



Beyond Nomenclature and Use: A Review of Ecological Methods for Ethnobotanists

Charles M. Peters

*Institute of Economic Botany,
The New York Botanical Garden*

Introduction

Quantitative Assessment of Species Density

Size and Shape of Sample Plots

Number of Sample Plots

Arrangement of Sample Plots

Field Procedures and Measurements

Which Plants to Count?

What to Measure?

Growth and Yield Studies

Selection of Sample Trees

Methodology and Data Collection

Reproductive Propagules

Plant Exudates

Vegetative Tissues

Selected Guidelines for Ethnobotanical Research: A Field Manual, 241–276

Edited by Miguel N. Alexiades

© 1996 The New York Botanical Garden

Defining the Resource Base

Conclusions

Acknowledgments

Literature Cited

Introduction

Although broadly defined as the study of the interrelationships between plants and people, ethnobotany has, in most cases, focused solely on compiling lists of the plant species used by different cultural groups. Recent studies have expanded on this concept somewhat by trying to quantify the relative importance of different plant uses (see Phillips, Chapter 9, this volume) or by focusing in greater detail on the actual pattern or intensity of use of different resources (see Zent, Chapter 10, this volume). These modifications notwithstanding, ethnobotany has remained primarily a static, descriptive endeavor. The core components of the discipline today—plant collection, plant identification, and the detailed documentation of plant uses at one point in time—are essentially the same as they were 100 years ago when Harshberger first coined the term *ethnobotany* (Harshberger, 1896).

The basic shortcoming of a purely descriptive approach is that it does not take into account the fact that things happen when people use plants. Destructive harvesting and overexploitation, for example, can gradually eliminate a plant species from the local environment. Deliberate planting, controlled harvesting, and forest management, on the other hand, can greatly increase the distribution and abundance of local resources. Species lists alone are insufficient to document these dynamic interactions. A particular plant resource may be recorded as having exceptional properties and a high use value during one ethnobotanical survey, but if the species that produces it occurs at low densities in the forest, is harvested destructively, or cannot regenerate under existing levels of exploitation, there is a very high probability that the resource will not even be noted in subsequent surveys. There is an ecological context within which people interact with plants, and the exploration of this territory can generate a host of new questions for inquisitive ethnobotanists. Perhaps it is time to go beyond the basic queries of What is the name of this plant? and

What is it used for? and to ask How do indigenous communities apply their knowledge of the local flora? and, perhaps most important, What are the long-term impacts of these actions?

The purposes of this review are to outline some of the basic ecological methods available for addressing the latter types of questions and to briefly introduce the reader to the literature on quantitative ecology and vegetation sampling. Particular attention is focused on the collection of quantitative density and yield data for different plant resources. The relative advantages and limitations of a variety of sampling procedures are discussed, and potential methodological problems are highlighted whenever appropriate. Most of the examples presented are taken from my own research in the tropical forests of Amazonia and Southeast Asia. This review is not exhaustive, nor is it a "cookbook" of ecological methods that can be applied without modification. The selection of an appropriate sampling scheme for ethnobotanical work will ultimately depend on the specific objectives of the research, the experience and judgment of the investigator, and the time, financial resources, and personnel available.

Quantitative Assessment of Species Density

Density, or the number of individuals per unit area, is probably the ecological parameter of greatest interest to the ethnobotanist. This basic statistic can tell the investigator how much of a given plant resource is available for exploitation and where the greatest abundance of harvestable material is located. If the individuals are measured as well as counted, size-specific density estimates can be obtained to assess whether the species is regenerating under exploitation. Quantitative estimates of species density also lay the foundation for ecological monitoring by providing a yardstick with which to measure the long-term sustainability of plant resource exploitation (Hall & Bawa, 1993; Peters, 1994).

For cultivated plants growing in house gardens or small agroforestry plots, it is sometimes possible to conduct a 100% inventory of all individuals to obtain a precise estimate of species density (e.g., Padoch & de Jong 1991; Rico-Gray et al., 1990). In

most situations, however, it is neither feasible nor warranted to count all of the individuals of a species, and some type of sampling methodology will be required. The major issues to be considered in selecting an appropriate sampling scheme for collecting density data are related to the size, shape, number, and arrangement of sample plots. Also important, of course, are the procedures and measurements used in the field, deciding which plants to count, and what variables to measure.

Size and Shape of Sample Plots

Beyond the general advice that larger plants require larger sample plots, there are few rules that govern the selection of an appropriate plot size for vegetation sampling. Plots of 1.0 m² are usually sufficient for use with herbaceous plants (Kershaw & Looney, 1985; Van Dyne et al., 1963), but shrubs and understory vegetation may require plots of from 16.0 to 100 m² (Lyon 1968; Myers & Chapman, 1953). A variety of plot sizes have been used to sample forest vegetation. For inventory work in tropical forests, Lang et al. (1971) and Knight (1975) recommended the use of 10×20 m plots; 1000-m² plots were used in the comparative studies of Holdridge et al. (1971) and Gentry (1982); and various investigators have used large, single plots of from 1.0 to 3.0 hectares (e.g., Anderson et al., 1985; Campbell et al., 1986; Gentry, 1990; Hubbell, 1979). Although 100-m² plots are frequently recommended for measuring the density of tree species in temperate hardwood forests (Mueller-Dombois & Ellenberg, 1974), some studies (e.g., Bormann, 1953; Whittaker, 1967) found that 1000-m² or 1400-m² plots gave better results in these plant communities.

Logistic factors are also important to consider in selecting a plot size. Large sample plots, which have a greater probability of encountering different patches or "clumps" of individuals than do small plots, will usually provide a better estimate of the mean density of a species (Gauch, 1982). Large plots, however, take longer to lay out and inventory, and, given that there are more individuals to measure and count, there are more chances to make mistakes. Small plots are immeasurably easier to lay out and count, but they frequently produce a density estimate with a large error term, especially if the plot size selected is smaller than

the average size of the natural aggregations formed by the species being surveyed (Greig-Smith, 1983). If most of the sample plots fall directly within a clump, the final density estimate will be too high. If most of the plots miss these clumps, or only partially bisect them, the density of the species will be underestimated. As can be appreciated, the selection of an appropriate plot size for density sampling represents a compromise between time, expense, and level of precision required.

In terms of overall sampling efficiency, the actual shape of the plot may be more important than its size. As is shown in Figure 1, plots of similar size can exhibit notable differences in the amount of perimeter, or "edge," depending on their shape. For a given sample area, circular plots have less edge than square plots, and square plots have less edge than rectangular plots or transects. The total perimeter of a rectangular plot is controlled by the ratio of plot width to plot length, and long, narrow transects have considerably more perimeter than short, wide ones. To illustrate the magnitude of this range, I used different plot configurations to calculate the maximum and minimum perimeter lengths for each transect area shown in Figure 1.

There are positive and negative aspects associated with the perimeter characteristics of a sample plot. On the positive side, a larger amount of edge means that the sample unit will usually bisect a greater number of different habitats and species patches and provide a more representative description of the study area. This benefit is enhanced by orienting the long axis of the transect at right angles to topographic or drainage features (Avery & Burkhart, 1983). The extensive use of transects in floristic surveys (see reviews in Campbell et al., 1986; Gentry, 1982) and forest inventory operations (FAO, 1973; Wood, 1989) is largely the result of the increased perimeter afforded by sample units with this shape.

On the negative side, plots that have a large perimeter or boundary also have a large number of boundary or border trees whose inclusion or exclusion from the sample must always be assessed. The treatment of border trees is a chronic source of error in plot sampling. Ideally, the investigator should carefully measure out from the centerline of the plot to every border tree to verify that it is actually "in." This, however, can be a very time-consuming process, and, in most cases, the distance is sim-

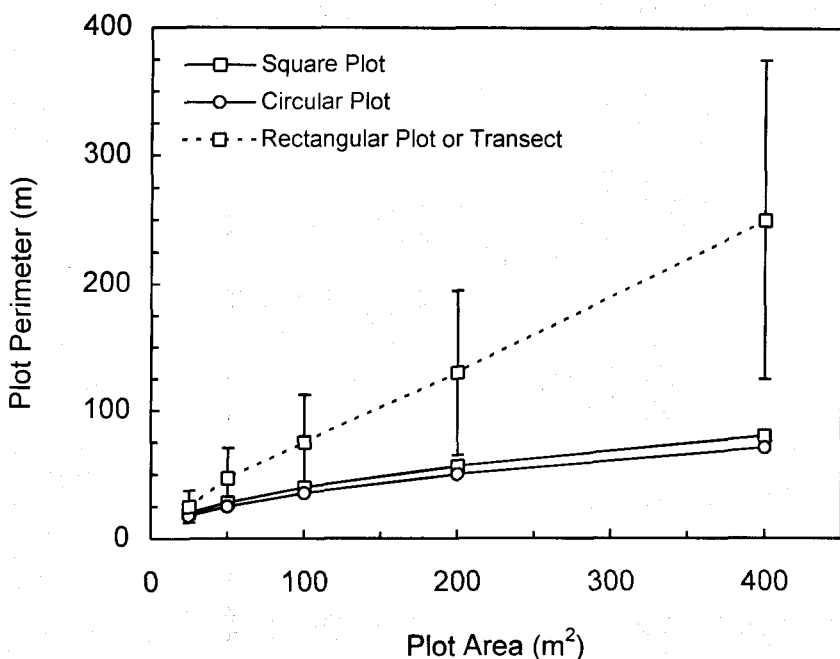


Figure 1. Relationship between plot boundary (in meters) and plot area (in square meters) for sample units of different sizes and configurations. Mean perimeter length is plotted for rectangular plots; minimum and maximum lengths for each rectangular plot area are indicated by vertical bars.

ply estimated visually by the investigator. The net result of this procedural shortcut is that too many stems are usually counted near the perimeter of a plot. Other sources of subjective bias can also enter in the treatment of border trees. For example, if the person responsible for determining whether a border tree is in or out is also the one who must climb the tree to collect the herbarium specimen, there is a tendency late in the day for large canopy trees to be out and for pole-sized, easily collectible stems to be in. Similarly, there is always the subtle temptation in floristic surveys to include the border trees that represent new species and to exclude those of species that have already been tallied.

Small sampling errors due to edge effects and the careless assessment of border trees can quickly add up. Consider for the moment a 10×1000 m transect run through a lowland tropical forest. If we assume that the questionable border area of the transect is 50 cm wide, there is approximately 1000 m^2 of perimeter,

or about 10% of the total plot area, within which a decision must be made as to which trees are in or out. Further assuming that the average density of trees in the forest is 700 stems/ha and that these stems are evenly distributed throughout the site, the investigator conducting the inventory should encounter about 70 border trees, or, stated another way, be confronted with at least 70 chances for committing a sampling error. To fully reap the statistical benefits of using rectangular plots and transects, special care should be taken to ensure that all plot boundaries are precisely maintained.

Number of Sample Plots

The total number of plots to use for collecting density data will necessarily depend on the spatial heterogeneity or patchiness of the species being sampled. Plant species that exhibit a regular or homogeneous spatial pattern can be sampled with fewer plots than species that grow in pronounced clumps. For species that are distributed at random throughout the study site, the precision of the density estimate will depend solely on the total number of individuals counted in the inventory, regardless of the size or shape of the individual sample plots used (Greig-Smith, 1983). As a general rule, the investigator should adapt a "more is better" philosophy and always try to sample as many plots as possible within a given habitat.

Several basic methods are available for determining the appropriate number of plots to use for a particular species or vegetation type. One method involves calculating the running mean, or variance, of the density estimates obtained from successive plot samples and then plotting these against the total number of plots sampled (Goldsmith & Harrison, 1976; Kershaw & Looney, 1985). In most cases, the variation in mean density will be quite high among the first plots sampled and then will gradually flatten out as the calculated sample mean begins to more closely approximate the true density of stems in the study area. An example of this technique is shown in Figure 2 using stem data collected from a series of 10 × 20 m contiguous plots in lowland dipterocarp forest within the Danau Sentarum Wildlife Reserve, West Kalimantan, Indonesia (Peters, unpubl. data). The curve exhibits a considerable amount of fluctuation up to a sample size of about

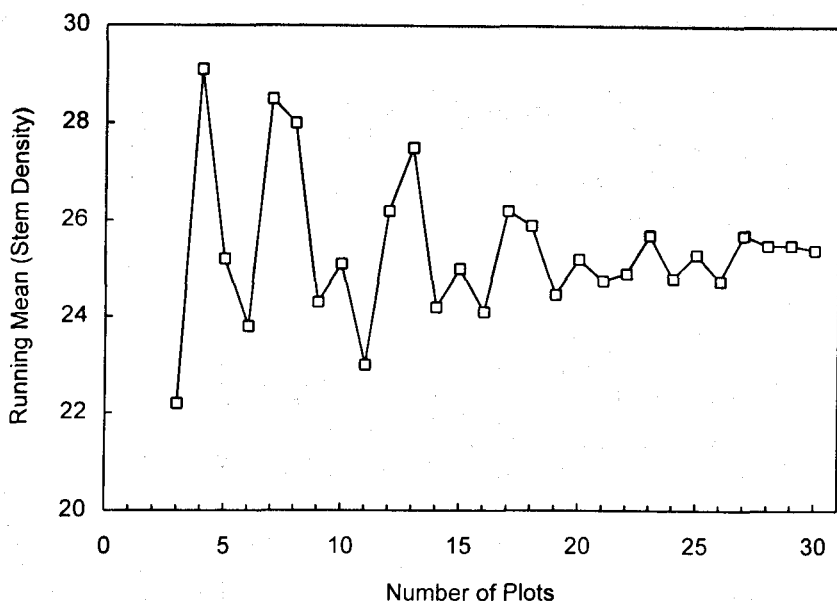


Figure 2. Running mean of number of stems (≤ 10 cm in diameter) per plot along a series of 30 contiguous 10×20 m plots. Data collected from a lowland dipterocarp forest in the Danau Sentarum Wildlife Refuge, West Kalimantan, Indonesia. (From Peters, unpubl. data).

22 plots (4400 m^2), at which point the variation in mean density starts to stabilize. This pattern suggests that a minimum sample size of approximately 25 plots (0.5 ha) would be sufficient for estimating the density of stems in this forest.

In many commercial timber surveys, it is common practice to set the sample intensity as a certain percentage of the total sample area to ensure that sufficient plots are sampled. Sample percentages of from 5% to 10% are usually standard (Avery & Burkhardt, 1983; Bonham, 1989). Assuming that a 5% sample percentage is desired, a 100-ha tract of community forest would need to be sampled with $50,000 \text{ m}^2$ of plots. This area could be obtained by using five $10 \times 1000 \text{ m}$ transects, fifty $20 \times 50 \text{ m}$ rectangular plots, or 100 circular plots with a radius of 12.62 m . The obvious problem with this method is that not all species and vegetation types require the same sample percentage. Blindly using a constant percentage in all situations will cause the investigator to use too many plots in some cases and not enough plots in others.

Several statistical techniques have been developed for estimating the exact number of sample units needed to obtain a given level of precision. These calculations, however, require an *a priori* estimate of the mean and variance of the population to be measured from a pilot survey or from a few preliminary plots that have been sampled in the study area. Detailed discussions of these techniques can be found in Husch et al., 1972; Köhl, 1993; and Philip, 1994. Most of these techniques require that the data be collected using a random sampling design (see below).

Arrangement of Sample Plots

A final consideration of great importance in the quantitative assessment of species density has to do with the way in which the plots are arranged throughout the study area. There are essentially two methods for deciding where to locate the sample units: the samples may be distributed regularly throughout the area in a systematic fashion, or they may be randomly located. A third method, the subjective placement of sample plots in typical or representative sites, is usually not recommended. Subjective sampling carries with it an excessive degree of personal bias, and the data collected using this procedure are not acceptable for any statistical tests involving the assessment of significance, such as *t*-tests, regression, correlation, or *F*-tests (Cochran, 1977; Greig-Smith 1983).

In systematic sampling, the sample units are spaced at fixed intervals. The location of the first sample is usually selected at random and all other samples are positioned according to a strict pattern. The actual sample units employed may be either transects or plots. Figure 3 illustrates the general layout of a systematic transect sample (Figure 3A) and a systematic plot sample (Figure 3B.). The square sample area shown is equivalent to a 100-ha tract of forest composed of three different forest types or land use categories (I, II, and III).

The transects shown in Figure 3A are 10 m wide and 1000 m long. Note that the transects are parallel and that they have been oriented at right angles to the rivers (solid black lines) so that all soil types and environmental conditions are intersected to provide a representative sample of the local vegetation. The 200-m interval between transects results in an overall sample intensity

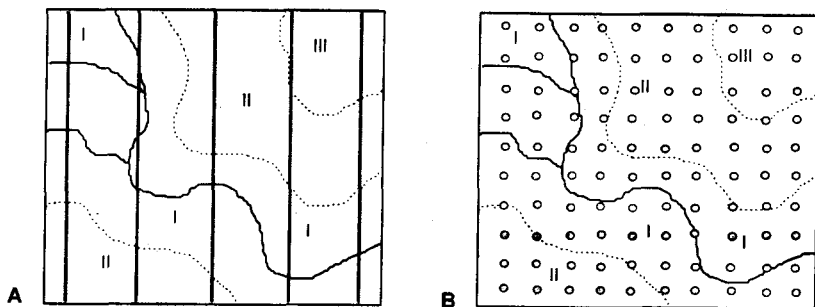


Figure 3. Arrangement of sample units in systematic sampling design. Square area shown represents 100-ha tract composed of three different forest types or land use categories (I, II, III). Solid black lines represent rivers; dotted lines represent type boundaries. **(A)** Systematic transect sample. Transects are 10 m wide, 1000 m long, and separated by 200 m, resulting in an overall sample intensity of 5%. **(B)** Systematic plot sample. Plots are circular, with a radius of 12.62 m, yielding a sample area of 500 m². Total sample intensity equals 5% (i.e., 100 plots \times 500 m² plot = 50,000 m², or 5.0 ha).

of 5%. The percent sampling intensity of a systematic transect sample is calculated simply by dividing the transect width (10 m) by the distance between transects (200 m) and then multiplying by 100. This feature is extremely useful in cases where the boundaries of a forest tract are known but the total area has yet to be determined. In the present example, a total of 5000 m of transect, or 50,000 m², were sampled. Given a sample percentage of 5%, the total area of the tract, if unknown, could have been calculated by multiplying the reciprocal of the sampling percentage (20) by the total sample area (50,000 m²) to give a result of 1,000,000 m², or 100 ha.

The basic design of a systematic plot sample is shown in Figure 3B. The plots are uniformly spaced throughout the forest in a grid pattern along north-south (10 columns) and east-west (10 rows) compass bearings. The circular configuration, with a 12.62-m radius (500 m²), of each of the 100 sample plots reduces the number of boundary trees and edge. The total sample area obtained by this design is 50,000 m² (100 plots \times 500 m² per plot), resulting in the same sample intensity (5%) as that provided by the transects. Given the even coverage provided by the grid layout, the plots can be oriented without worrying about the topography or drainage features of the site.

Forest inventories based on systematic sampling present several distinct advantages relative to other sample designs. First, they provide a good estimate of population means and totals because the sample area is spread out over the entire study site. Second, they are faster and less expensive to conduct than randomized designs because the location of the sample units is based on fixed directional bearings and distances. Locating transects or plots in the field is greatly facilitated, and travel time between sample units is minimized. Third, because the entire site is traversed in a regular, controlled pattern, supplementary forest type or land use information can be collected and easily mapped during field operations. Finally, systematic sampling does not require a priori knowledge of the total area of vegetation to be sampled.

A systematic design, however, has one major disadvantage. There is no satisfactory way to estimate the precision or sampling error of the data collected, because statistical variance computations require a minimum of two randomly selected sample units (Grieg-Smith 1983; Husch et al., 1972). In systematic sampling, only the location of the first plot or transect is selected at random; the remaining sample units follow a predetermined and regular pattern. This would not be a problem if all of the trees in a housegarden, agroforestry field, or managed forest were distributed at random and exhibited no pattern of variation. Unfortunately, the individuals in a biological population are rarely, if ever, arranged independently of each other, and there is a high degree of natural variability. It is, therefore, impossible using systematically collected data to separate the variability attributed to randomness from that naturally exhibited by the population. Although worthy of note, this limitation detracts very little from the overall utility of systematic sampling. In practice, the lack of an estimable sample error means only that the density data from two different areas cannot be compared statistically.

A random sampling design, on the other hand, provides not only mean and total density values but also an estimate of the precision of those values. The calculation of a standard error ($s_{\bar{x}}$) and confidence limits (CL) from the sample data, for example, allows the investigator to state that, at any given probability level, the true density value for the population or species lies within a certain specified range (Snedecor & Cochran, 1967).

Two examples of random inventory designs are illustrated in Figure 4. As before, the total area shown is 100 ha. There are 100 circular sample plots (each with a radius of 12.62 m), and the sample intensity is 5%. In simple random sampling (Figure 4A), all 100 plots are randomly located throughout the study area. A convenient method for determining the location of random plots is to place a transparent grid over a base map, aerial photo, or satellite image of the area and to draw randomly generated pairs of Cartesian coordinates for each plot. After marking the location of all plots on the base map or photo, derive compass bearings and distances from a central starting point to describe their relative position in the field.

The example shown in Figure 4B represents a stratified random sampling design. Although at first glance the pattern seems identical to that of simple random sampling, the important difference is that the plots have been "stratified" by forest type. The number of sampling units allocated to each forest type is determined by the percentage of the total area represented by each type, so that larger forest types contain a greater number of plots. Besides providing a more precise and efficient sample design, stratification also helps to avoid the uneven distribution or

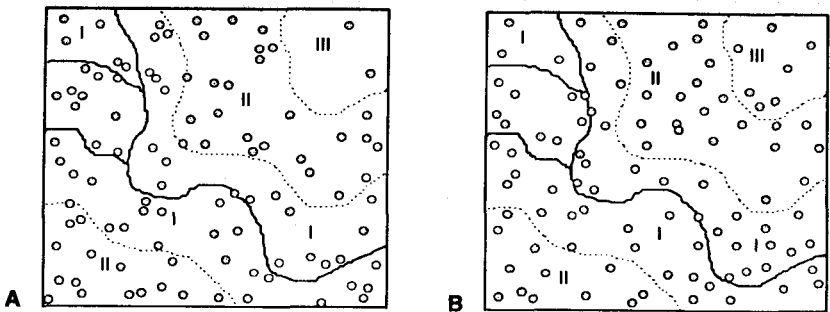


Figure 4. Arrangement of sample units in random sampling design. Square area shown represents 100-ha tract composed of three different forest types or land use categories (I, II, III). Solid black lines represent rivers; dotted lines represent type boundaries. All (100) plots are circular, with a radius of 12.62 m (total sample area 500 m²); plot location is based on randomly selected coordinates. Overall sample intensity is 5%. **(A)** Simple random sample. Plots are located randomly throughout the entire area. Note that forest type III (upper right corner of figure) is sampled by only two plots. **(B)** Stratified random sample. The number of plots allocated to each forest type is based on percentage of total area represented by that type. Note that forest type III has now been sampled with seven plots

clumping of plots that frequently occurs with random sampling (Adlard, 1990; Philip, 1994). In Figure 4A, for instance, a large area of type I forest remains unsampled. The same procedure used to locate plots for simple random sampling can be used for stratified random sampling. Random coordinates that place a plot within a forest type requiring no further sampling are simply rejected.

The clear advantage of simple random sampling is its statistical rigor. Precise confidence limits can be assigned to all of the data, and, given information on the natural variability of the population (e.g., from a preliminary sample), the minimal number of plots that need to be used to adequately describe the forest can be calculated. There are, however, several disadvantages to a random design. The plots can be very difficult to locate in the field, and much time is wasted traveling from one plot to the next. In some cases, the random selection of plot location may leave significant sections of the study area unsampled. Perhaps the greatest limitation, however, is that random sampling does not allow the regular, grid-based observations necessary for detailed forest type or land use mapping.

Both random and systematic sampling are routinely used to collect plant density data. Wood (1989) reported that systematic sampling is the preferred design for commercial timber inventories in Africa and Southeast Asia, whereas random sampling is more strongly favored in Latin America. Of the 36 tropical countries surveyed in Wood's study, fixed-area plots had a higher frequency of use (44%) than transects (34%). In terms of ethnobotanical research, systematic (Kinnard, 1992; Lepofsky, 1992) or subjective (Balée, 1994; Boom, 1989; Prance et al., 1987) transects seem to be the preferred sampling method, although some investigators have opted for a random (Pinedo-Vasquez et al., 1990) or stratified random (Irvine, 1989; Salick, 1989) design. Single 1.0-ha sample plots have also been used in several recent studies (e.g., Fong, 1992; Phillips, 1993).

Field Procedures and Measurements

Regardless of the sampling design selected, great care should be taken in locating and laying out the sample plots in the field. If the plots fall in the wrong place, or are the wrong size, it is very

likely that the final density estimate obtained will also be wrong. Of special importance in this regard is the correction for slope. Ensuring consistency and comparability requires that all distance and area measurements be made along the horizontal. Measuring 20 m along a 10% slope yields a very different horizontal distance (19.9 m) than measuring 20 m along a 40% slope (18.6 m). In both cases, however, the horizontal distance obtained is less than that desired. In terms of geometry, measuring along a slope is like measuring the hypotenuse of a right triangle, when what the investigator should really be trying to measure is the base of the triangle.

Failure to correct for slope can lead to significant measurement errors. Take, for example, the case of a 10×1000 m transect laid out along a constant 30% slope. If no correction is made for the topography, every 20 m measured along the transect will be 0.8 m too short and each 10×20 m segment of the transect will contain 192 m^2 , rather than 200 m^2 . By the end of transect, the sample unit will be 40 m too short and will contain 400 m^2 less than it should. The density data from this sample are clearly not comparable to those collected from a 10×1000 m transect laid out along flat terrain.

Table I shows one way to avoid this problem. Slope corrections are tabulated for different distances and percent slopes. As indicated in the table, the measurement of 10 horizontal meters along a 30% slope requires a distance of 10.44 m. Meter tapes or ropes can be prepared in advance to facilitate the use of these correction factors in the field. If measurements are made at 10-m intervals, for example, a 15-m tape or rope can be marked or knotted at the appropriate distances for the range of slopes expected to be encountered in the field. One crew member pulls the tape 10 m, while a second stays behind to take a slope reading (e.g., with a clinometer or Abney level). If the slope was determined to be 50%, then the tape would be extended to the 11.2 m mark before setting a plot stake or tallying the next 10-m segment of the transect.

Which Plants to Count?

Two points in particular should be addressed in deciding which plants to count in each of the plots. The first is related to the

Table 1. Slope corrections for different distances and percent slopes. Table values indicate the distance along a slope that must be traveled to obtain the horizontal distance indicated by the column heading.

Slope (%)	Horizontal distance (m)				
	5	10	15	20	25
10	5.02	10.05	15.07	20.10	25.12
15	5.06	10.11	15.17	20.22	25.28
20	5.10	10.20	15.30	20.40	25.50
25	5.15	10.31	15.46	20.62	25.77
30	5.22	10.44	15.66	20.88	26.10
35	5.30	10.59	15.89	21.19	26.49
40	5.39	10.77	16.16	21.54	26.93
45	5.48	10.97	16.45	21.93	27.41
50	5.59	11.18	16.77	22.36	27.95
55	5.71	11.41	17.12	22.83	28.35
60	5.83	11.66	17.49	23.32	29.15
65	5.96	11.93	17.89	23.85	29.82
70	6.10	12.21	18.31	24.41	30.52
75	6.25	12.50	18.75	25.00	31.25
80	6.40	12.81	19.21	25.61	32.02
85	6.56	13.12	19.69	26.25	32.81
90	6.73	13.45	20.18	26.91	33.63
95	6.90	13.79	20.69	27.59	34.48
100	7.07	14.14	21.21	28.28	35.36

minimum size limit of the individuals to be included in the sample. The second has to do with deriving an operational definition of the word *individual*. Both of these issues should be resolved before fieldwork is begun.

The minimum size limit used in an inventory exerts a controlling influence on the total number of plant stems that have to be counted. In most cases, the smaller the minimum size limit, the greater the number of sample plants that are included. To illustrate this relationship specifically for trees, inventory data from 1.0-ha samples of hill dipterocarp forest and managed forest orchard in West Kalimantan, Indonesia, are shown in Figure 5. The stem counts from these samples (Peters, unpubl. data) were grouped into 5.0 cm diameter classes. In both environments, the increase in the number of sample trees is linear down to a diameter of about 25–30 cm. Further decreases in minimum diameter,

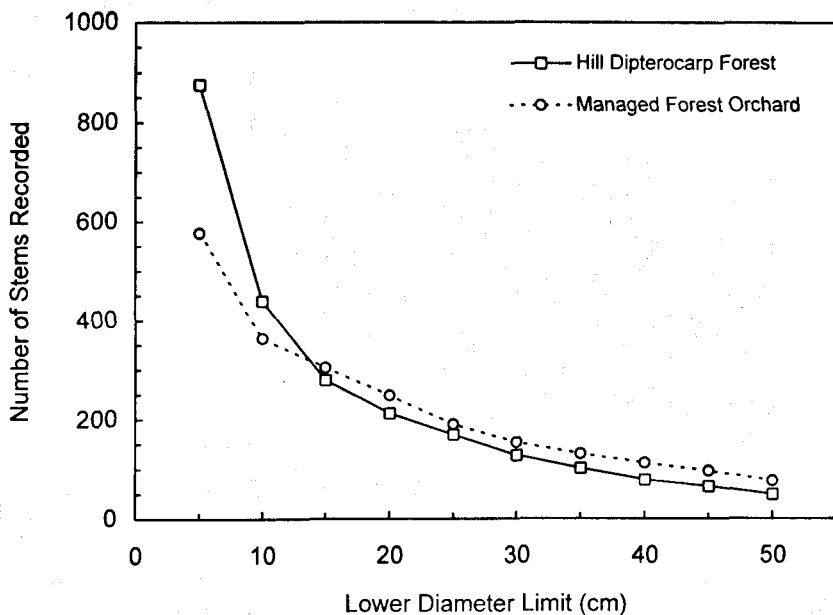


Figure 5. Number of stems recorded in 1.0-ha samples of hill dipterocarp forest and managed forest orchard using different minimum diameter limits. Data for hill dipterocarp forest were collected in the Raya-Pasi Nature Reserve; managed forest orchard data are from the Dayak village of Bagak Sahwa. Both sites are located in the Sambas district of West Kalimantan, Indonesia. (From Peters, unpubl. data.)

however, result in an exponential increase in the number of stems; the density of 5.0-cm sample trees ($n=876$) is almost twice that of 10.0-cm trees ($n=439$) in the unmanaged forest. The lower number of small stems in the managed forests reflects the periodic thinning and selective weeding practiced by local Dayak communities (Padoch & Peters, 1993).

The pattern shown in Figure 5 underscores the trade-offs involved in choosing a size limit for inventory work. A smaller minimum size increases the amount of information obtained from the sample, but it also greatly increases the time and expense of fieldwork. Larger diameter cut-offs significantly speed up field operations, but they may result in an unrepresentative sample of certain plant resources. Many important forest fruits, for instance, are understory and midcanopy species, and these resources would be completely missed by adopting the 20–40 cm diameter limit used in many commercial timber surveys (Heins-

dijk & de Bastos, 1965; UNESCO, 1978). As a compromise between time invested and information obtained, a large number of ethnobotanical studies (e.g., Phillips, 1993; Pinedo-Vasquez et al., 1990; Prance et al., 1987) and floristic surveys (e.g., Campbell et al., 1986; Gentry, 1988) have used a 10-cm minimum diameter limit. Studies focused specifically on the collection of density data for acaulescent palms, lianas, bamboos, or herbaceous plants would, of course, require the use of an even smaller minimum size limit.

For most tree species, it is relatively easy to distinguish one genetic individual, or *genet*, from another. They originate from a single seed, produce a single trunk, and occupy a well-defined and exclusive space in the forest. The situation is a bit more complicated for bamboos, caespitose palms, and many herbaceous plants that reproduce vegetatively and have the ability to form dense, multistemmed clumps. In these species, a single genetic individual may be represented by innumerable clonal shoots, or *ramets*. Faced with an impenetrable stand of bamboo, or a rattan clump with over 200 spiny stems, or a dense sward of grass with innumerable tillers, the investigator may be hard pressed to even distinguish where one individual stops and another begins. What should be counted—the individual clumps (*genets*), the individual stems (*ramets*), or both?

Probably the best rule of thumb in these cases is to try to quantify the same vegetative unit as that which is actually exploited as a resource. If an entire sward of grass (i.e., one genet composed of many ramets), for instance, is pulled up and used as thatch, counting the number of clumps per plot will probably yield a reasonable estimate of the density of this resource. Bamboo poles (i.e., ramets), which are harvested individually, would best be assessed by counting all of the culms in each plot. In some cases, however, it is clearly not feasible to count all of the individual ramets produced by a species (e.g., a dense cluster of rattan or an extensive bamboo forest). Counting all the clumps (*genets*) in the plot and then choosing a subsample of individuals of varying size for counting ramets is a useful strategy for getting around this problem. Once all of the plot data have been collected, regression analyses can be used to derive a predictive equation describing the relationship between genet size and number of ramets. Based on the total number of clumps counted in

the plots, this equation can be used to provide a rough estimate of the total number of harvestable rattan canes or bamboo stems growing in the study area.

What to Measure?

Although the collection of density data is essentially a counting process, there is much to be gained by measuring the size of each of the sample individuals encountered in the plots. For example, it is useful to know that each hectare of your study area contains 100 fruit trees >10 cm dbh. Determining that 40 of these trees are over 30 cm in diameter and of sufficient size to actually produce fruit is a finding of even greater utility. Information about the size distribution of individuals in a population can also frequently provide indirect evidence about the regeneration success of that species. A population composed of 100 trees per hectare with all of the individuals over 50 cm in diameter is very different from one composed of 100 trees per hectare with 20 trees 50 cm in diameter, 30 trees of 30 cm dbh, and 50 trees of 10 cm dbh. The latter population appears to be regenerating itself quite well. The former is probably destined to disappear from the site as soon as the big trees die. The really important question here is not so much What is the total density of species A within the study area? as What is the density of different-sized individuals [i.e., the size structure] of species A on the site? Answering this question requires that the sample plants be measured as well as counted.

The most frequently measured and easily obtainable expression of tree size is diameter at breast height, or dbh (approximately 1.4 m above the ground). In cases where extreme buttress formation, wounds, or forked boles preclude the measurement of dbh, a section of clear trunk immediately above the problem area should be measured, and a note that this was done should be recorded near the diameter measurement for that individual (Philip, 1994). Depending on the resource, dbh may not always be the most meaningful size parameter to measure. Basal diameter (i.e., at the ground level) is a more useful index to classify the size structure of shrub populations (e.g., Peters & Vazquez, 1987; Reid et al., 1990), and height, although difficult to measure with precision in closed forest, may be the only alternative for

members of those taxa (e.g., palms, bamboos, and tree ferns) that do not grow in diameter (Ash, 1987; Bullock, 1980; Pinard, 1993; Piñero et al., 1977).

Growth and Yield Studies

An ethnobotanist assessing the use of resources by a local population is interested in *how many* individuals of a particular species are growing in local forests, agroforestry fields, or home gardens. These density estimates, however, are only half of the plant-use equation. To really evaluate the quantity of resource available to local populations, one must also quantify *how much* of the desired resource is produced by each of these individuals. Foresters routinely collect this type of data by monitoring the radial increment of timber trees, and there is a large and detailed literature on the growth and yield characteristics of commercial timber species (e.g., Adlard, 1990; Alder, 1980; Wan Razali et al., 1989). The situation, however, is quite different for nontimber resources. Virtually nothing is known about the fruit, oil seed, latex, or resin yield of forest species or local cultivars, even for the most valuable and widely exploited market species. How many Brazil nuts does a large *Bertholletia* tree produce? How much rattan cane does a wild *Calamus* clump make in a year? What is the rubber yield from a large *Hevea* tree in the lowland forests of Amazonia? Although fertile ground for ethnobotanical inquiry, these questions remain essentially unanswered.

Selection of Sample Trees

The basic objective of a yield study is to provide a reasonable estimate of the quantity of resource produced by a given species growing in a particular habitat. As it is rarely feasible to monitor all of the individuals of a selected species, data collection will necessarily focus on a subsample of plants. If at all possible, the selection of these sample plants should be stratified by two main variables—diameter, or some other indicator of plant size, and site condition.

There are several good reasons for conducting a stratified sample. Regardless of the species or type of resource produced, plant size exerts a major influence on yield. Large plants, because of

their better canopy position, larger leaf area and root mass, and greater availability of stored carbohydrates, are usually significantly more productive than smaller plants (Kozlowski et al., 1991). The actual parameter of interest, therefore, is not simply mean production but the size-specific production rate of the species. As will later be discussed in this section, the exact nature of the statistical relationship between size and yield is an important tool for deriving an estimate of the total annual productivity of a given plant resource.

Plant productivity also varies with respect to certain site parameters. Even after the effect of size has been accounted for, most species will usually exhibit higher yields in some sites than in others. The key variable of interest may be soil depth and fertility, soil moisture, canopy cover, relative slope, forest type, or presence and absence of cultural treatments such as thinning, weeding, or mulching. Whatever variables are selected, the important thing is that the investigator be able to stratify or partition the local habitat along these lines. The selection of slope as a growth parameter, for example, would require some knowledge of the areal extent of different slope conditions within the study area, as well as an estimate of the density of plants occurring in each slope class. These data, or those for soil condition, canopy cover, forest type, or management status, could be collected during the inventory work by taking the appropriate measurements in each sample plot.

There are no hard and fast rules to determine the number of sample plants that should be selected to assess the yield characteristics of a particular species. In many cases, the issue will resolve itself on the basis of the relative density of the species in different site conditions, the size distribution of the population, the number of individuals in each diameter or height class, and the actual time and expense available for conducting yield studies. As was discussed earlier in regard to the number of sample plots, the greater the number of individuals that can be sampled, the greater the accuracy of the final estimate.

Using the results from the plot survey as a guide, the investigator should randomly select individuals from different size classes and habitats. If at all possible, the number of sample trees selected from each size class should be the same in each site condition or forest type. Ideally, every size class should be sampled

by *at least* three individuals so that some index of variability (such as standard error) can be calculated. Size classes can sometimes be lumped together to achieve this objective if fewer than three individuals per class are available. A reasonable level of precision can be ensured if the total number of sample plants selected in each site condition falls within the range of 25–30 trees (e.g., 5–6 trees in each of five size classes).

After the sample plants to be measured have been selected, these individuals should be located in the field, sequentially numbered, and permanently labeled with paper or plastic tags. If sufficient time and funding are available, additional information such as crown area or number of leaves (for palms), canopy cover, and distance to and size of nearest-neighbor trees can also be collected from each sample individual at this time. These data can later be grouped into classes and compared statistically to provide a more detailed analysis of size-specific productivity.

Methodology and Data Collection

The exact sampling procedure used in the yield studies will necessarily vary with the type of resource being measured. For ease of discussion, the innumerable useful products produced by plants can be divided into three main groups based on the origin of the plant tissue or compound being used: reproductive propagules (e.g., fruits, seeds, and accessory tissues), plant exudates (e.g., latexes, resins, and gums), and vegetative tissues (e.g., stems, leaves, roots, barks, and apical buds). Although fruits, nuts, and oilseeds are different commodities, their production by individual trees can be measured using a similar methodology.

Reproductive Propagules

The production of fruits and seeds is measured at discrete intervals throughout the fruiting season using either direct counts or a random sample of the area under the crown of adult trees. For small trees that produce few fruits of relatively large size (e.g., shrubs, some palms, and cauliflorous trees), direct counts of fruit can be employed with reasonable precision (Dinerstein, 1986; Peters & Vazquez, 1987; Piñero & Sarukhan, 1982; Sork, 1987). It is usually a good idea to make replicate fruit counts on the same individuals until a consistent number is obtained. If nuts or seeds

are the actual resource of interest and the fruits in question are multiseeded, a large number of mature fruits should be opened ($n=50-100$) and the seeds counted to determine the average number of seeds per fruit.

Direct counts can also be used with species that produce multiple fruits in large infructescences (e.g., many palms) by harvesting these structures when mature and carefully counting the number of fruits (e.g., Anderson et al., 1985; Phillips, 1993). An alternative strategy is to first record the total number of infructescences produced by the tree and then to harvest a subsample for counting individual fruits. Given the ease of direct counts, it is tempting to use this procedure on large-fruited canopy trees by scanning the crown with binoculars. This technique, however, is not recommended as there is no way to mark the fruit that have already been counted, and it is extremely difficult to survey the entire crown of a large tree without some degree of overlap or repetition. Chapman et al. (1992), for example, compared visual estimates of fruit production by two canopy species in Uganda with actual counts of the total fruit crop collected under the crowns of the trees and found correlations of only marginal significance between the two data sets.

Tall forest trees that produce more fruit than can be counted individually must be sampled using small plots or specially constructed fruit traps (see review in Green & Johnson, 1994). A critical assumption involved in using this method is that a large percentage of the fruits will fall directly under the crown of the adult tree. For most commercial fruits, nuts, and oilseeds, which are relatively large and heavy, this assumption probably is valid. However, sampling under the crown of a tree will not account for the fruits and seeds that are eaten or dispersed by animals before they fall, and, as a result, the data collected will not represent the total number of fruits produced. This limitation notwithstanding, the use of fruit traps or plots does provide a reasonable estimate of postpredation or harvestable yield, which may actually be a more relevant and useful measurement for the ethnobotanist than is total yield.

The first step in the sampling process is to determine the exact area of the vertical projection of the crown of each sample tree. This is accomplished by measuring out from the trunk of the tree to the outermost branches of the crown along at least four

radii. On the basis of these measurements, the actual projection or "shadow" of the crown is sketched on millimetric graph paper and its area calculated using the appropriate formula for that configuration (e.g., circle or ellipse). A stratified random design is then used to allocate the sample plots or traps within this area. The crown area is divided into four quadrants of similar area; the boundaries of these quadrants are determined by four perpendicular radii extending out from the trunk. Random coordinates are then chosen to position the samples within each quadrant. The reason for this stratification is that fruits rarely fall in a symmetrical or regular pattern under a tree. Prevailing winds and the relative position of fruit-laden branches usually cause more fruits to fall on one side of the crown than the other. Dividing the crown projection into quadrants will ensure that regions of both high and low fruit density are sampled.

There are two options for determining the number of sample plots or traps to be used under each tree. A constant percentage of the crown area can be sampled (Howe, 1980; Howe & Vande Kerckhove, 1981), or, alternatively, a constant number of traps can be used irrespective of crown area (Howe, 1977; Peters, 1990; Peters & Hammond, 1990). The former method requires that a greater number of samples be located under large trees than small ones, and certain statistical tests may be complicated because of the unequal sample sizes (Sokal & Rohlf, 1981). The latter method samples smaller trees more intensively than large ones. If a fixed sampling percentage is desired, a sufficient number of traps should be used to sample about 10% of the total crown area. If a constant number of traps of sample units is used, a total of 8–12 traps or plots (i.e., 3–4 per quadrant) should be located under each tree.

Although traps may have a slight advantage over plots in that fruits cannot roll out of the sample unit, plots are faster to lay out and easier to maintain. The most common plot size used in fruit production studies is a 1×1 m square. The plots under each tree should be numbered sequentially, the corners staked, and the boundaries clearly delineated with plastic string or flagging. Raking the plot down to mineral soil and maintaining it free from leaves and vegetation can greatly facilitate the locating of fallen fruits.

Fruit traps can be from 0.5 to 1.0 m² in size and of either a

square or circular configuration. The smaller traps are somewhat more stable and easier to transport to the field. Square traps are constructed by first making a box frame out of 1×4 cm wooden battens and then stretching a piece of 2-mm nylon netting tightly over the bottom and affixing it with tacks or staples (see description in Adlard, 1990). Circular traps can be made out of stiff wire or plastic tubing; larger fruits require a stronger and more durable trap than smaller ones. A 79.8 cm diameter circle has an area of 0.5 m; a 112.8 cm diameter circle provides a 1.0-m^2 sample area. Nylon screening is used to make a loose, concave net (approximately 30 cm deep), which is then tied or clipped to the circular frame; plastic clothespins work very well for clipping the bag to the hoop. Both square and circular fruit traps should be elevated about 50 cm off the ground using treated wooden stakes or PVC pipe, and the number of the trap should be clearly marked on the leg or frame.

Fallen fruits start to decompose quite rapidly on the forest floor, and there is always the possibility that some fruits will be eaten or removed by animals before they are counted. Fruit predation between sampling periods can also be a problem with traps, which are easily climbed by squirrels and other forest rodents. Reviewing the traps or plots as frequently as possible, preferably twice a week, will help prevent these potential sources of error. At each sampling period, the number of immature and mature fruits in each sample unit should be carefully counted and all of the reproductive material removed from the plot. Screens, stakes, and plot boundaries should also be checked at this time and repaired if necessary. The biweekly sampling of each tree should be continued until at least two consecutive fruit counts give null results.

Plant Exudates

The measurement of plant exudate yield requires some a priori knowledge of the traditional tapping or collection technique used for a particular species. Of special importance is information concerning the frequency with which the trees are usually tapped. Through a continual process of trial and error over the years, experienced collectors have undoubtedly determined the tapping method and harvest schedule that produces the greatest amount of latex, resin, or gum. The objective here is to actually quantify this yield.

Perhaps the easiest way to obtain these data is through careful participant observation; work with an experienced local assistant and follow him around as he taps the sample trees (see Barrera de Jorgenson, 1993; Dove, 1993; Gianno, 1986). The exudate obtained from each tree is measured (by weight or liquid volume depending on the resource) and recorded in the field, and, with the help of the local assistant, an initial estimate is made of the frequency with which the tree can be tapped. Several sequential tappings should be measured to obtain some idea of the variability in yield, as well as to observe the tree's response to the wounding caused by harvesting. Depending on the particular tapping regime employed, daily, weekly, or monthly production rates are then calculated for each sample tree and exudate under study.

Vegetative Tissues

The variety of vegetative structures exploited as resources—e.g., stems, leaves, barks, roots, apical buds—can be divided into two groups based on the physiological response of the plant species to harvesting: the plant species will either survive and later regenerate the vegetative structures removed, or it will be killed by harvesting the tissue. The former group includes leaves, branches, and the bark and apical buds of certain species; the latter includes most types of stem tissue, roots, and bark. Different sampling methodologies are required to estimate the productivity of these two groups.

For species that exhibit regrowth or sprouting, the basic idea is to first quantify the existing stock of harvestable resource and then to monitor the rate at which these resources are replenished by the plant. The periodic collection of palm leaves provides a useful example to illustrate this concept. Working with experienced collectors, the investigator records the average number of leaves harvested from the crown of each sample individual together with data on the total number of leaves per crown. The residual leaves on each individual should be marked with paint or tags to differentiate them from the new leaves that are later produced. After an adequate period of time has passed for the new leaves to fully elongate, the palm is reharvested and the leaf number is again determined. This procedure should be followed through at least two cycles of harvest and new leaf production to get some idea of whether the rate of leaf production decreases in

response to repeated harvest. The mean yield figure for each tree represents the total number of new leaves harvested throughout the sample period. The final result should be adjusted to reflect a yearly production rate. Similar methodologies have been used in studies of plant demography to quantify the leaf replacement rates of palms (Bullock, 1980; Lugo & Rivera, 1987; Oyama 1990; Sarukhan, 1978), cycads (Clark & Clark, 1987), and tree ferns (Tanner, 1983).

The procedure for collecting yield data for species that are killed by harvesting is a bit more complicated. Measuring root growth or the production of bark, for example, is plagued with methodological difficulties, and these resources require a two-step sampling scheme involving allometric analyses and growth studies. During the first phase of this process, the preselected sample plants are felled, dissected, and carefully measured to obtain an estimate of the size-specific bark or root volume for that species. Regression analyses are then used to derive a predictive equation relating plant size to quantity of resource present. The slope of this regression line can eventually be used to predict yield.

The problem, however, is that there is, as yet, no time dimension or rate associated with the production of the resource. What is lacking is information about the rate at which these plants grow from one size class to the next. Collecting this information requires selection of a second subsample of plants representing a range of different sizes and habitats; growth of the subsample plants is monitored for at least one year. Diameter growth is the best parameter to measure for most tree species, and these data can be collected by using dendrometer bands (Bormann & Kozlowski, 1962; Liming, 1957) or by making periodic diameter measurements on the same sample trees. In the latter method, painting a line on each of the sample trees to indicate the original point of measurement is highly recommended. Height growth is clearly a more meaningful parameter to measure for palms, herbs, and understory plants.

Combining the data sets from the allometric and growth studies provides a reasonable estimate of the productivity of a particular root or bark resource. For example, if the bark biomass of a 20-cm *Cinchona* tree is 11.0 kg and the bark biomass of a 25-cm tree is 20 kg (Hodge, 1948), a 20-cm tree growing at 0.5 cm per year would produce approximately 900 g of bark tissue a year. If

necessary, the order of the allometric and growth studies can be reversed, with the growth studies being conducted first and the same sample trees later being harvested and analyzed. This strategy, which requires only one group of sample trees, may be warranted for species occurring in low-density populations.

Solitary rattans are an especially difficult subject for yield studies. In many respects, the harvest of these resources is identical to logging in that entire stems are removed and, for many species, there is no resprouting (Dransfield & Manokaran, 1994). The problem is that there is no easily measurable indicator of growth, such as diameter (dbh), as is used by foresters to estimate productivity. Rattans, like all palms, have no secondary meristem and exhibit no radial growth. They produce new stem tissue (cane) solely by extension growth. For small and intermediate-sized individuals, height growth can be measured directly to obtain an estimate of cane yield. Measuring the height increment of the larger, more valuable canes, some of which may be 40–50 m long, is quite a bit more difficult and requires tree climbing.

There is usually no way to get around this problem. Basing yield figures solely on the extension growth of smaller, and frequently slower-growing, individuals will lead to an underestimate of productivity. Periodic controlled harvests can be used to estimate the local stock of rattan cane, but this procedure ignores the critical issue of size-specific yield. Perhaps the only recommendation that can be made is to try to measure at least a few large-sized canes. To achieve this objective, one must climb each individual to locate its apical bud or growing point. A point on the stem immediately behind the bud should then be permanently marked with paint and tied with flagging to facilitate relocation. After 6–12 months, the climber should enter the canopy again and carefully measure the distance from the paint mark to the end of the apical bud. The average growth rate taken from several large canes could then be applied to all large-sized, canopy individuals.

Defining the Resource Base

The results from the fieldwork described thus far can be integrated to estimate the total quantity of harvestable resource pro-

duced by different plant populations within the study area. These data, which represent the size of the resource base for a particular species, can be used to analyze local patterns of resource exploitation, to forecast future yields and harvest revenues, and to assess the ecological sustainability of current harvest levels. They are, in essence, the foundation upon which resource management is based.

Two pieces of information are needed for this analysis: size-specific production data from the growth and yield studies and the size structure of the population obtained from the inventory data. Although the following discussion uses rattan as an example, the basic procedure for estimating total population yield is essentially the same for all types of plant resources.

The size-specific production data collected in the yield studies are first grouped by habitat or site class and then regression analyses are performed to derive a predictive equation describing the relationship between plant size and productivity. In some cases, the functional relationship between these two variables will not be linear, and the data may require some type of transformation (e.g., conversion to logarithms) before they are analyzed (Sokal & Rohlf, 1981). It is also possible that after inspection the data could be best described using curvilinear or polynomial regression techniques. Whatever degree of analysis is used, however, the objective is to produce a result that is both biologically meaningful and statistically significant.

One example of the form that this relationship might take is shown in Figure 6 using data collected for *Calamus schistoacanthus* Bl. in the Danau Sentarum Wildlife Reserve in West Kalimantan, Indonesia (Peters, unpubl. data). This rattan is an important source of cordage and weaving material for local fishing communities, and large quantities are harvested every year for both subsistence use and sale. The species occurs naturally in high-density stands in the seasonally flooded forests of the reserve. Growth data were collected in 1994 from a total of 78 *C. schistoacanthus* individuals representing a range of height classes. As is indicated by the regression line in Figure 6, there is a linear relationship between plant size and extension growth for canes up to 5 m in length, and the relationship appears to be strong enough from a statistical standpoint ($r^2 = 0.774$; $P < 0.01$) that stem growth can be reliably predicted from cane length.

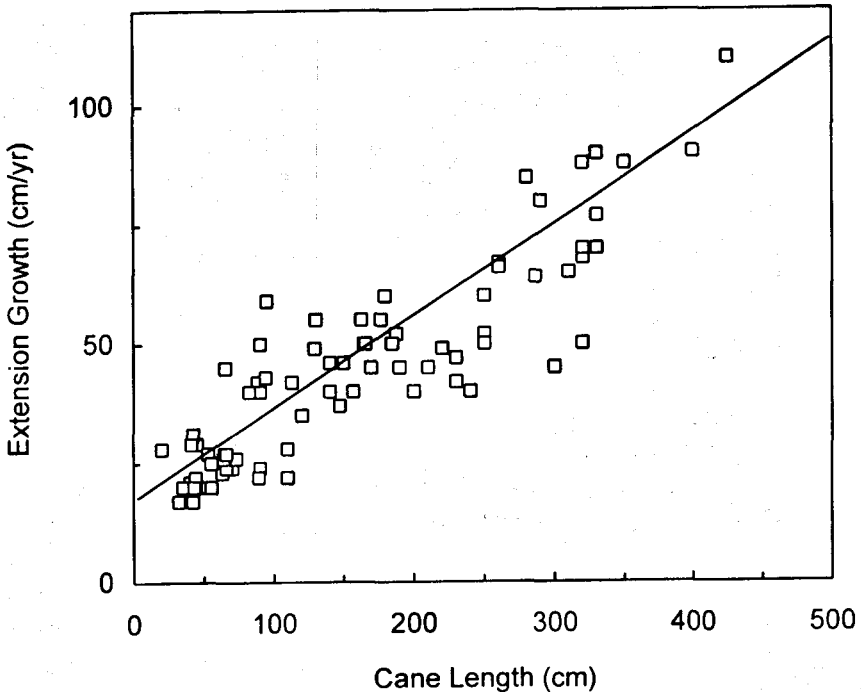


Figure 6. Annual extension growth as related to cane height for *Calamus schistoacanthus* ($n=78$) growing in the seasonally flooded forests of the Danau Sentarum Wildlife Reserve, West Kalimantan, Indonesia. The regression line is based on the general linear model, $\text{growth} = a + b (\text{height})$; the parameter values and coefficient of determination are: $a = 15.76$, $b = 0.186$, $r^2 = 0.78$, $P < 0.01$.

The final equations obtained from the yield studies are used to estimate the collective productivity of each of the appropriate size classes (i.e., those containing individuals of reproductive or merchantable size) in the population. These estimates are calculated by substituting the midpoint of each size class as the dependent, or y , variable in the yield equation. The average yield value for each size class is then multiplied by the actual number of individuals within that class to obtain a class total. Summing these totals over all size classes provides an estimate of total population yield. Care should be taken to include only productive individuals in these calculations. The male trees of dioecious species, for example, obviously should be omitted from an analysis of total fruit yield.

The inventory and growth data for *C. schistoacanthus* presented

in Table II illustrate this procedure. The density data (Peters, unpubl. data) were collected from 1.4 km of 10-m wide transects sampled at the Danau Sentarum Wildlife Reserve in the same area as the yield studies. At least three points of interest are illustrated by the data shown in Table II. The first is related to the extremely high density of the *C. schistoacanthus* populations at Danau Sentarum. Over 750 individual clumps per hectare were recorded in the transects, and, on the basis of stem counts made on a subsample of individuals, these clumps contained an estimated 12,200 canes/ha. The fact that *C. schistoacanthus* is one of the few Bornean rattans that can tolerate severe seasonal flooding is probably largely responsible for the notable abundance of this species.

The second point is that the total cane yield by the species is a function of both the size-specific growth rate and the density of canes in each size class. The larger size classes grow faster, but they represent a small percentage of the total annual productivity because of the limited number of clumps of this size in each hectare. Small clumps are very abundant, but they exhibit a growth rate that is less than a third of that shown by taller canes. Most

Table II. Estimated annual yield of rattan cane by a 1.0-ha population of *Calamus schistoacanthus* growing in seasonally flooded forest at the Danau Sentarum Wildlife Reserve, West Kalimantan, Indonesia

Height class (m)	Clumps/ha	Canes/ha ¹	Estimated mean growth (cm/yr) ²	Total growth/class (m)
0.0–1.0	327	3177	25.1	797.4
1.0–2.0	174	2234	43.6	974.0
2.0–3.0	99	1679	62.3	1046.0
3.0–4.0	57	1277	80.9	1033.1
4.0–5.0	36	1066	80.9	862.3
5.0–6.0	32	1251	80.9	1012.1
6.0–7.0	18	930	80.9	752.4
7.0+	11	653	80.9	528.3
Total	754	12,267		7005.6

¹ Estimates of the number of canes (ramets) per clump (genet) were based on counts of a subsample of clumps ($n=46$) and calculated using the equation $\log_{10}(\text{number of canes}) = a + b(\text{height of cluster})$; $a=0.927$, $b=0.121$, $r^2=0.82$.

² Size-specific growth estimates were calculated using the regression equation $\text{growth} = a + b(\text{height})$, where $a=15.76$, $b=0.186$, $r^2=0.78$.

of the rattan cane produced each year by the *C. schistoacanthus* population comes from individuals of intermediate size (2.0–6.0 m tall).

Finally, the data shown in Table II provide some indication about what a sustainable level of rattan harvest might be from these populations. An estimated total of 7005 m of cane per hectare are produced every year by *C. schistoacanthus*. Of this total, 3155 m of cane are produced by individuals of merchantable size (i.e. the four size classes >4.0 m tall). Given that the minimum length of harvested cane is usually 4.0 m, this figure represents a mean annual productivity of approximately 790 canes/ha. Every year, at least half of the individuals in the 3.0–4.0 m size class will grow into the 4.0–5.0 m merchantable class. The “in-growth” of these smaller individuals expands the local rattan resource base by about 650–700 new merchantable canes. If we assume that the density estimates are representative and that the measured growth rates are maintained over time, the data in Table II suggest that about 700 *C. schistoacanthus* canes per hectare per year could be harvested on a sustained-yield basis from the flooded forest of Danau Sentarum.

Conclusions

Ethnobotany and plant ecology are natural partners, and their collaboration can contribute greatly to the study of people and plants. Coupling plant use information with quantitative data on the distribution, abundance, and yield of different resources provides a useful new framework for addressing the question, How important is this species? Perhaps of even greater relevance given current realities, however, is that this integrated focus also allows the investigator to probe deeper into the questions, How quickly is this resource being used up? What can be done to prevent overexploitation? The conservation and rational use of the innumerable plant resources “discovered” by ethnobotanists over the last 100 years will inevitably require the collection of density and yield data. Although the fact is seldom mentioned, ethnobotanical research is really the first step toward effective resource management. The more ambitious the first step, the faster effective resource management can be achieved.

Acknowledgments

I thank Miguel Alexiades for his patience and persistence in motivating me to write about the links between ecology and ethnobotany. Fieldwork in West Kalimantan, Indonesia, was conducted under the auspices of the Indonesian Institute of Science (LIPI) in collaboration with Tanjungpura University in Pontianak, and the continual support of those two institutes is gratefully acknowledged. Funding for the research in Indonesia was provided by the World Environment and Resources Program of the John D. and Catherine T. MacArthur Foundation. I thank Wim Giesen, Eddy Zulkarnain, Budi Suriansyah, and Julia Aglionby for their invaluable assistance during the rattan surveys at Danau Sentarum.

Literature Cited

- Adlard, P. G.** 1990. Procedures for monitoring tree growth and site change: A field manual. Tropical Forestry Paper No. 23. Oxford Forestry Institute, Oxford.
- Alder, D.** 1980. Forest volume estimation and yield prediction. FAO Forestry Paper 22/2. Food and Agriculture Organization of the United Nations, Rome.
- Anderson, A. B., A. Gely, J. Strudwick, G. L. Sobel & M. G. C. Pinto.** 1985. Um sistema agroforestal na várzea do estuário amazônica (Ilha das Onças, Município de Barcarena, Estado do Pará). *Acta Amazonica Supplement* 15: 195–224.
- Ash, J.** 1987. Demography of *Cyathea hornei* (Cyathaceae), a tropical tree fern from Fiji. *Australian Journal of Botany* 35: 331–342.
- Avery, T. E. & H. E. Burkhardt.** 1983. Forest measurements. McGraw-Hill, New York.
- Balée, W. L.** 1994. Footprints of the forest: Ka'apor ethnobotany—The historical ecology of plant utilization by an Amazonian people. Columbia University Press, New York.
- Barrera de Jorgenson, A.** 1993. Chicle extraction and forest conservation in Quintana Roo, Mexico. Thesis. University of Florida, Gainesville.
- Bonham, C. D.** 1989. Measurements for terrestrial vegetation. John Wiley, New York.
- Boom, B. M.** 1989. Use of plant resources by the Chacobo. *Advances in Economic Botany* 7: 78–96.
- Bormann, F. H.** 1953. The statistical efficiency of sample plot size and shape in forest ecology. *Ecology* 34: 474–487.
- & **T. T. Kozłowski.** 1962. Measurement of tree growth with dial gauge dendrometers and vernier tree-ring bands. *Ecology* 43: 289–294.

- Bullock, S.** 1980. Demography of an undergrowth palm in littoral Cameroon. *Biotropica* 12: 247–255.
- Campbell, D. G., D. C. Daley, G. T. Prance & U. N. Maciel.** 1986. Quantitative ecological inventory of terra firme and várzea tropical forest on the Rio Xingu, Brazilian Amazon. *Brittonia* 38: 369–393.
- Chapman, C. A., L. J. Chapman, R. Wingham, K. Hunt, D. Gebo & L. Garder.** 1992. Estimators of fruit abundance of tropical trees. *Biotropica* 24: 527–531.
- Clark, D. A. & D. B. Clark.** 1987. Temporal and environmental patterns of reproduction in *Zamia skinneri*, a tropical rain forest cycad. *Journal of Ecology* 75: 135–149.
- Cochran, W. G.** 1977. Sampling techniques. 2nd ed. John Wiley, New York.
- Dinerstein, E.** 1986. Reproductive ecology of fruit bats and the seasonality of fruit production in a Costa Rican cloud forest. *Biotropica* 18: 37–318.
- Dove, M. R.** 1993. Smallholder rubber and swidden agriculture in Borneo: A sustainable adaptation to the ecology and economy of the tropical forest. *Economic Botany* 47: 136–147.
- Dransfield, J. & N. Manokaran (eds.).** 1994. Rattans. Plant Resources of South-East Asia No. 6, Plant Resources of South East Asia Foundation, Bogor, Indonesia.
- Food and Agriculture Organization (FAO).** 1973. Manual of forest inventory with special reference to mixed tropical forest. Food and Agriculture Organization of the United Nations, Rome.
- Fong, F. W.** 1992. Perspectives for sustainable utilization and management of Nipa vegetation. *Economic Botany* 46: 45–54.
- Gauch, H. G.** 1982. Multivariate analysis in community ecology. Cambridge University Press, Cambridge, U.K.
- Gentry, A. H.** 1982. Patterns of neotropical plant species diversity. *Evolutionary Biology* 15: 1–84.
- . 1988. Tree species richness of upper Amazonian forests. *Proceedings of the National Academy of Sciences* 85: 156–159.
- (ed.). 1990. Four neotropical forests. Yale University Press, New Haven, Conn.
- Gianno, R.** 1986. The exploitation of resinous products in a lowland Malayan forest. *Wallaceana* 43: 3–6.
- Goldsmith, F. B. & C. M. Harrison.** 1976. Description and analysis of vegetation. Pages 85–155 in S. B. Chapman, ed., *Methods in plant ecology*. John Wiley, New York.
- Green, D. F. & E. A. Johnson.** 1994. Estimating the mean annual seed production of trees. *Ecology* 75: 642–647.
- Greig-Smith, P.** 1983. Quantitative plant ecology. 3rd ed. University of California Press, Los Angeles.
- Hall, P. & K. Bawa.** 1993. Methods to assess the impact of extraction of non-timber tropical forest products on plant populations. *Economic Botany* 47: 234–247.
- Harshberger, J. W.** 1896. Purposes of ethnobotany. *Botanical Gazette* 21: 146–154.

- Heinsdijk, D. & M. A. de Bastos.** 1965. Forest inventory on the Amazon. FAO Report No. 2080. Food and Agriculture Organization of the United Nations, Rome.
- Hodge, W. H.** 1948. Wartime *Cinchona* procurement in Latin America. *Economic Botany* 2: 229–257.
- Holdridge, L. R., W. C. Grenke, W. H. Hatheway, T. Liiaing & J. S. Tosi.** 1971. Forest environments in tropical life zones: A pilot study. Pergamon Press, Oxford.
- Howe, H. F.** 1977. Bird activity and seed dispersal of a tropical wet forest tree. *Ecology* 58: 539–550.
- . 1980. Monkey dispersal and waste of a neotropical tree. *Ecology* 61: 944–959.
- & **G. A. Vande Kerckhove.** 1981. Removal of wild nutmeg (*Virola surinamensis*) crop by birds. *Ecology* 62: 1093–1106.
- Hubbell, S. P.** 1979. Tree dispersion, abundance and diversity in tropical dry forest. *Science* 203: 1299–1309.
- Husch, B., C. I. Miller & T. W. Beers.** 1972. Forest mensuration. Ronald Press, New York.
- Irvine, D.** 1989. Succession management and resource distribution in an Amazonian rain forest. *Advances in Economic Botany* 7: 223–237.
- Kershaw, K. A. & J. H. H. Looney.** 1985. Quantitative and dynamic plant ecology. 3rd ed. Edward Arnold, London.
- Kinnard, M. F.** 1992. Competition for a forest palm: Use of *Phoenix reclinata* J. J. Jacquin by human and non-human primates. *Conservation Biology* 6: 101–107.
- Knight, D. H.** 1975. A phytosociological analysis of species-rich tropical forest on Barro Colorado Island, Panama. *Ecological Monographs* 45: 258–254.
- Köhl, M.** 1993. Forest inventory. Pages 243–332 in L. Pancel, ed., *Tropical forestry handbook*. Springer-Verlag, Berlin.
- Kozlowski, T. T., P. J. Kramer & S. G. Pallardy.** 1991. The physiological ecology of woody plants. Academic Press, New York.
- Lang, G. E., D. H. Knight & D. A. Anderson.** 1971. Sampling the density of tree species with quadrats in a species-rich tropical forest. *Forest Science* 17: 395–400.
- Lepofsky, D.** 1992. Arboriculture in the Mussau Islands, Bismark Archipelago. *Economic Botany* 46: 192–211.
- Liming, F. C.** 1957. Homemade dendrometers. *Journal of Forestry* 55: 575–577.
- Lugo, A. & C. Rivera.** 1987. Leaf production, growth rate, and age of the palm *Prestoea montana* in the Luquillo experimental forest, Puerto Rico. *Journal of Tropical Ecology* 3: 1151–1161.
- Lyon, L. J.** 1968. An evaluation of density sampling methods in a shrub community. *Journal of Range Management* 21: 16–20.
- Mueller-Dombois, D. & H. Ellenberg.** 1974. Aims and methods of vegetation ecology. John Wiley, New York.
- Myers, E. & V. J. Chapman.** 1953. Statistical analyses applied to a vegetation type in New Zealand. *Ecology* 34: 175–185.

- Oyama, K.** 1990. Variation in growth and reproduction in the neotropical dioecious palm *Chamaedorea tepejilote*. *Journal of Ecology* **78**: 648–663.
- Padoch, C. & W. de Jong.** 1991. The house gardens of Santa Rosa: Diversity and variability in an Amazonian agricultural system. *Economic Botany* **45**: 1166–1175.
- & **C. Peters.** 1993. Managed forest gardens in West Kalimantan, Indonesia. Pages 167–176 in C. S. Potter, J. I. Cohen & D. Janczewski, eds., *Perspectives on biodiversity: Case studies of genetic resource conservation and development*. AAAS Press, Washington, D.C.
- Peters, C. M.** 1990. Plant demography and the management of tropical forest resources: A case study of *Brosimum alicastrum* in Mexico. Pages 265–272 in A. Gomez-Pompa, T. C. Whitmore & M. Hadley, eds., *Rain forest regeneration and management*. Cambridge University Press, Cambridge.
- . 1994. Sustainable harvest of non-timber plant resources in tropical moist forest: An ecological primer. Biodiversity Support Program, Washington, D.C.
- & **E. J. Hammond.** 1990. Fruits from the flooded forests of Peruvian Amazonia: Yield estimates for natural populations of three promising species. *Advances in Economic Botany* **8**: 159–176.
- & **A. Vazquez.** 1987. Estudios ecologicos de camu-camu (*Myrciaria dubia*) I. Produccion de frutos en poblaciones naturales. *Acta Amazonica* **16/17**: 161–174.
- Philip, M. S.** 1994. *Measuring trees and forests*. 2nd ed. CAB International, Wallingford, U.K.
- Phillips, O.** 1993. The potential for harvesting fruits in tropical rainforests: New data from Amazonian Peru. *Biodiversity and Conservation* **2**: 18–38.
- Pinard, M.** 1993. Impact of stem harvesting on populations of *Iriartea deltoidea* (Palmae) in an extractive reserve in Acre, Brazil. *Biotropica* **25**: 2–14.
- Pinedo-Vasquez, M., D. Zarin, P. Jipp & J. Chota-Iuma.** 1990. Use-values of tree species in a communal forest reserve in northeast Peru. *Conservation Biology* **4**: 405–416.
- Piñero, D. & J. Sarukhan.** 1982. Reproductive behavior and its individual variability in a tropical palm, *Astrocaryum mexicanum*. *Journal of Ecology* **72**: 977–991.
- , **J. Sarukhan & E. Gonzales.** 1977. Estudios demográficos en plantas. *Astrocaryum mexicanum* Liebm. I. Estructura de la poblaciones. *Boletín de la Sociedad Botánica de México* **37**: 69–118.
- Prance, G. T., W. Balée, B. M. Boom & R. L. Carneiro.** 1987. Quantitative ethnobotany and the case for conservation in Amazonia. *Conservation Biology* **1**: 296–310.
- Reid, N., J. Marroquin & P. Beyer-Munzel.** 1990. Utilization of shrubs and trees for browse, fuelwood and timber in the Tamaulipan thornscrub, northeastern Mexico. *Forest Ecology and Management* **36**: 61–79.
- Rico-Gray, V., J. G. Garcia-Franco, A. Chemas, A. Puch & P. Sima.** 1990. Species composition, similarity, and structure of Maya homegardens in Tixpeual and Tixcacaltuyub, Yucatan, Mexico. *Economic Botany* **44**: 470–487.