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QUANTITATIVE SAMPLING PRINCIPLES IN COTTON IPM

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Defining the Objectives of Sampling
Population Sampling (Parameter Estimation)
Commercial Monitoring (Decision Estimation)
Sequential Sampling

Defining the Sample Unit
Sample Method Efficiency
Sampler Efficiency
The use and economic benefits derived by adapting improved pest management approaches are covered in detail throughout this book. With few exceptions, the derivation of the biological relationships that went into developing the associated pest management components, the implementation of this information, and the benefits derived from these improvements were and are conditional on the availability of accurate, reliable, and easy-to-use monitoring procedures.

Most of the pioneering work in developing sampling techniques in cotton has been in the area of entomology. Although the fairly recent renewal of our national concern over pesticide-related problems has helped to promulgate the development of quantitative sampling procedures, few are presently available for assessing the abundance and impact of pathogens, weeds, and nematode species on cotton. This in part reflects the relatively short period of time that scientists in these disciplines have been involved with cotton IPM. Although specific sampling procedures are discussed throughout this chapter, the major emphasis is on principles of sampling. Although we will rely largely on the more readily available entomological examples, all basic principles and many of the procedures discussed are also appropriate for sampling nematode, pathogen, and weed species and for assessing the development of the cotton crop. For readers who require more detail on particular sampling procedures, Bohmfalk et al. (1983), Ellington et al. (1984a), Hamer (1980), Lincoln (1978), Lloyd et al. (1983), Smith et al. (1983), and Sterling and Lincoln (1978) review sampling of arthropods inhabiting cotton; weed sampling is reviewed by Baldwin and Santelmann (1980), Ellington et al. (1984a), Fay and Olson (1978), Flint et al. (1981), and Kirk et al. (1972); while Butterfield and DeVay (1977), Ellington et al. (1984a), Huismann and Ashworth (1974), Tabachnik et al. (1979), Toler et al. (1981), and Weinhold (1977) discuss various assay methods for sampling soil pathogens; finally, readers are referred to Barker and Campbell (1981), Ellington et al. (1984a), Ferris (1985), and McSorley (1987) for nematode sampling.
DEFINING THE OBJECTIVES OF SAMPLING

When sampling, the objective may be to understand and predict the distribution, abundance, and possibly the interaction of a population(s) with the host crop. Or, the objective may be to apply the information gathered to aid in managing a crop. The reason for the distinction is that population sampling in the first case and decision sampling in the second have different objectives (Ruesink and Kogan, 1982). The emphasis on population sampling is on the reliability of the parameter being measured, that is, the closeness of the sample mean to the true population mean (Sokal and Rohlf, 1969). Decision sampling emphasizes the reliability of the resulting estimate of the pest’s status, that is, whether the pest (or other group) is above or below the action threshold. It may not be necessary to have a highly reliable estimate of population density when determining whether or not a pest requires control, particularly if the density is far above or far below its action threshold (Fig. 5.1).

Figure 5.1 Comparison of confidence intervals for (A) population (parameter estimation) sampling and (B) commercial (decision) monitoring.
The objective of population sampling may be to estimate the number of organisms on an absolute basis (i.e., number per unit area). Since the results of this type of sampling may be used to derive functional relationships, a greater number of sample units may have to be examined to achieve sufficient reliability in the estimate (Fig. 5.2). Population sampling and decision sampling may also require different sample units. An accurate sampling method may not be necessary for commercial monitoring, particularly when estimates of the efficiency of sampling procedures at capturing the target species are known, enabling absolute density estimation (see the sample unit section).

Failure to distinguish between the objectives of population and management sampling may result in scouts spending much more time and money sampling than is necessary. For example, Rothrock and Sterling (1982) estimated that by using sequential decision plans, pest management sampling costs could have been reduced by $48,000 to $73,200 per year ($2.64 to $3.96 per hectare) over a six-year study when compared to traditional population-type sample methods in a south Texas IPM program. The result is the ex-
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penditure of time, effort, and money for population-type sampling when only decision sampling is needed. Similarly, unless researchers define their hypotheses sufficiently well and understand the constraints associated with a particular sampling procedure, they may end up with data and results that are too variable and therefore of little use.

Although the objectives of population sampling and commercial monitoring differ, it is important to keep in mind that the development of an accurate, reliable, and easy-to-use commercial monitoring program is dependent on having someone go through the steps of researching the target species, in the process quantifying in great detail their distribution patterns and the factors responsible for the observed patterns. Ecological theories and appropriate commercial monitoring procedures can in turn be developed from the more detailed distributional data.

Population Sampling (Parameter Estimation)

When a researcher is deriving a parameter estimate, a considerable investment in time may be required, while a farmer or crop consultant is often interested in knowing only whether a pest is above or below some economic or action threshold. The greater the reliability necessary for the estimate, the larger must be the sample size. Too often the sample size taken is not related to the number required, and the reliabilities of the estimates are often too low or too high. This equates to poor data at one extreme and unnecessary costs at the other.

Karandinos (1976) presented a series of equations for use in estimating sample sizes. Ruesink (1980) and Wilson and Room (1982) incorporated Taylor’s power law into Karandinos’s equations. Equations (5.1) and (5.2) are generalized equations for estimating sample sizes for population sampling.

\[ n = t_{\alpha/2}^2 D_x^{-2} \bar{x}^{-2} \]  
\[ n = t_{\alpha/2}^2 D_p^{-2} p^{-1} q \]  

where \( t_{\alpha/2} \) = standard normal variate for a two-tailed confidence interval
\( D_x \) = proportion defined as the ratio of half the desired confidence interval to the mean (\( D_x = \text{C.I.}/2\bar{x} \) for enumerative sampling),
\( D_p \) = proportion defined as the ratio of half the desired confidence interval to the proportion of infested sample units (\( D_p = \text{C.I.}/2p \) for binomial or presence-absence sampling)
\( a, b \) = Taylor’s coefficients (Taylor, 1961)

A general feature of these equations is that species whose distribution patterns are more clumped, reflected by higher \( a \) and \( b \) coefficients (see Taylor’s power law), require a greater sample size for a given level of reliability (Fig. 5.3). Also, a smaller sample size is required at higher densities for a given
level of reliability using the enumerative sample size equation (Wilson, 1985). The smaller the value of $D_x$ and $D_p$, the greater is the required sample size. Wilson also indicated that for binomial sampling, unlike enumerative sampling, sample size first decreases then increases as $p$ and corresponding density increase. This is due to a small confidence interval about $p$, corresponding to a very large confidence interval about $X$, at $p$ values approaching unity (Fig. 5.4).

**Commercial Monitoring (Decision Estimation)**

When a field is monitored to determine a pest’s economic status or to determine whether a natural enemy is at a level capable of suppressing a pest’s population density, the farther $X$ or $p$ are from the economic threshold (see Chapter 6), the smaller the sample size required to estimate whether the pest is above or below the threshold. While the number of sample units to estimate a population density with a given level of reliability is independent of an economic threshold, the threshold plays an integral part in determining the required sample size when a management decision is being made. Figure 5.5 illustrates the number of cotton terminals (upper 12 cm of the plant) that would need to be examined comparing population sampling and commercial monitoring for the cotton bollworm. The number of sample units that would
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Figure 5.4 Relationship between $D_x$ and the confidence interval about a mean and $D_p$ and the confidence interval about $p$.

Figure 5.5 Number of cotton terminals that would need to be examined comparing population sampling and commercial monitoring, with the upper 12 cm of the plant as the sample unit.
Table 5.1 Comparison of the Number of Sample Units Required to Estimate the Population Density (Research Sampling) or Economic Status (Decision Sampling) of Cotton Bollworm Based on Egg Sampling ($S^2 = 1.83\bar{x}^{1.13}$, $\alpha = 0.05$, $\beta = 0.05$)

<table>
<thead>
<tr>
<th>$D_S$</th>
<th>$X$</th>
<th>$D_P$</th>
<th>$P$</th>
</tr>
</thead>
<tbody>
<tr>
<td>0.1</td>
<td>704</td>
<td>271</td>
<td>174</td>
</tr>
<tr>
<td>0.2</td>
<td>176</td>
<td>71</td>
<td>47</td>
</tr>
<tr>
<td>0.3</td>
<td>81</td>
<td>33</td>
<td>22</td>
</tr>
<tr>
<td>0.4</td>
<td>47</td>
<td>20</td>
<td>14</td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>$E_T$</th>
<th>$X$</th>
<th>$E_T_P$</th>
<th>$P$</th>
</tr>
</thead>
<tbody>
<tr>
<td>0.5</td>
<td>22</td>
<td>5</td>
<td>4</td>
</tr>
<tr>
<td>2.5</td>
<td>71</td>
<td>7</td>
<td>4</td>
</tr>
<tr>
<td>4.5</td>
<td>2</td>
<td>10</td>
<td>125</td>
</tr>
<tr>
<td>6.5</td>
<td>2</td>
<td>4</td>
<td>16</td>
</tr>
</tbody>
</table>

be required for commercial monitoring increases exponentially as density approaches the economic threshold from either above or below. Unlike research sampling, the number of units that would have to be examined changes as the threshold changes (Table 5.1). The number of sample units requiring examination for commercial monitoring increases as the species density approaches an economic threshold, and changes as the threshold changes.

As with any estimation procedure, there are errors associated with making management decisions. Two types of errors in decision making are (1) the probability of concluding that the population level is above the threshold when it is not, and (2) the probability of concluding that the population level is below the threshold when it is not (Sterling and Pieters, 1979) (Fig. 5.6). Wald (1947) classified these as alpha and beta errors. In theory, an acceptable level of error is determined for alpha by minimizing the total cost involved with monitoring and the costs involved with spraying when not required (labor, machinery, pesticides, resurgence, secondary outbreaks, etc.) and for beta by minimizing the total cost involved with monitoring and the cost of pest damage due to not spraying when justified (Wilson, 1982).

In its simplest case, consider two cotton crops, one irrigated and the other dryland. In the case of a pest such as spider mites or root-knot nematode, the yield loss of both crops is linearly related (within bounds) to pest density above some threshold response level (see Fig. 5.2), with the absolute loss per pest unit being greatest on the irrigated cotton. Because the relationship between alpha and monitoring and pesticide disruption related costs are
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Figure 5.6 α and β error rates for commercial monitoring. α, Probability of initiating a control action when not required; β, probability of not initiating a control action when justified.

about equal for both irrigated and nonirrigated cotton, Fig. 5.7A indicates that both fields would justify equivalent alpha error rates, while the irrigated field would have a much lower beta error rate due to the greater potential for yield loss per pest unit (Fig. 5.7B, C). It also can be shown that as pesticide costs increase or crop value decreases, more intensive sampling or lower alpha and beta errors are justified.

Equations (5.3) and (5.4) (Wilson, 1985), which incorporate the alpha and beta error rates, were derived from the central limit theorem.

\[ n = t_{\alpha}^2 \text{or } \beta | \bar{x} - T |^{-2} a \bar{x}^b \]  
\[ n = t_{\alpha}^2 \text{or } \beta | p - T |^{-2} pq \]  

where T is the economic threshold expressed as a mean density per sample unit for enumerative sampling [Eq. (5.3)], or as a proportion of infested units for presence-absence or binomial sampling [Eq. (5.4)].

Sequential Sampling

Equations (5.3) and (5.4) and the more broadly known equations developed by Wald (1947) fit into the specialized area of sampling referred to as sequential sampling. Many sequential sampling plans have been developed to aid in the management of cotton insect pests (Allen et al., 1972; Rothrock and Sterling, 1982a,b; Sterling, 1976; Sterling and Frisbie, 1981; Sterling and Pieters, 1979; and Wilson et al., 1983a). Sequential sampling plans are avail-
Figure 5.7 Effect of pest damage on the appropriateness of alpha and beta error rates for irrigated and dryland cotton. After Wilson (1982).
Defining the Objectives of Sampling

able for most of the key pests, for both enumerative and binomial sampling procedures, but they are not in extensive use by crop scouts or consultants. Adoption of sequential sampling as an aid in pest management decision making has been shown to reduce sampling costs by 40 to 60% compared with procedures having comparable average error rates. Only when a pest’s population density approaches its economic threshold does the required sample size begin to increase rapidly. During the remainder of a season the required sample size for sequential sampling will be considerably less than that for other quantitative procedures.

From a statistical point of view there is no doubt that the use of sequential sampling can result in considerable saving in sampling costs compared to procedures having comparable average error rates. Unfortunately, the use of one or more existing quantitative sampling procedures as a baseline for evaluating the merits of sequential sampling means very little to a field scout. The problem is not necessarily due to scouts using inherently inferior sampling procedures; the problem is that most scouts make treatment decisions based on sample sizes that are too small. The result is that scouts commonly have unacceptably high error rates in their decision making relative to the value of the fields that they manage. Too often this equates to an excessive, use of less than optimally timed pesticides. This problem is not easily solved. Growers rarely understand the economic consequences of inadequate sampling; and as long as the direct farmer cost of pesticide misuse (spray costs and resistance costs) remains relatively low, and the value of accurate and reliable management decisions is perceived as being relatively low, we can expect farm managers to err on the conservative side.

One criticism of sequential sampling that deserves addressing is its use for assessing the relative abundance or treatment status of more than one insect species. The greater the number of species that are being assessed at any time, the higher is the probability that one or more of these will be sufficiently close to its economic threshold to warrant examining a prede-termined upper limit number of sample units. A general rule of thumb is that sequential sampling will result in a savings in time when using a particular sampling procedure if the number of species or separate age classes being recorded is less than five (Wilson, 1985). Above that number, a scout will probably obtain comparable error rates using a fixed-sample-size approach.

The primary problem with the general lack of adoption of any new sampling plan, including sequential sampling, is that the traditional sampling method must be abandoned or highly modified. Most decision makers, scouts, and consultants are reluctant to abandon methods that have been used for a long time. Sequential sampling procedures have been routinely used by nonagricultural industries since shortly after World War II. As the use of higher technology continues to move into agriculture, the conservatism inherent to agriculture will give way to broader acceptance of procedures such as sequential sampling.
DEFINING THE SAMPLE UNIT

Individuals involved with the use or development of sampling procedures are often concerned with not catching all the organisms present on the sample unit, believing that the best procedure is the one that captures the greatest percentage of the “bugs” in the immediate area examined. Procedures that are most catch-efficient, however, are usually time consuming, often involve the use of expensive equipment, and are often impractical because of the cost involved in their use. A procedure need not have a high catch efficiency if it can be related to an absolute procedure.

Sample Method Efficiency

Few methods provide a truly absolute estimate of density per sample unit, particularly across all the species or age classes sampled. When used to sample predators or pests on cotton, the sweep net has an average efficiency of approximately 10% (Smith et al., 1976; Fleischer et al., 1985; Wilson and Room, 1982). A 10% efficiency value means that a sampler is detecting only an average of 1 in every 10 predators in the area sampled. This percentage is species age and crop stage specific (McGroarty and Croft, 1978; Wilson and Gutierrez, 1980). For some cotton predators the efficiency of the sweep net has been reported to be as low as 3.5% (Wilson and Gutierrez, 1980), while the efficiency approaches 0% if used to sample the eggs of lepidopteran pests (Wilson and Room, 1982). Similarly, root-knot nematode soil extraction techniques capture only 10 to 30% of the juvenile nematodes in the sample (Chapter 6). In comparison, in evaluating four methods of sampling used in perennial crops, Kirk et al. (1972) determined that all methods overestimated weed infestation compared with a complete count method. Unless the scout or researcher realizes the inefficiency of a sampling procedure, a very distorted view develops of the importance of a pest species, its natural enemies, or the growth of the crop.

Factors such as sample unit size, time of day (possibly a temperature or light effect), and stage of crop growth have also been shown to influence the relative efficiency of several sampling procedures (Dumas et al., 1962, 1964; Gertsch and Riechart, 1976; Fillman et al., 1983; Hutchison and Pitre, 1982; Shepard et al., 1974; Wilson and Gutierrez, 1980). Depending on the distribution of the species within the plants and the part of the plant that is sampled, a variable proportion of the individuals will be recovered (Byerly et al., 1978). Figure 5.8 shows a schematic representation of the cumulative vertical distribution of several cotton arthropods. As might be expected, a method that samples only the upper strata of the plant will successfully capture but a very small proportion of those species distributed farther down the plant. Bishop (1981), Fye (1972), Lesar and Unzicker (1978), Nyffler (1982), Wilson and Gutierrez (1980), and others have confirmed that the within-plant distribution patterns of pests and their natural enemies, although
overlapping, are often very distinct and that the catch efficiency of a sampling procedure depends on the part of the plant sampled. Procedures such as using the sweep net to sample vegetation are more efficient at capturing species located in the outer and upper canopy of the crop. Some relatively absolute cotton sampling methods, such as bag samplers, clamshell samplers, Chlorox rinse separators, and for some species visual sampling procedures have a high catch efficiency. They sample nearly the total available habitat (Byerly et al., 1978; Ellington et al., 1984b; Garcia et al., 1982; Leigh et al., 1970, 1984; Wilson and Room, 1982). As an extreme example, the clamshell method or variants thereof, enclose whole plants, and with the exception of highly motile pests or pests that are not easily captured by the subsequent berlese or alcohol extraction technique, the extraction efficiency approaches 100%.

The calibration of relative sampling procedures using absolute sampling has been reviewed by Kogan and Pitre (1980). They suggest correlating numbers collected by relative methods against numbers from absolute techniques. This method also works when comparing two relative sampling procedures. The first step is to treat the most consistently efficient procedure over the range of densities and age class sampled as the independent variable and the second procedure as the dependent variable (Fig. 5.9). The two must be scaled to the same unit or unit area (Morris, 1955). As an example, a 50-sweep sample unit (0.4 m in diameter) covers the area equivalent to approximately 200 plants when the plants are at a density of 10 plants per
square meter. Therefore, for a sweep net sampler and a whole-plant sampler to have the same efficiency, the sweep method has to catch 200 (50 \times 0.4 \times 10) times as many per its sample unit as would the single-plant procedure. After first multiplying the sweep counts by the correction factor 0.005 (= 1/200), if the regression intercept does not differ significantly from zero, possibly implying a constancy in relative efficiencies for the range of densities compared, it may be advisable to force the regression through the origin (Zar, 1974). For some a correlation approach may be a more acceptable alternative to this regression. However, because the regression approach is extremely robust, ignoring the assumption of measurement without error for the independent variable appears to cause little problem.

$$\bar{x}_2 C_{2,1} = b_{2,1} \bar{x}_1$$ (5.5)

where $b_{2,1}$ is the relative efficiency of procedure 2 compared to procedure 1 and $C_{2,1}$ is the scalar for equating the catches to the same unit area.

For a large number of sampling procedures, the foregoing or similar methods can be used to derive relative efficiency estimates (Fleischer et al., 1985;
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Smith et al., 1976; Leigh et al., 1984; Wilson and Room, 1982). The difficulty that some researchers have encountered when attempting to correlate different procedures can largely be attributed to low density estimates, often combined with an inadequate number of sample units and samples. The lower the density, the greater the relative variability about an estimate (see Fig. 5.9)—therefore, the difficulty in correlating counts, and thus the need for a larger number of sample units and samples.

Relative efficiency estimates are necessary when deriving thresholds for use with different sampling procedures. From a commercial perspective the need is obvious. It does little good to talk about action thresholds except within the context of the sampling procedure being used. Until efficiency estimates are available, it is difficult to quantitatively compare and determine which of two or more procedures is best. When possible, relative sampling procedures should be compared with a procedure that captures most of the organisms in the sampled area. Such comparisons allow nearly absolute density estimation, using sampling procedures having low catch efficiencies, but ease of operation and low cost.

Sampler Efficiency

Intuitively, there is one major limitation to comparing the relative "catch efficiency" of two sampling procedures. Whenever skills are required in using a sampling procedure, it is very likely that different samplers will have unequal skills and thus it is unlikely that any two samplers can consistently find equal numbers of insects under similar conditions. Thus it may be as important to calibrate individuals based on some index of skill as it is to calibrate the sampling methods. For example, with sweepnet sampling, a stronger person may be able to force the net through the plant and capture greater numbers of arthropods than can a relatively weaker person. On the other hand, a sampler with good eyesight and instincts may have natural skills in finding arthropods or detecting disease symptoms, skills that cannot be taught to someone with poor eyesight or instinct. Thus it seems likely that the difficulty in calibrating sampling methods may correlate with the skills required for accurate sampling (i.e., visual > sweepnet > knockdown > mechanical vacuum > traps or separation techniques).

Lincoln (1978) is one of the few to evaluate differences between persons when using skill-dependent sampling methods. Scouts showed great differences in their abilities to count either insects or fruit. For example, one scout counted 207% more shed squares than another. Wilson et al. (1983a) found that one sampler recorded only 42% and 65% as many of the leaves infested with mites and predators, respectively, as found by three other samplers; in this case the low efficiency was due to excessive haste by that sampler when examining leaves.

The relative catch efficiency of different samplers is as important as the catch efficiency of different sampling procedures. From a management per-
spective, not knowing the efficiency of a scout will result in misapplication of pesticides. From an ecosystem perspective, this variable efficiency can result in a misinterpretation of the interactions between two or more species. This last point is made apparent by looking at the values derived using a simple diversity index such as the Shannon-Weaver index (see Collier et al., 1973) before and after correcting for catch efficiency (Fig. 5.10). If a sampling procedure were to have zero catch efficiency for one or more species, the bias would be even more extreme. More complicated or more realistic species diversity indices would similarly show a bias in such a comparison.

Relative Cost Reliability

The baseline for evaluating a sampling procedure is how much it costs to obtain an estimate with a given level of reliability. All sampling procedures differ in their catch efficiency (for each species) as well as their ease and cost of operation. The best or most cost-reliable sampling procedure is not necessarily the most catch-efficient. Rummel et al. (1980), as an example, were able to show that the boll weevil pheromone trap index method, although less accurate, was both more reliable and less costly than was the previously used square damage procedure, and correlated best to subsequent weevil damage. The best procedure provides an estimate with a defined level
of reliability at the least cost (Wilson et al., 1982). To determine and compare the cost reliability of two or more sampling procedures requires

1. Relative or absolute efficiency estimates
2. Variance–mean relationship (enumerative) or proportion infested–mean relationship (presence–absence)
3. Cost (time) required to collect and examine each sample unit.
4. Cost (time) required to move between sample sites.

Costs required to obtain an estimate or make a decision with a given level of reliability can be compared to determine which of two sampling procedures is better (Wilson et al., 1982).

\[
\frac{C_1}{C_2} = \frac{n_1(\theta_1 + \phi_1)}{n_2(\theta_2 + \phi_2)}
\]

(5.6)

where \( C_i \) = cost for a given level of reliability for the ith sampling procedure
\( \theta_i \) = time (cost) required to examine an individual sample unit using the ith sampling procedure
\( \phi_i \) = time (cost) required to move from sample unit to sample unit for the ith procedure; assumes that the time taken to walk into a field to the first sample unit is about equal to the time taken to walk between units
\( n_i \) = number of samples required for an estimate with a given level of reliability; for purposes of population sampling, Eqs. (5.1) (enumerative) and (5.2) (presence–absence) are appropriate for estimating the \( n \) values; while Eqs. (5.3) and (5.4) are correspondingly used for commercial monitoring.

When the ratio of \( C_1/C_2 \) is greater than unity procedure 1 costs more for a given reliability level than does procedure 2. When two enumerative procedures are compared, Eq. (5.6) requires Taylor's coefficients which will differ for each species and for each sampling procedure. It is possible to compare an enumerative and a presence–absence procedure (the latter not using Taylor's power law). When such a comparison is made, it is necessary to use \( D \) values that give comparable confidence intervals for Eqs. (5.1) and (5.2). The \( D \) value used for a mean is necessarily different from the corresponding \( D_p \) value about \( p \) (see Fig. 5.4). The problem does not arise when comparing the sample-size equations used in commercial monitoring since the somewhat arbitrary aspect of choosing \( D \) is replaced by the difference between the economic threshold, \( T \), and the estimated density, \( \bar{x} \), or proportion of infested sample units, \( p \).

For some species the \( C_1/C_2 \) ratios may change with respect to one or more variables, such as plant density, as illustrated in Fig. 5.11 comparing a terminal subsampling plan \((N, C)\) with a whole-plant sampling plan \((N, C)\).
when sampling for cabbage loopers. It may also change due to sampling costs changing with pest density, or the relative efficiency of a sampling procedure, or the efficiency of a sampler changing through time (Wilson and Gutierrez, 1980). In enumerative sampling, samples with high densities of target organisms may be extremely costly to count. One sampling method may be better at lower densities and another better at higher ones. Density also has the effect of changing variance, which directly affects the number of samples required for a given level of reliability. When the cost reliability of two or more sampling procedures is compared for more than one species, one can determine which procedure is the best by taking a weighted average of the $C_1/C_2$ ratios [see Eq. (5.6)]. If this average is greater than unity, method 2 is best, and vice versa. For pest management purposes it may be better to use the sampling procedure that is most cost reliable for the key pest, which may be the best or at least an adequate procedure for the majority of other species being sampled. If information is required for several species, some of which cannot be cost-reliably sampled with the same procedure, more than one sampling procedure should be used. Whether to use one or more sampling procedures, and which ones, should be determined in light of their costs and practicality.

Figure 5.11 Effect of pest density on the relative cost reliability of two sampling procedures. After Wilson et al. (1982).
SAMPLING FREQUENCY AND FORECASTING

Sampling frequency is usually determined subjectively and often has little bearing on the phenology of the crop, pests, or natural enemies. In its simplest form, phenology models that use pheromone traps to establish a biofix can be used to determine when subsequent samples might be taken. Tummala and Haynes (1977) predict that as use and dependence upon such models increases, there will be a concomitant decrease in costs associated with monitoring. The degree to which monitoring can be reduced, however, depends on the reliability of these forecasting techniques and upon the current level of monitoring. The current degree of monitoring may be far less than required based on the relative costs of sampling and the cost of making inappropriate management decisions. The number of unknowns or “black boxes” in our understanding of pest management systems promoted calendar-type sampling in the same manner as it promoted a calendar or prophylactic spray strategy.

Sampling frequency can be estimated quantitatively in several ways. From an optimization perspective, however, the following should be considered.

1. The phenology of the pest species
2. The rate at which a pest’s population is increasing (affected by abiotic and biotic factors) and the proximity of pest densities to economic or action thresholds
3. The damage potential and cost of controlling the pest species at different crop stages [relates to the economics of incremental costs and incremental benefits of control actions (Headley, 1982)]

To date, the greatest amount of work on sampling frequency in cotton has focused on spider mites (Plant and Wilson, 1985; Wilson, 1985; Wilson et al., 1985). Figure 5.12 illustrates results from a simple forecasting model (Wilson, 1985) which meet the three requirements listed above. This procedure is appropriate for univoltine or multivoltine species which have extremely short generation times. The timing of samples is based on a curvilinear projection based on the rate of pest infestation estimated from previous samples, where the projected time interval between successive samples decreases as the estimated population density approaches the economic threshold. This phenomenon is very closely analogous to sequential sampling where the closer the population is to the economic threshold, the greater the number of samples required to estimate with a given level of reliability whether the population is above or below the threshold. The projected time interval between samples also decreases more variable the data, due either to lower-quality data (fewer sample units or greater sampler error) or intrinsically more variable data (a species attribute).
Although very little research has been conducted on optimizing sampling frequency, research efforts in cotton and soybeans using novel approaches ranging from Bayesian analysis, modifications to conventional sequential analyses, to fairly simple but robust regression techniques (Pedigo and van Schaik, 1984; Plant and Wilson, 1985a; Wilson, 1985; Wilson et al., 1985) bode well for further improvements in the development and use of forecasting techniques in crop-pest management.

**COMMUNITY-WIDE MANAGEMENT**

An area that deserves special notice is community-wide management (see Chapter 13). The notion that populations must be managed on a community basis versus a field-by-field approach is a major departure from most current sampling programs, and influences the sampling procedures employed.
Highly vagile species may require a community-wide or even multinational monitoring approach to make management decisions. Less vagile species can be sampled and managed on a field-by-field basis or even a smaller-area basis. Sampling of arthropods, weeds, nematodes, and pathogens of cotton can be viewed as a continuum, with some being sampled on a community, state, national, or multinational basis, while for others spot sampling within a particular field may optimize costs and benefits.

In cotton, multinational sampling may be required for *Heliothis zea* (Boddie) in order to predict its seasonal dynamics since this insect may migrate long distances (Hartstack et al., 1976, 1982). Fusarium wilt, in comparison, would more likely require a valley-wide community approach to monitor and control its rate of spread. The community-wide bollworm management program in Arkansas developed by Phillips and his co-workers has demonstrated that a considerable reduction in pesticide usage can be achieved with this approach (see Chapters 10 and 13).

**DISPERSION PATTERNS**

This chapter would be incomplete were we not to cover some of the basic principles of dispersion and distribution. Spatial patterns characterize species and are determined by their interactions with the biotic and abiotic environment. For most sample unit sizes used in estimating parameters or in making management decisions, the observed spatial pattern is clumped (Taylor et al., 1978; Wilson and Room, 1983). Very few species have uniform spatial patterns. The reported inability to distinguish spatial patterns from random is often due to low density. At low densities, the *variance/mean* ratio approaches 1.0; while as density increases, the ratio becomes increasingly greater than 1.0. This lack of statistical sensitivity is also commonly aggravated by an inadequate sample size. The smaller the sample size, the greater the variability about a species variance–mean curve and accordingly, the more difficult it is to distinguish the pattern from randomness, particularly at lower densities.

A species' spatial pattern can also be affected by sample unit size (Wilson, 1985). The perceived pattern can differ from the actual pattern due to the sample unit size being either larger or smaller than what is biologically relevant for the species being sampled. A sample unit size of 50 sweeps of a net, for example, has no bearing on the underlying biologically based pattern of the species being sampled. The size chosen as the sample unit justifiably is one of convenience, and although often based on the biology of the target organisms, is nevertheless chosen more to aid in assessing populations of the organism than for understanding the mechanisms involved with determining the underlying pattern.

A common dispersion statistic is the ratio of the standard deviation over the mean (sIX), referred to as the coefficient of variability (CV). The CV is
an appropriate statistic for comparing relative deviations in that unlike the variance/mean ratio, its value is unitless. CV values can provide considerable insight into the interaction of a species with its environment. In general, as sample unit size increases, the CV decreases. Using a progressively smaller sample unit size can also result in a decreasing CV. Figure 5.13 demonstrates this point for two hypothetical species, indicating that the "biologically relevant sample unit size" (Wilson, 1985), that is, that sample unit size which captures the essence of a species' spatial pattern, estimated by the maximum CV value, may differ from one species to the next. The figure also demonstrates that the underlying heterogeneity of the environment can also produce a second local maximum CV. This might reflect a preference for a particular soil gradient or stage of host crop development, again providing useful information about the interaction of a species with its abiotic and biotic environment.

Distribution Functions

Probability distribution functions have also been used to describe the spatial pattern of a range of species in cotton (Wilson et al., 1983b). Many of these distribution functions are frequently constrained by unrealistic assumptions, such as discussed with the negative binomial distribution function (Taylor et al., 1978). Many of these constraints, however, are of little or no importance when using distribution functions as sampling or monitoring tools. Arthropods found in cotton fields generally exhibit a clumped pattern of
dispersion (Pieters and Sterling, 1973; Reiley and Sterling, 1983; Wilson and Room, 1983), which is often characterized by fit to the negative binomial distribution (Wilson et al., 1983b). Weeds, nematodes, and pathogens similarly are often found aggregated in “hot spots” within a field. These spots represent colonization sites from which further spread may occur. Such areas may also be particularly favorable for population buildup, such as with root-knot nematode in a sandy soil streak within a field comprised largely of a clay loam soil. The Poisson distribution function may sometimes provide a reasonable fit for certain arthropods under some conditions (Kuehl and Fye, 1972; Wilson and Room, 1983), such as when using a 50-sweep sample unit, which masks the underlying distribution pattern, for reasons discussed previously. For most species using most sampling procedures, however, their pattern within their environment differs considerably from random, with the Poisson distribution pattern consequently being an inappropriate description.

Taylor’s Power Law

Fracker and Brischle (1944), Hayman and Lowe (1961), and Taylor (1961) each independently developed an equation that relates in a dynamic fashion the commonly used and simple variance and mean statistics. This equation replaces the complex probability distribution functions used in describing distribution pattern of a species. Each of these persons found that the variance changes with the mean in a nonlinear but easily predictable manner, described by

\[ S^2 = ax^b \]  

(5.7)

where \( a \) and \( b \) are species and sample unit specific coefficients which together describe a species’ spatial pattern.

Taylor’s power law, as this equation has become known, although basically an empirical curve fit to an exponential function, has proven extremely useful as a component in developing sampling procedures (Ruesink, 1980; Wilson, 1985). Taylor’s \( a \) and \( b \) coefficients are estimated using log-log, transformations or nonlinear regression techniques (Miller, 1971; Taylor, 1961). Both estimation procedures are subject to some error (Miller, 1971; Wilson, 1985) and preliminary analyses should be attempted using different techniques before deciding which is best for a particular data set.

Both \( a \) and \( b \) are near constant for a species category over a fairly wide range of conditions. Taylor et al. (1978) consider the coefficient \( b \) constant for a species, with only \( a \) affected by sample unit size. Banerjee (1976) and others (see Wilson, 1985) show that both \( a \) and \( b \) are subject to change due to age-specific dispersal, mortality, and sample unit size. From Eq. (5.7) it can be demonstrated that a high \( a \) value would not necessarily imply a clumped spatial pattern, since a low \( b \) value could result in a uniform dis-
Figure 5.14 Taylor’s a and b coefficients interacting to effect a species distribution pattern.

Distribution pattern for a wide range of densities. Similarly, a high b value need not imply a clumped pattern at all densities since its effect on variance can be counteracted by a low a value (Fig. 5.14).

In deriving Taylor’s coefficients for approximately 30 categories of arthropods and plant parts for three sampling procedures in cotton, Wilson and Room (1983) found that the value of a ranged from 0.80 to 6.37, while the value of b ranged from 0.93 to 1.40. Wilson et al., (1983a) using a single leaf sample unit, estimated an a value of 6.16 and a b value of 1.54 prior to population crash, for the active stages of Tetranychus spp., a highly clumped organism. Since relatively few species in cotton are more clumped than are spider mites, these values may be near the upper limit for mobile stages of organisms in this crop (Table 5.2). As has been discussed in the sections “Population Sampling” and “Commercial Monitoring,” the magnitude of Taylor’s a and b coefficients reflects the degree of clumping of the particular species and has a profound effect on the number of sample units that must be examined to obtain an estimate with a given level of reliability (see Fig. 5.3).

Proportion Infested–Mean Relationship

The preceding section shows how the $S^2 - \bar{x}$ relationship dynamically categorizes a species’ spatial pattern. The relationship between the proportion of sample units that are infested with a species and the density per sample unit can be equally useful (Ingram and Green, 1972; Sterling, 1975, 1978;
Sterling and Pieters, 1979; Wilson 1982, 1985; Wilson et al., 1983). For any species, as its density increases, the proportion of infested sample units increases (Fig. 5.15). The rate at which the proportion of infested sample units $p$ increases with density is characteristic for a particular species.

When a species is randomly distributed, $p$ is estimated by

$$p = 1 - e^{-x}$$  \hspace{1cm} (5.8)$$

where $p$ is the proportion of sample units with one or more of the particular species or category being recorded.

When a species with a clumped pattern is compared with one that is randomly distributed but at equal densities, $p$ will be less for the species having the clumped distribution pattern. The reverse occurs comparing a uniformly and a randomly distributed species.

Since Eq. (5.8) is not a suitable description for most species, both Ingram and Green (1972) and Sterling (1975, 1976, 1978) used polynomial regressions to describe the relationship between the proportion of infested sample units and density for several pests of cotton. Wilson and Room (1983) expanded the use of the proportion infested–mean relationship by developing a mathematically and biologically more tractable binomial model which can incorporate a variance–mean relationship in the form of Taylor's power law:

$$p = 1 - e^{-\frac{s^2}{k}}(S^2(k-1)-1)^{-1}$$  \hspace{1cm} (5.9)$$

$$p = 1 - e^{-\frac{s^2}{k}}(a^b-1)(a^b-1)^{-1}$$  \hspace{1cm} (5.10)$$
### Table 5.2 Taylor's Coefficients for Cotton Plant Parts and Arthropods

<table>
<thead>
<tr>
<th>Category</th>
<th>Bag</th>
<th>a</th>
<th>b</th>
<th>( r^2 )</th>
<th>( n^a )</th>
</tr>
</thead>
<tbody>
<tr>
<td>Cotton fruit</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Squares</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Bolls</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Open bolls</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lepidoptera: Noctuidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Heliothis armigera</em> (Hübner)</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>H. punctigera</em> Wallengren</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>White eggs</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Brown eggs</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Total eggs</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Very small larvae (&lt;3 mm)</td>
<td>1.43</td>
<td>1.12</td>
<td>0.90</td>
<td>29</td>
<td></td>
</tr>
<tr>
<td>Small larvae (3–7 mm)</td>
<td>1.05</td>
<td>1.01</td>
<td>0.95</td>
<td>30</td>
<td></td>
</tr>
<tr>
<td>Medium-sized larvae (7–19 mm)</td>
<td>0.84</td>
<td>0.95</td>
<td>0.98</td>
<td>22</td>
<td></td>
</tr>
<tr>
<td>Large larvae (&gt;19 mm)</td>
<td>1.00</td>
<td>0.98</td>
<td>0.92</td>
<td>11</td>
<td></td>
</tr>
<tr>
<td><em>Earias huegeli</em> Rogenhofer larvae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Hemiptera: Cicadellidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Austroasca viridigrisea</em> (Paoli)</td>
<td>1.61</td>
<td>1.17</td>
<td>0.97</td>
<td>42</td>
<td></td>
</tr>
<tr>
<td>Hemiptera: Lygaeidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Oxycarenus luctuosis</em> M.&amp;S. adults</td>
<td>4.62</td>
<td>1.43</td>
<td>0.95</td>
<td>37</td>
<td></td>
</tr>
<tr>
<td><em>Geocoris lubra</em> (Kirkaldy) adults</td>
<td>1.60</td>
<td>1.12</td>
<td>0.96</td>
<td>13</td>
<td></td>
</tr>
<tr>
<td>Hemiptera: Miridae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Campylomma livida</em> Reuter adults</td>
<td>1.52</td>
<td>1.11</td>
<td>0.93</td>
<td>12</td>
<td></td>
</tr>
<tr>
<td><em>C. livida</em> nymphs</td>
<td>0.89</td>
<td>0.97</td>
<td>1.00</td>
<td>11</td>
<td></td>
</tr>
<tr>
<td>Coleoptera: Coccinellidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Verania frenata</em> Er. adults</td>
<td>1.87</td>
<td>1.18</td>
<td>0.93</td>
<td>7</td>
<td></td>
</tr>
<tr>
<td><em>Coccinella repanda</em> Er. adults</td>
<td>1.14</td>
<td>1.01</td>
<td>0.81</td>
<td>23</td>
<td></td>
</tr>
<tr>
<td><em>Diomus notescens</em> Blackburn adults</td>
<td>0.86</td>
<td>0.96</td>
<td>1.00</td>
<td>17</td>
<td></td>
</tr>
<tr>
<td>Coleoptera: Melyridae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Laius bellulus</em> (Guerin) adults</td>
<td>0.99</td>
<td>1.00</td>
<td>0.98</td>
<td>20</td>
<td></td>
</tr>
<tr>
<td>Araneida: Oxyopidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Oxyopes</em> spp. adults</td>
<td>0.93</td>
<td>0.98</td>
<td>1.00</td>
<td>8</td>
<td></td>
</tr>
<tr>
<td>Araneida: Salticidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Salticidae</em> spp. adults</td>
<td>0.86</td>
<td>0.93</td>
<td>0.84</td>
<td>11</td>
<td></td>
</tr>
<tr>
<td>Araneida: Clubionidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Chiracanthium diversum</em> (Koch) adults</td>
<td>3.30</td>
<td>1.39</td>
<td>0.87</td>
<td>25</td>
<td></td>
</tr>
<tr>
<td><em>C. diversum</em> immatures</td>
<td>0.87</td>
<td>0.97</td>
<td>1.00</td>
<td>12</td>
<td></td>
</tr>
<tr>
<td>Araneida: Theridiidae</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Arachaearanea veruculata</em> (Urquhart) adults</td>
<td>0.80</td>
<td>0.94</td>
<td>0.99</td>
<td>19</td>
<td></td>
</tr>
<tr>
<td><em>A. veruculata</em> immatures</td>
<td>0.88</td>
<td>0.97</td>
<td>1.00</td>
<td>14</td>
<td></td>
</tr>
</tbody>
</table>


*Regression for categories with at least 7 days’ data.*
Table 5.2 Taylor's Coefficients for Cotton Plant Parts and Arthropods (continued)

<table>
<thead>
<tr>
<th>Sampling Method</th>
<th>Visual</th>
<th>Sweep</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>$a$</td>
<td>$b$</td>
</tr>
<tr>
<td>Visual</td>
<td>2.79</td>
<td>1.19</td>
</tr>
<tr>
<td></td>
<td>2.54</td>
<td>1.22</td>
</tr>
<tr>
<td></td>
<td>2.50</td>
<td>1.18</td>
</tr>
<tr>
<td></td>
<td>1.22</td>
<td>1.05</td>
</tr>
<tr>
<td></td>
<td>1.17</td>
<td>1.04</td>
</tr>
<tr>
<td></td>
<td>1.16</td>
<td>1.03</td>
</tr>
<tr>
<td></td>
<td>1.14</td>
<td>1.03</td>
</tr>
<tr>
<td></td>
<td>1.30</td>
<td>1.06</td>
</tr>
<tr>
<td></td>
<td>1.96</td>
<td>1.21</td>
</tr>
<tr>
<td></td>
<td>6.37</td>
<td>1.40</td>
</tr>
<tr>
<td></td>
<td>1.21</td>
<td>1.04</td>
</tr>
<tr>
<td></td>
<td>1.61</td>
<td>1.11</td>
</tr>
<tr>
<td></td>
<td>1.33</td>
<td>1.06</td>
</tr>
<tr>
<td></td>
<td>1.15</td>
<td>1.03</td>
</tr>
<tr>
<td></td>
<td>1.14</td>
<td>1.04</td>
</tr>
<tr>
<td></td>
<td>1.49</td>
<td>1.09</td>
</tr>
<tr>
<td></td>
<td>1.33</td>
<td>1.07</td>
</tr>
<tr>
<td></td>
<td>1.29</td>
<td>1.06</td>
</tr>
<tr>
<td></td>
<td>1.44</td>
<td>1.08</td>
</tr>
<tr>
<td></td>
<td>1.14</td>
<td>1.03</td>
</tr>
<tr>
<td></td>
<td>2.05</td>
<td>1.18</td>
</tr>
<tr>
<td></td>
<td>1.03</td>
<td>1.01</td>
</tr>
<tr>
<td></td>
<td>1.65</td>
<td>1.13</td>
</tr>
</tbody>
</table>
If baseline data are available relating \( p \) to the density of a species, a quick estimate of density can be obtained by recording for each sample unit whether they are infested (Fig. 5.16). Wilson et al. (1981) found that it took as much as 2 hours to record the number of mites on a single heavily infested leaf, while presence—absence sampling takes about 1 minute per leaf, including the time required to walk the field.

Equations (5.9) and (5.10) can be used to describe the \( p-X \) relationship for all three major types of distribution patterns. As the variance approaches the mean as a limit, both equations collapse to the nonzero term of the Poisson distribution function (Wilson et al., 1983a). Taylor's \( a \) and \( b \) coefficients (see Table 5.2) enables estimates of \( p \) for any density. It should be kept in mind that Taylor's \( a \) and \( b \) coefficients are affected by sample unit size and would have to be derived for different sample unit sizes.

In case of an organism whose spatial pattern does not differ significantly from randomness, \( S^2/\bar{x} = 1 \), assuming in this case that this ratio is a true representation of \( \sigma^2/\mu \). Similarly, if \( S^2/\bar{x} \) is greater than unity, the spatial pattern is clumped; if \( S^2/\bar{x} < 1.0 \), the spatial pattern is uniform. In comparison, the relationship between \( p \) and \( \bar{x} \) can also be used to categorize a species spatial pattern. For the Poisson distribution, we know that \( p = 1 - e^{-x} \). Rearranging, we get

\[
1 - p = e^{-x} \quad \text{or} \quad \log_e(1 - p) = -\bar{x}
\]

and finally,

\[
-\log_e(1 - p)/\bar{x} = 1.0
\]
Table 5.3 Classification of Spatial Patterns for Cotton-Inhabiting Organisms

<table>
<thead>
<tr>
<th>Spatial Pattern</th>
<th>Variance/Mean Ratio</th>
<th>$-\log_e(1 - p)/\bar{x}$</th>
</tr>
</thead>
<tbody>
<tr>
<td>Aggregated or clumped</td>
<td>&gt;1.0</td>
<td>&lt;1.0</td>
</tr>
<tr>
<td>Random</td>
<td>= 1.0</td>
<td>= 1.0</td>
</tr>
<tr>
<td>Uniform</td>
<td>&lt;1.0</td>
<td>&gt;1.0</td>
</tr>
</tbody>
</table>

As with the $S^2/\bar{x}$ ratio, it can be shown that when the variance is greater than the mean as occurs with an organism with a clumped distribution pattern, $-\log_e(1 - p)/\bar{x} < 1$.

The similarity between the classification of a distribution pattern using the variance/mean ratio, or the $-\log (1 - p)/\bar{x}$ ratio, is not fortuitous, and both lend themselves to fairly rapid evaluation of a species spatial pattern (Table 5.3).

**FUTURE DIRECTIONS**

As the concept of integrated pest management evolves and as higher-technology decision aids such as pest and crop models become increasingly available, there will probably be much greater flexibility with regard to the choice of strategies, tactics, and timing of actions taken. In the past it has been thought necessary to reduce the complexity of the decision-making criteria to simple and easy-to-remember rules such as: spray for bollworms on July 15 or spray for boll weevils at the appearance of the first matchhead square. The next step in the evolution toward IPM was to add sampling and economic thresholds to the statements above, or to use sampling procedures which, although having low catch efficiencies, can be related to an absolute method, are accurate, and are easy to use with minimal training. The pheromone-trap index system developed by Rummel et al. (1980) to sample boll weevils, the sequential sampling plans developed by Allen et al. (1972) and Sterling (1975) and his colleagues, or the presence–absence or binomial sampling and forecasting procedures developed by Plant and Wilson (1985a) and Wilson et al. (1983a) for sampling spider mites are good examples of such procedures.

Decision rules of the future may well incorporate some of the older and simpler rules, but with the use of computers we can afford to integrate the concept of a continuum while maintaining flexibility in the decision process. The concept of a continuum recognizes that conditions are not static; they are not the same from one year to the next. Many variables, such as soil moisture, fertility, natural enemies, economics, physiological time, weather, and others, will affect the numbers of pests that can be tolerated at any period (Pedigo et al., 1986; Sterling, 1984; Wilson, 1986). Thus, under dryland conditions, for example, fewer cotton fleahoppers can be tolerated on
plants that have begun to set squares if soil moisture is in short supply than if soil moisture is plentiful. Computers can handle all these variables concurrently and provide recommendations that have been optimized for all relevant conditions. Similarly, the use of expert systems rule bases and the entire area of artificial intelligence will greatly increase the robustness and realism of the decision-making process while maintaining an outer veneer of user friendliness (Jones, 1985; Plant and Wilson, 1985; Stone et al., 1986).

What will be the need of field sampling in the future of IPM? At minimum, sampling will still be required to validate insect model predictions. Complete reliance on models, or for that matter, reliance on any unilateral management approach, is never wise. As the management of cotton systems continues to evolve, simple, reliable, and easy-to-use crop and pest monitoring procedures will probably remain as one of the major foundation blocks for an ecologically sound and economically feasible pest management program.

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GENETICALLY MODIFIED COTTONS IN IPM Bt cotton varieties expressing the Cry 1Ac protein from Bacillus thuringiensis subsp. kurstaki were first registered in Australia in 1996 (INGARD®) and gradually increased in area under an industry agreed deployment strategy which limited use to 30% of the cotton area. Two gene (Cry IAc/Cry 2Ab) varieties (Bollgard II) have been commercialised from 2004/05 and have now completely replaced Ingard varieties. All Bt varieties are grown under a comprehensive management strategy designed to minimise the risk of resistance evolving in Helicoverpa armigera, the main