of cabbage root flies can be increased with extracts of host chemicals like isothiocyanates.[23]

Documentation of emergence patterns of generational peaks has been greatly aided by the use of traps baited with semiochemicals. When these traps are located over a wide area, they provide a relatively time-efficient method by which to follow the population as it changes from the pupal to adult stage. Baited traps are commonly used to provide information on emergence patterns of pestiferous insects from several orders including Diptera (e.g., apple maggot), Coleoptera (e.g., boll weevil), and Lepidoptera (e.g., codling moth).

MONITORING FOR INSECTICIDE RESISTANCE

Although this topic most properly fits under another chapter, monitoring adult populations by pheromone traps may also be used for monitoring insecticide susceptibility. Reidl et al.[24] developed a procedure which utilized pheromone traps to screen field populations of the codling moth for resistance to azinphosmethyl. Collected moths were assayed by leaving them in the polybutene adhesive and then applying a dose of azinphosmethyl to the thoracic dorsum or venter. This use of pheromone traps for monitoring insect populations was later modified to incorporate insecticide concentrations directly into the adhesive material of the pheromone traps for the pink bollworm[25] and the beet armyworm.[26]

LOCALIZED SAMPLING

INTERCROP MOVEMENT

Besides the long-range movement studies of Lepidoptera mentioned earlier, many serious insect pests move freely between crops within a more localized region, and the spatial aspects of population change can play a major role in the timing and intensity of pest outbreaks on certain crops.[27] To a large extent our ability to manage insect pests and reduce crop losses depends upon our capacity to monitor and forecast the pest population and determine the likelihood of an infestation in a specific area or field. In many of our agricultural systems, an insect may use a sequence of host crops over space and time, and thus an understanding of intercrop movement becomes essential. The knowledge that *Lygus* species developed in safflower and alfalfa and then moved to cotton, provided growers with the opportunity to use well-timed sprays to control *Lygus* on these "nursery" crops and avoid the more drastic damage which would occur on cotton.[28] A similar situation of pest movement between crops was also reported by Kennedy and his co-workers in North Carolina.[29,30] In their studies

they found that the two-spotted spider mite, *Tetranychus urticae*, overwintered in the vegetation along field borders on field corn; then in the spring crawled into the crops of adjacent fields where they developed large populations. As the crops senesced in July and August, the mite populations moved from the field corn to other high value crops such as peanuts, melons, and tomatoes. Similar movement patterns from low value crops onto high value crops also have been illustrated with onion thrips in New York. In this situation onion thrips overwinter in wheat and alfalfa,[31] in the early spring develop on these two crops and oats, and then leave these crops as they senesce or are harvested.[32,33] Monitoring of the insect populations can allow us to focus the major management effort on the nursery crop and reap the benefits from that effort in reduced problems on the other crops.[27] Often, however, there are economic constraints to implementing this logic, such as the low value of some nursery crops or the situation where different growers own the fields.

WITHIN-FIELD SAMPLING

When monitoring insect populations in the field, one must consider the purpose of the sampling because this will influence where the samples should be taken. For example, if one is trying to simply detect the presence of an insect, it would be advisable to take samples only from those areas which, through previous experience and an understanding of the insect ecology, are more likely to be infested. If, on the other hand, the objective is to obtain an overall estimate of the field population, then a representative sample which minimizes bias is desired. The classical approach is to randomize some aspect of the sampling procedure to eliminate sampling bias. Random samples can be taken using a number of different strategies.[2] In unrestricted random sampling, the sites to be selected for sampling can be defined by using random numbers tables. While this method will eliminate sample bias, it may not be efficient for minimizing the variance and may lead to an inefficient use of time in the field. Other options are stratified random samplings in which the area is divided into a number of equal-sized subdivisions or strata, and one sample is selected from each strata. An alternative to random sampling is systematic sampling. In this case samples are taken at fixed spatial intervals. This approach has the advantage of determining patterns of infestation in the field which can be incorporated into sampling methods. With the use of today's computer graphics programs data from systematic sampling can be easily inspected for such patterns. This approach has been used successfully with many crops including European corn borer infestations in sweet corn,[34] onion thrips on onions,[35] and onion thrips and Lepidoptera on cabbage.[104]

The consequences of within-field distribution of insects is especially apparent in the case of aphid and leafhopper-vectored plant viruses. Both insects are capable of spreading the viruses over long distances, but the distance a particular virus is spread from a source is related to the dispersal of the vector and the persistence of the virus in the vector.[36] With nonpersistent viruses, which are retained in the vector for only a short time, the pattern of infection within a field typically shows a strong initial border effect as a result of insects first landing on the hosts along field margins, inoculating these hosts, and losing the virus in the process. As flights into the field continue or as secondary infection spreads, the strong initial border effect diminishes.

WITHIN-PLANT DISTRIBUTION

Sampling costs can be improved by reducing the time expended in estimating population densities. One common approach used to achieve this goal in agricultural systems is to subsample. In terms of within-plant distribution, subsampling refers to assessing arthropod presence or numbers on only a portion of a normal sample unit. For example, Harcourt[37] reported that the Colorado potato beetle population on tomato plants could be accurately estimated by counting only the basal third of the plant, rather than the entire plant. This

approach can also be used in binomial (presence-absence) sampling: two-spotted spider mite populations, *Tetranychus urticae* (Koch), can be rapidly appraised by determining the percent of plants infested on the uppermost fully expanded trifoliate.[38] Such sampling plans are possible because a predictive relationship has been established between whole plant counts and subsamples. Thus, sampling plans using subsamples require substantial background information and are generally the result of considerable research effort.

Not all subsampling is based on visual estimation of populations. Much of the common equipment used for arthropod sampling simply provides subsamples of plants. Sweep nets and devices used for suction sampling are good examples. These typically remove only that portion of a population on a plant that is near the top or accessible to the equipment. As in the population estimates based on percent infestations or visual counts, assessments resulting from the use of such equipment will be affected by changes in population distributions within plants.

Subsampling plans based on within-plant distribution are often complicated by changes in distribution over time. Physiological and physical changes in plants as they age are frequently implicated as primary factors affecting preferred habitats within plants. Kennedy[39] noted that apex leaves, as sites of protein synthesis, and the oldest leaves, which are typically undergoing leaf proteolysis, were frequently the preferred sites for aphid attack because of high soluble nitrogen levels. This hypothesis has been generally supported by observations on *Brassica* species indicating that *Myzus persicae* (Sulzer) prefers the oldest foliage and *Brevicoryne brassicae* (L.) prefers the youngest leaves.[40,41] However, the distribution has been shown to change with plant age; no differences in locational preference are noted for physiologically homogeneous small plants[42] while *B. brassicae* prefers the oldest leaves on flowering plants.[43] Similarly, secondary plant chemistry, which influences within-plant distributions, is also known to change with plant age as well as between seasons.[44,45]

Plant resistance factors, whether they be caused by chemical or morphological characteristics, can alter the distribution of insects and hence affect sampling. Different concentrations of plant chemicals can be expected to change the within-plant distributions of insects such as in the case of DIMBOA, a resistance factor for European corn borer in maize, which varies within the plant, with the highest levels in root, stalk, and whorl tissues.[46] In the case of cabbage lines which have been documented to be resistant to onion thrips, it was observed that both susceptible and resistant lines had similar numbers of thrips, but the distribution of thrips within the plant was different: the susceptible lines had the majority of thrips in the head and lower populations on the frame of leaves, and on the resistant lines the situation was reversed.[47]

Environmental variation alters the locational preferences of arthropods, and diel patterns in temperature and/or humidity can affect the within-plant distribution of beetle larvae[48] and cabbage loopers.[49] Diel variation in light intensity has been correlated with pea aphid sampling by sweep nets.[50] Similar circadian patterns in sticky trap catches of males and females of *Liriomyza* species also have been reported.[51] Frost patterns within plants, as well as rainfall and other weather-related effects, could reasonably be expected to affect the optimal within-plant locations for arthropod survival. In a related fashion, some locations within plants could be expected to be more protected from pesticides, thereby allowing populations to survive in specific protected areas within the plant canopy. Thus, pesticide applications could alter the relationship between plant subsamples and whole plant counts, thereby affecting the accuracy of the subsampling procedure.

An insect's movement patterns between fields, within fields, and within plants will influence our ability to detect the insect or estimate its density. Thus, an understanding of the insect's ecology and its interactions with surrounding habitats, the environment, and the plant will aid greatly in designing sampling programs that are realistic and effective aids for making pest management decisions.

DEVELOPMENT OF A SAMPLING PLAN

Regardless of whether a sampling plan is being developed for a region or a more localized area, the plan should be constructed with the following factors in mind: precision, efficiency, and the value of the information which is collected. Precision and efficiency can be addressed through statistical procedures, but determining the value of information has been more of a qualitative factor. Recent papers[52,53] have tried to evaluate sampling information in integrated pest management (IPM) programs more objectively using criteria from decision theory. Based on a study of leek moth infesting leek in Holland[53] and European corn borer in New York,[105] it was found that intensive field sampling for leek moth before applying an insecticide was not economical, but that field sampling for European corn borer was. This points out the need to examine the economics of each crop so that appropriate sampling strategies can be devised.

SELECTION OF A SAMPLE UNIT

Choice of a sampling unit will vary with the information desired. Most frequently, sampling in agricultural ecosystems focuses on determining if pest or beneficial insect populations have reached a threshold level which requires action. For this purpose, several types of information are of particular interest: (1) estimates of a population density per unit area, (2) assessments of percentage infestation or parasitism, (3) estimation of damage per unit area and, less typically, (4) absolute population counts. However, regardless of the type of information desired, certain general considerations are valid.

A variety of criteria for selecting a sample unit have been suggested in the literature.[54] Although not all of the suggested requirements are practical in every situation, many of the recommendations have broad applicability. Ideally, a sample unit should meet most, if not all, of the following requirements.

Requirement 1

Each sample should have an equal chance for selection. Unfortunately, utilizing a completely random selection process in a large agricultural field often requires an excessive amount of time and effort on the part of the sampler. As a result most fields are surveyed in a prearranged pattern (e.g., U-, V-, or X-shape), with samples collected periodically along the transects.[55] However, this approach effectively can include a random factor by selecting the interval between samples by chance. Random numbers can be found in published tables,[56] or generated by pocket calculators, dice, last few numbers on the odometer of your vehicle, numbered slips of paper drawn from a hat, etc.

Inclusion of a randomizing factor is critical for preventing errors that are associated with arbitrary sampling. Typically, samplers who select plants arbitrarily introduce some bias into the program. For example, some individuals will preferentially select samples which show signs of feeding damage or stress. These samples often have a greater probability of containing pest arthropods, thus skewing the data toward higher population densities. As samplers become tired or are pressed for time, some will unintentionally begin to select samples which are less likely to require substantial counting or recording time, thereby inadvertently producing low population estimates.

Requirement 2

The proportion of an insect population using the sample unit as a habitat should remain constant throughout at least each sampling event. If the organism to be sampled changes within-plant location during the course of the day, then population estimates will vary accordingly. For example, the largest instars of the beet armyworm, *Spodoptera exigua* (Hübner) (Lepidoptera: Noctuidae), are negatively phototactic.[57] Foliar sampling in celery

during midday invariably produces lower counts than during crepuscular periods because the larvae have either moved into the center of the plant where detection is unlikely, or they have left the plant and hidden underground. In an analogous fashion, larvae of the cabbage looper, *Tricoplusia ni* (Hübner), change location within cabbage plants in response to temperature[58] or distribution in relation to light and gravity.[59] Therefore, sampling only a subset of outer leaves on the plant may not provide consistently accurate population estimates. A wide variety of arthropod preferences and behaviors following circadian rhythms which would affect larval, pupal, and adult sampling have been observed for plant pests and beneficial arthropods.[60-63] Besides these behaviors, the age structure of the population will affect biological monitoring practices. Populations in which the age of the individuals is distributed with respect to time will have only a portion of their numbers subject to sampling at any point in time by any method which does not sample all age classes.[64] Thus, precise estimation of populations requires that either the sampling unit be independent of diel periodicities, that samples be collected during specific times or environmental conditions, that a "correction factor(s)" be determined for samples from a habitat collected over time, or that the age structure of the population be taken into consideration.

Additional variability in population estimates will occur if the sampling technique becomes more or less efficient due to environmental conditions which vary over time. Sweep net sampling is often less proficient at collecting arthropods early in the morning if the plants are laden with dew. Techniques, such as suction sampling, which often rely on escape behaviors to maximize collections, may have reduced efficiency in the cooler periods of the day when insects are less active.

Requirement 3

There should be a reasonable balance between the variance produced when data are collected from a given sample unit and the cost (time, labor, and equipment) inherent in assessing that unit. Generally, a preferred sample unit would be the minimum size which allows an adequate number of replications on a given date to produce averages with a meaningful (i.e., useful) variance. Sampling all the leaves on a plant would provide accurate information for each plant, but if only a few plants could be sampled in the time available, the population estimate for the field could be dramatically inaccurate. A better procedure would utilize a subsample of leaves from the plant, with more samples collected from each field. For example, green peach aphids, *Myzus persicae* (Sulzer) (Homoptera: Aphididae), are most efficiently sampled by monitoring the preferred oldest leaves of broccoli plants.[65] Whole plant counts would require surveying the upper and lower surfaces of up to 23 leaves per plant, when the population could be reliably estimated (coefficient of determination = 0.92) from counts on the lower surfaces of the oldest leaf. The increase in numbers of samples possible with the same amount of effort (cost) provides the additional advantage of increasing the probability of finding initial or clumped infestations of arthropods or occurrences of damage.

Requirement 4

Whenever practical, the sample unit should be as close to the "natural habitat unit"[66] as possible. This can be defined as the area within which an insect is likely to spend most of its time in a given developmental stage. For example, in many agricultural settings the individual plant or specific locations on the plant are the natural habitat units for pest species. Only rarely do aphid nymphs or early instar lepidopterous pests move between plants, and the pupae of most insects are entirely sessile. Once settled, many scale insects lose their ability for movement. Of course, there are many exceptions, including the armyworms and other highly mobile species, but repeatedly sampling individual plants or subsamples of plants for damage can overcome even this problem.

The primary weakness of this requirement occurs when one attempts to sample arthropods which have no discreet habitat (e.g., a continuum of plants such as grasses, or even row crops which grow to produce single canopies in which individual plants are difficult to identify). Many larvae feed underground on the roots of grasses and other plants which may not provide a distinct habitat unit. In these cases, arbitrarily chosen quadrats or other artificial plot selections need to be utilized. Selection of the size and shape of the quadrats then becomes dependent on the amount of effort needed to sample each, the numbers that can be sampled in a given time period, the randomization of variances or what variances are generated, and whether the locations can be adequately randomized.

DETERMINING THE NUMBER OF SAMPLES REQUIRED

In general, a large number of samples improve the reliability of an estimate of a population density. However, because sampling can be expensive in terms of time and effort, population estimates are often made with as few samples as possible which will provide a given level of reliability. Karandinos,[67] Li,[68] Ruesink,[69] and Snedecor and Cochran[1] describe in detail the formulas needed to calculate optimum sample sizes using reliability defined by the coefficient of variability and by formal probabilistic statements. The coefficient of variability is the simpler and more readily accessible approach. The formula for calculating the number of samples required for a given level of variability is

$$n = \left(\frac{\sigma}{\mu \cdot c.v.}\right)^2$$

where n is the number of samples required, μ is the unknown mean of the sample, σ is the variance, and c.v. is the coefficient of variation of the mean, defined as:

$$c.v.(\overline{X}) = \frac{\sigma/\sqrt{n}}{\mu}$$

For practical purposes, this technique requires that a series of samples be collected, and from these some preliminary estimates of the mean (= Xe) and the variance (= Se) can be included in the revised formula:

$$\hat{n} = \left(\frac{Se}{\overline{X}e \cdot c.v.}\right)^2$$

Selecting an appropriate level for the coefficient of variation will vary with the degree of reliability desired. Increasing reliability by decreasing the coefficient of variation will obviously increase the numbers of samples required. Finding a balance between the numbers of samples economically possible in a field survey program and an acceptable level for the coefficient of variation is often one of the more difficult decisions a sampler must make.

Such calculations of the numbers of samples required should be made repeatedly during the season. Many field scouts and scientists assume that the number of samples required does not change during a crop season or between years. Actually, there is compelling evidence for a need for sample size modification with variation in larval size, as population density changes, and with alterations in within-field distribution of populations. Use of this incorrect assumption adds substantially to the errors incurred in population estimation. Examples from the literature, as well as some additional techniques for estimating population sizes with fixed levels of precision, are presented in the next section of this chapter dealing with population distributions.

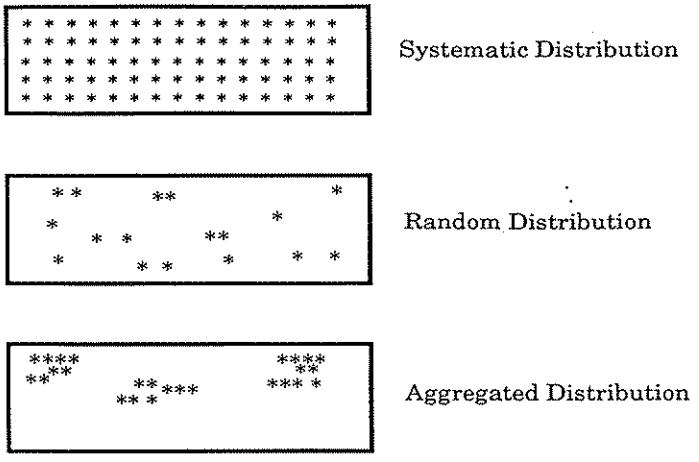

FIGURE 1. Potential spatial distribution patterns of arthropods.

WITHIN-FIELD DISTRIBUTION OF ARTHROPODS

Arthropod populations can assume one of three spatial patterns within fields: systematic distributions, random organization, or aggregations (see Figure 1).

As populations tend toward aggregation, the coefficient of variation of the mean increases, requiring that more samples be collected to achieve a given level of reliability (see the previous formula). Measuring such distributions has therefore been the subject of considerable study. Not surprisingly, relatively few arthropod species have been reported to have systematic distributions. Some ant colonies may appear regularly distributed as a result of territorial or foraging activity, but even apparently homogeneous environments such as pastures and row crop fields are usually heterogeneous enough that populations are not regularly distributed over wide areas. As noted by Walter[70] and Taylor,[71] even random distributions are rare. Aggregated distributions are by far the most common reported in the literature. These clumped distributions can be caused by a variety of factors in agricultural systems, including heterogeneous within-field environments, protection or self-defense, mating, feeding, and oviposition patterns.[2,72]

Several approaches have been suggested for analyzing the spatial distributions of arthropods. These fall into three broad categories: the theoretical models of dispersion, the indices of dispersion, and the regression-based indices. These approaches and some related concerns are discussed briefly in this chapter specifically in relation to sampling; for computer programs for distributional analyses to produce sampling plans, the reader is referred to the chapter authored by Nyrop and Binns.

The theoretical models of dispersion, such as the negative binomial,[73,74] Poisson,[75] and the Neyman type A,[76] can be powerful tools for determining how arthropods are distributed between sample units. In addition, these models can be used to develop sampling programs; the negative binomial has been used extensively.[77,78] However, these models do have significant limitations, many of which are discussed in detail by Taylor.[71]

One of the more substantial problems associated with the theoretical models is that, depending on how the sample units are structured, the same data can be made to fit either the random or aggregated distribution pattern.[79,80] Typically, data sets incorporating arthropod counts do not include samples which have all possible counts from the lowest to the highest numbers recorded. For example, if aphids are counted on 100 host plants and the maximum number recorded is 100 aphids per plant, there is a high probability that plants will not be observed that have exactly 57 aphids, etc. The presence of these "blanks" in the data cause mathematical problems with the analyses. A common procedure for eliminating this difficulty

is to segregate the count data into frequency classes. Thus, all counts of 0 to 5 fall into class no. 1, counts from 6 to 15 fall into class no. 2, and so forth. Unfortunately, the use of frequency classes presupposes some biological significance of the groupings chosen when, in fact, the segregation is often based on statistical needs. Because of this, the same data set can be made to fit a variety of the theoretical models depending on how the data are segregated into the frequency classes.

Partly in response to this problem, a variety of dispersion indices were developed which are not dependent on frequency classes. Examples of commonly reported indices include the mean/variance relationship, Green's coefficient,[81] Morisita's original[82] and standardized indices,[83] and Lloyd's patchiness index.[84] Each of these indices has advantages and disadvantages in certain circumstances, and the reader is referred to Myers[85] and Trumble et al.[86] for additional information. These techniques provide information on the distribution of organisms, but have not been specifically adapted to the development of sampling plans. However, one interesting use which has evolved is the practice of examining changes in arthropod dispersion over time and among locations or crop years to determine if the regression-based indices can be validly used for a given data set.[86-88] Although the regression techniques can themselves be used to document such variation over time, there is considerable value in showing agreement among several indices.[85]

The two most commonly reported regression-based indices are Taylor's power law (TPL)[89,90] and Iwao's regression of mean crowding on the mean.[91-93] These procedures have been used by other researchers to develop sequential classification or estimation plans.

The key parameters of Taylor's power law, $s^2 = am^b$, are estimated from the regression equation:

$$\log (s^2) = \log (a) + b \log (m)$$

where s^2 is the variance, m is the population mean, and a and b are the intercept and slope coefficients, respectively. Student's t-tests then can be used to test the hypothesis that the "coefficient of aggregation" (b) = 1, or if the log (a) = 0. According to Taylor,[90] if b is significantly greater than one (unity), then the population is aggregated. If b is not different from one, the population is randomly distributed. Values of b significantly less than unity indicate a systematic distribution. Taylor has suggested that the slope is characteristic for a species. In TPL, the intercept value is not given biological significance and is said to vary with the sampling technique employed.

Green[94] reported a method which utilizes the regression parameters of TPL to generate fixed precision level counting plans. These can be used to determine the cumulative numbers of arthropods which must be counted to estimate population densities at some given level of precision. The formula

$$\log \mathrm{T}n = \frac{\log (\mathrm{Do}^2/a)}{b - 2} + \frac{b - 1}{b - 2} \log n$$

calculates Tn, the cumulative number of organisms counted, where n = the sample size and Do = the fixed level of precision. Most reports either provide a range of precision levels or simply use a precision level of 25% (0.25), a value considered sufficiently accurate for use in pest management programs.[2] An example of a fixed level precision sampling plan is presented in Figure 2.

Finch et al.[95] rearranged Green's[94] formula to allow calculation of the numbers of samples required to estimate populations at various mean densities such that:

$$\log n = (\log a - 2 \log \mathrm{Do}) - (2 - b) \log x$$

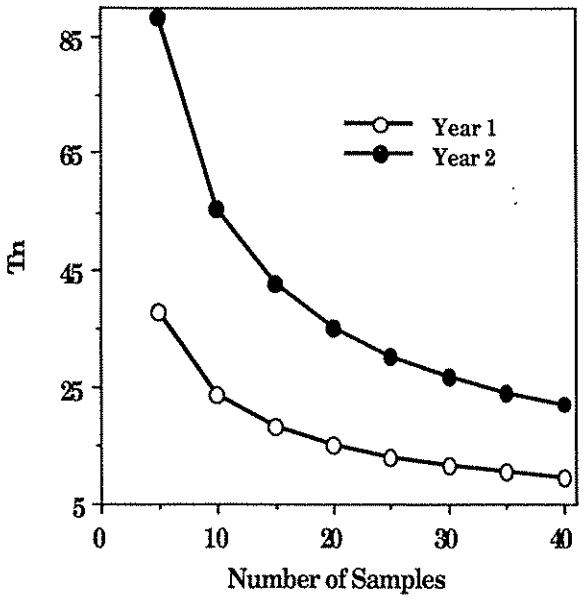

FIGURE 2. A fixed level sequential counting plan for a hypothetical population of arthropods generated using Green's formula. The intercept coefficients of Taylor's power law (TPL) used in this plan were year 1, $a = 1.05$ and year 2, $a = 1.75$. Slopes for both years were held constant at $b = 1.5$. The precision level was fixed at 25%. In practice, samples would be collected until the cumulative number of arthropods exceeds the stop line value for the number of samples collected. In this approach, the intercept (a) from TPL is given considerable statistical importance and can dramatically affect the numbers of samples required to estimate population density; the stop line in year 1 requires that twice as many arthropods be counted as in year 2.

where a and b are from TPL, $n =$ the number of samples, Do = a given level of precisions, and x = population density. An example of a sequential counting plan based on Finch's formula has been provided in Figure 3. Although both Green's[94] formula and the Finch et al.[95] revision are powerful tools which have proved valuable in many sampling programs, both are generally limited to use with populations which have distributions generating TPL slope values of less than 2.0.

Iwao's regression model follows the formula, $m = a + \beta m$, where a = the intercept on the ordinate $\mathring{m}$ = mean crowding, and β = the slope of the regression line formed when $\mathring{m}$ is regressed on the mean. Mean crowding is derived by solving Lloyd's[84] formula

$$\mathring{m} = m + (\sigma^2/m - 1)$$

and substituting in the x and s^2 from the count data. Student's t-tests can then be used to determine if a (estimated by a) or β (estimated by b) differ from 0 or 1. According to Iwao,[91] the "index of basic contagion" describes an individual as the basic unit of the population when $a = 0$, or a group of individuals as the basic unit if $a > 0$. The slope, which Iwao[91] termed the "density contagiousness coefficient", distinguishes how the basic population units are distributed within their habitat; distribution is random when $b = 1$ and becomes increasingly aggregated as b increases.

Utilizing the distributional information from the regression of mean crowding on the

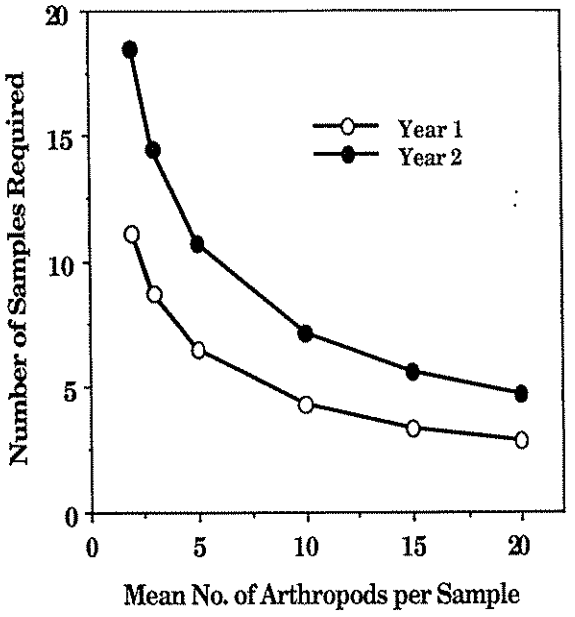

FIGURE 3. Fixed precision level stop lines for the required number of samples for arthropod population at various density levels. The intercept coefficients of Taylor's power law used in this plan were year 1, $a = 1.05$ and year 2, $a = 1.75$. Slope values ($b = 1.40$) and precision level values (Do = 25%) were held constant for both years. As seen in Figure 2, fluctuations in a can result in major differences in the sampling plan. Using the formula of Finch et al.,[95] a 60% variation in the intercept values between years resulted in at least 50% more samples being required in year 2.

mean, Kuno[96] developed a sequential sampling plan based on the concept of a critical density per sample (e.g., economic injury level). The upper and lower limits (see Figure 4) can be calculated using the formulae:

$$\text{upper limit} = nm_0 + t\sqrt{n\{(a+1)m_0 + (b-1)m_0^2\}}$$
$$\text{lower limit} = nm_0 - t\sqrt{n\{(a+1)m_0 + (b-1)m_0^2\}}$$

where, n = the number of samples, m_o = the critical density, t = the t value for some level of confidence, and a and b are the intercepts and slopes generated by Iwao's regression of m on the mean. Although this technique has been widely reported in the literature, a study by Nyrop and Simmons[97] demonstrated that the procedure may produce inaccurate upper and lower limits because the actual error rates can deviate substantially from those assumed by Iwao.[91] Therefore, they suggest that simulations be used to determine the correct error rates for inclusion in the model.

Binomial or presence-absence sampling plans developed by Nachman,[98] Wilson and Room,[99] and Ward et al.[100] require considerable input to develop a program, but generally offer substantial savings in time and effort in the field. In practice, these plans relate a given proportion of samples containing a given arthropod species to a mean number of arthropods per sample. For example, when sampling for aphids in cereal grains, the individual only needs to determine if a given proportion of tillers are infested to decide if a control action is required.[101] This approach can allow increased numbers of samples to be examined in substantially less time than programs which require that the arthropods be counted and

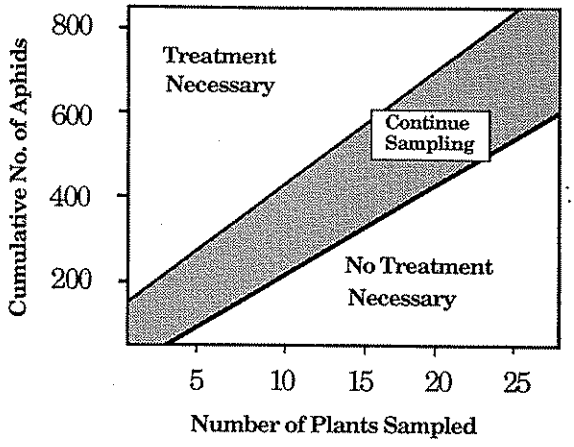

FIGURE 4. An example of Iwao's sequential sampling plan used for aphids on strawberries.[88] In application, randomly chosen plants are sampled in sequence, and sampling stops when the cumulative number of aphids fall outside the shaded area between the upper and lower limits. Theoretically, sampling could continue indefinitely, but some upper number of plants typically is chosen beyond which samples will not be collected.

tabulated. For a more complete discussion of binomial sampling, the reader is referred to the chapter by Nyrop and Binns.

All of the aforementioned distributional models and indices are powerful tools which can maximize a sampler's efficiency while minimizing effort. However, because arthropod distributions can vary (1) over time,[88,102] (2) with changes in population density,[71] (3) after pesticide application,[38,103] (4) among host plants, and (5) between geographic locations,[86] caution should be used in developing and recommending sampling programs. One of the more common errors in the literature is the collection of dispersion information from untreated fields and the subsequent development of sampling plans for fields which receive pesticide applications. Such plans ignore potential changes in distribution and may produce unintentionally conservative programs which result in excessive and unnecessary use of pesticides.

CONCLUSION

The goal of pest management is often defined as the reduction of the use of pesticides to the minimal level necessary to produce a crop of high quality. Since the information derived from monitoring a pest population is essential in helping to make decisions on the use of pesticides, monitoring pest populations is fundamental to achieving reduced pesticide usage. Population monitoring can be used to collect information in a number of different situations. It may be used to help prevent pests from becoming established (quarantine) as well as serving as a means for determining whether an economic threshold has been exceeded. In all of the different situations, however, it is essential to define the purpose of monitoring, quantify the value of sampling information derived from monitoring, and incorporate an understanding of the insect's behavior and ecology into the monitoring program. Knowing when an insect is emerging, when it will migrate from one crop to another, where it will land in a field, how it will disperse within that field, and how it will disperse on its host plant are essential elements in developing a precise and time-efficient monitoring program.

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