Before initiating a survey or a monitoring program of any group of organisms in an area, an investigator should carry out some preliminary background research. Essential materials for the research include maps of the area and descriptions of its climate, geology, and vegetation. Learning to recognize the woody plant species and major plant associations likely to be encountered is particularly important. Knowledge of the anticipated species diversity and distributions of the macrofungi is also very helpful. Such background information increases an investigator’s understanding of the habitat requirements of the fungi encountered, aids in delimiting plots that accurately represent the habitats present, and helps in the design of an effective sampling regimen. Color photographs of the vegetation and landscape also are useful.

In any study, the investigator must choose where, when, and how to collect data. Field studies are particularly sensitive to timing and location of observations. Macrofungi exhibit patterns of diversity that are related largely to substratum and host availability, and their fruiting (and, hence, our opportunity to observe them) is climate driven. The productive seasons for sampling macrofungi and the frequency and duration of sampling are dictated largely by the local climate. Fungi fruit when temperatures are above freezing and moisture is available. Different species, however, exhibit different fruiting phenologies, which vary from year to year and at different elevations and latitudes. Maximum richness of fruiting species occurs only during brief periods and differs among years. Environmental variables and ecological processes that affect the likelihood of recording a species during a study must be considered irrespective of the study’s goals.

### Seasonality and Year-to-Year Annual Variation

Within a geographic region fruiting is influenced by elevation and latitude and their effects on temperature and precipitation (Ohenoja 1993). Thus, a particular species may fruit at different seasons across wide geographic distances or along strong elevational gradients. In addition to different seasonal peaks of abundance for individual species, annual variation in presence of sporocarps can be enormous. For example, only 5% to 20% of the ectomycorrhizal species encountered at each of eight sites in Olympic National Park fruited in 2 successive years (Fig. 8.1). Combining the data from all sites, about 30% of the species were detected in both years (O’Dell and Ammirati 1994; O’Dell et al. 1999). Similarly, Winterhoff (cited in Arnolds 1992) studied five sites for 5 years and found that from 8% to 88% of the total reported species from a single site were detected in any given year. Lodge (1996) found that several species in the Entolomataceae fruited every second or third year in a wet subtropical forest in Puerto Rico, whereas a few other species were found only during 1 year of a 13-year survey. Schmit et al. (1999) reported a significant annual difference in species composition during a 3-year study of macrofungal diversity in oak-dominated forests of the Chicago region. In a 21-year study of fungal fruiting
Collecting and Describing Macrofungi

**FIGURE 8.1** This cumulative species-accumulation graph shows the increase in the number of recorded species with additional samples over time (Data from T. O’Dell). If the community is relatively homogeneous, the curve will level off (reach an asymptote) more quickly than if several communities are mixed together. The asymptote indicates the species richness for the area. For macrofungi, up to 8–12 years of sampling may be required to approach an asymptote. In general, methods used to estimate the total species richness for an area will not work well if no ‘shoulder’ is present in the species accumulation curve.

Phenology in Switzerland, Straatsma and colleagues (2001) found that some species fruited only during the 1 year that was particularly rich in species and that fruiting abundance and species richness were correlated. Species richness estimators did not stabilize during the 21 years.

The challenge of measuring species diversity of macrofungi increases in years when many species fruit simultaneously at a given site. Unless a large, efficient workforce is available, specimens may decay before they can be adequately documented, resulting in significant loss of data. That situation, together with the fact that some species may fruit only 1 year out of 4 or more, underscores the need for long-term (at least 5-year, preferably 10-year) studies. The time scale of fungal succession is not well known, but the first observations of some species on a site may reflect their recent migration into the community. Annual variation in species recorded because of variable fruiting patterns is difficult to distinguish from succession.

In temperate regions with summer drought, spring and fall are the main fruiting seasons. The “fall” season is progressively later at lower latitudes, in some regions occurring after the winter solstice. In temperate regions with summer rain and at high elevations and latitudes, summer may be the most important season for fruiting. Unusual weather events can trigger unusual patterns of fruiting, however; in regions with dry summers, for example, fruiting of species rarely observed in the region may follow a summer shower. Although the fruiting of macrofungi most often is limited by lack of precipitation, excess moisture also can prevent fruiting in some species.

Temperature also has a major impact on macrofungi fruiting, an effect that may not be limited to the fruiting season. Ohenoja (1993) documented the effects of year-round temperatures on fall fruiting of macrofungi. She found that temperature interacted with the habitat and ecological guild of the fungus in stimulating or retarding fruiting. For example, warm summers increased the production of mycorrhizal sporocarps in spruce forests but had no effect on sporocarp production in pine forests.

If a measure of species richness is the goal of a study, then baseline data for a site may involve intense collecting over a period of several days when sporocarp production is high. Ideally, however, one samples fleshy macrofungi in an area every 1–2 weeks throughout the fruiting season to maximize the number of species observed. When such a schedule is not possible, it is useful to do a pilot study (see discussion of complementarity...
tests under “Determining Adequate Sampling,” later in this chapter) before deciding when and how often to sample. Studies of annual variation and seasonality of macrofungal fruiting have been limited largely to Europe. Few studies have examined multiple taxa or lasted long enough to have predictive value. Some data on seasonality, however, may be gleaned from herbarium records.

**Vegetation**

The type of vegetation in an area affects the species richness and composition of macrofungi at that site. Grasslands, deserts, forests, tundra, and other habitats all have characteristic species. Plant species composition also influences the number and species of macrofungi present because plants constitute the habitat and energy source for most fungi, and all fungi show some degree of host or substratum specificity.

Vegetation zones or plant associations are useful criteria to use when dividing a landscape for sampling. Many fungi occur only in association with particular families or genera of plants. Villeneuve and colleagues (1989), Bills and associates (1986), and Nantel and Neuman (1992) found the distribution of ectomycorrhizal fungus species to be correlated with forest type (forests dominated by deciduous trees versus conifers). O’Dell and others (1999) found precipitation to be better than vegetation type as a predictor of species richness and community structure of ectomycorrhizal fungi at local scales. The large variation found among sites in a single forest zone in their study points to the need for sampling multiple sites with similar host tree species (Figs. 8.2 to 8.5).

**Geography**

Mycologists with broad geographic experience were surveyed to determine what factors they believed most strongly affect diversity in the groups that they study (Lodge et al. 1995). For the fungi treated in this chapter, diversity of habitats rather than geography was believed to have the strongest influence on fungal species richness. In much of Europe, tree diversity is low as a result of local extinctions of species caused by Pleistocene glaciation, and it is probably not coincidental that Europe was ranked lowest in fungal diversity when compared with other regions, although it has been collected more intensively (Dennis 1986). Some ectomycorrhizal fungi (e.g., Cortinariaceae) and discomycetes were thought to be most diverse at middle and high latitudes, whereas Agaricales in general (especially saprobic Tricholomataceae) were considered most diverse at low latitudes.

![FIGURE 8.2 Ectomycorrhizal fungi include those with fleshy indented caps and thick wrinkles instead of gills, such as *Gomphus* (shown here) and species of *Cantharellus* (Chantarelles). (Photo by T. O’Dell)
FIGURE 8.3 Ectomycorrhizal fungi include all species of Lactarius (milkcaps), including *L. indigo* shown here, and species of the related genus, *Russula*. Latex presence, color and color changes, taste, odor, and spore-print color, and color reactions of pileus, lamellae, and stipe to FeSO₄, should be recorded to identify species in this family, Russulaceae. (Photo by G. Mueller)

FIGURE 8.4 Ectomycorrhizal fungi include some of the typical gilled agaric fungi, such as this *Laccaria laccata*. (Photo by G. Mueller)
FIGURE 8.5 Most species of boletes, such as this Suillus spraguei, form ectomycorrhizal associations with tree roots. Boletes can be recognized by their texture, tubes that peel easily from the cap flesh, and their typical habit of fruiting on the ground. (Photo by G. Mueller)

**SUCCESSION**

Succession of macrofungi must be considered from several perspectives. First, there are successions of sporocarp production on particular substrata, although all species may be present in the substrata from the beginning. Herbivore dung, for example, has characteristic fungi that fruit consecutively over time (Chapter 21). Succession involving changes in community composition often are related to changes in the quality of the substratum. Hedger (1985) found, for example, that some species of *Lepiota* only grow well on leaf litter that previously has been decomposed by other fungi, such as certain *Marasmius* species. Similarly, trunks of large fallen trees host a cadre of fungi that fruit early in log decomposition and others that fruit only later (Lange 1992; Renvall 1995; Heilmann-Clausen 2001). One approach for determining changes in fungal community composition over long periods is to study a chronosequence. To document changes in the composition of the macrofungal community on decomposing beech logs, Heilmann-Clausen (2001) studied a 30-year record of aerial photographs of a Danish forest to determine when each tree more than 60 cm dbh had died. He used that information to divide the 130 logs into seven age cohorts and determined the macrofungi present on the logs in each one.

Secondly, successional changes occur in the vegetation at a site. Such changes may have a direct impact on fungi through the establishment: of new host taxa and changes in the amount and quality of available organic matter. Fungi are also likely to be affected by changes in understory microclimate. Lodge and Cantrell (1995b) found that the opening of the forest canopy in the Caribbean National Forest in Puerto Rico (El Verde field station) by hurricane Hugo caused the extirpation of several mycelia of *Collybia johnstonii*, especially on ridges where the canopy has been slowest to close. That leaf-decomposing fungus has superficial mycelia and hyphal strands that are very sensitive to drying.

Apparently, ectomycorrhizal fungi on host root systems also exhibit succession. Some species fruit in very young stands of trees, some are restricted to older stands, and others occur across many age classes (Mason et al. 1983; Dighton et al. 1986; Luoma et al. 1991; Deacon and Fleming 1992; Last et al. 1992; O’Dell et al. 1992a, 1992b). Ectomycorrhizal succession is poorly documented, having been studied largely in plantations of trees outside of their native ranges. Successional phenomena illustrate the need for including diverse microhabitats and a range of stand ages among the sampling sites when studying the macrofungal diversity of a landscape.

**COLLECTION, CULTURE, AND PRESERVATION**

After sampling design, the core of any fungal diversity study is the collection and documentation of specimens. That includes collecting sporocarps at the field site,
Collecting and Describing Macrofungi

labeling them, taking photographs, taking tissue cultures, setting up spore prints, writing descriptions, and preserving the specimens for later identification. Adequate time must be budgeted for all of those activities. One approach is to spend part of the day in the field and the remainder at the laboratory or field station working up specimens. Alternatively, if field sites are remote and collecting is good, 2 or 3 full days of collecting can be followed by up to a week of laboratory work. In that case, the specimens must be prioritized in order of preparation, with the most fragile and important specimens processed first and the remaining specimens stored in as cool a place as possible.

Identification of macrofungi relies heavily on the attributes of fresh specimens, and it is generally better to prioritize documenting these attributes over identifying specimens to species. If time permits, identifying a specimen is beneficial but not at the expense of documenting its morphological characters or letting other specimens spoil.

COLLECTING

Data management starts before collecting begins because specimens should be labeled as they are collected. Investigators must decide in advance which ecological or habitat data to record, prepare field labels, choose a cataloging system, and so on.

Not every specimen encountered needs to be collected. Deciding whether to collect a particular mushroom is not a trivial problem and depends on the condition and quality of the specimen as well as the goals of the study. General collecting in an area for inventory purposes or for a generic monograph allows one to be selective among specimens, although representatives of all species encountered in a plot-based study should be collected. Larger amounts of material are required for biomass or chemical studies.

For many taxa it is important to have data on all stages of sporocarp development. Thus, collectors should obtain sporocarps exhibiting a range of developmental stages for each taxon. Mature sporocarps for some groups, such as some Pezizales, may appear decrepit. Often a careful search of an area of several meters radius around the initial collection site will yield a full range of developmental stages. In general, however, it is usually best to leave behind sporocarps that are too immature or too old to provide useful data (Figs. 8.6 to 8.8).

Making a collection requires care and patience. Specimens are removed from the substratum by excavating around the base of the stipe to reveal any volva, rooting base, bulb, or attachment to a sclerotium or buried substrata. Such substrata include wood, cones or fruits, other fungi, and animals. If the specimen is on wood or litter, including some of the substratum with the collection will facilitate its identification. Unidentified plants near the specimen also can be included with the

FIGURE 8.6 It is best to collect all of the developmental stages of macrofungi to identify them and to document morphological variation; that is true for the species of *Crucibulum* shown here. (Photo by G. Mueller)
FIGURE 8.7 Fruit bodies in all stages of development are required for confident identification of almost all species of puffballs and earthballs, including the *Geastrum* species shown here. If only immature specimens are collected, it is sometimes possible to observe the color changes through maturation of the spores in the interior (the gleba) for several days after they have been collected. (Photo by D. J. Lodge)

FIGURE 8.8 Some agaric fungi, such as the *Coprinellus (Coprinus) mexicanus* shown here, change rapidly as they mature, so all stages should be collected. Most species of *Copinus* (in the broad sense) digest themselves as the spores mature thus destroying the structures on the cap surface and the gill edges that are critical for identification. (Photo by D. J. Lodge)
collection for later identification. After the site is examined carefully, it can be described as wet, boggy, dry, or whatever is appropriate. Collectors also should note if digging into the substratum was required to remove the sporocarp, if rhizomorphs were associated with it, or if it was attached to a moss. Those observations should be recorded on field labels, although if a specimen is collected properly, the substratum and/or rhizomorphs still should be apparent when the specimen is examined during processing (Fig. 8.9).

Sporocarp color is often important in the identification of macrofungi. Collectors should note color changes caused by bruising or handling of specimens. In many agaric genera, loss of moisture from the pileus occurs rapidly after collecting (such specimens are termed hygrophanous), so it is helpful to note the general color in the field. For example, color changes in basidiomes of some Cortinarius, Ramaria, and Mycena species can be rapid and, if undetected, can lead to incorrect descriptions and identifications. Small or fragile basidiomes of species, such as some Lepiota and Psathyrella, may have delicate veil material that can be damaged or lost on handling. Such features should be noted in the field.

**EQUIPMENT**

Every mycologist has his or her preferred collecting paraphernalia, and to a degree preferences depend on the taxa being collected. At least four items are required for collecting macrofungi: (1) a tool for cutting and digging, (2) a container or wrapping material for each specimen, (3) a larger container for transporting specimens in the field and back to the lab, and (4) a label for each specimen.

A thick-bladed, moderately sharp knife can be used to cut woody substrata or dig in soil. Some collectors carry both a knife and a trowel for collecting sporocarps from soil. Different types of fungi occurring on wood require different types of collecting equipment. An ax or hatchet often is needed to extract wood to a depth sufficient to enable identification of the host if it is unknown. However, a mallet and wood chisel, a heavy sheath knife,
FIGURE 8.10 A sheath knife or a folding knife with a locking blade is one of the most useful pieces of collecting equipment for macrofungi. Fungi fruiting on the ground, such as the species of *Macrocybe* shown here, should be pried up from the base to ensure that none of the basal parts are lost. (Photo in Costa Rica by G. Mueller)

or a folding knife with a locking blade are usually sufficient for removing the fungus. A pair of pruning shears and a folding pruning saw are also helpful for cutting smaller diameter twigs and branches to a uniform length. Care must be used to avoid undue damage to the plant if collecting from a living tree (Figs. 8.10 and 8.11).

The type of container or wrapping material used for individual specimens depends in part on the type of fungi being collected. Aluminum foil, waxed paper bags, or sheets of waxed paper rolled into packets and twisted in opposite directions at the ends often are used for fleshy fungi. Fragile specimens are best wrapped in aluminum foil or placed in small boxes. Foil keeps the specimens from dehydrating, affords good protection, remains intact when wet, and easily can be reused. Small plastic boxes, vials, fishing tackle or toolboxes, or other containers also work well (Fig. 8.12). Boxes prevent fungi from being crushed or bruised, keep them moist, afford protection from rain, and allow for easy viewing and comparison of specimens. Paper bags of various sizes work well for “woody” basidiomycetes (e.g., bracket fungi) and small, durable ascomycetes.

 Appropriately wrapped specimens are placed in a basket, bucket, pack basket, backpack, or bag (plastic or otherwise) chosen to protect the specimens from being crushed and for ease of carrying in the field (Fig. 8.13). Basidiomes of *Amanita* and certain other fungi will reorient their pilei after collection (the pileus is positively
FIGURE 8.11 Macrofungi that fruit on wood, such as this tooth fungus in the genus *Hericium*, can present a challenge for collectors. If the wood is soft, a sheath or buck knife or a knife with a locking blade can be used to remove the specimen with part of the substratum. Some collectors prefer to use a hatchet or a chisel and rubber mallet to remove specimens from wood. (Photo by G. Mueller)

FIGURE 8.12 Plastic boxes are useful for holding fungi, especially small and medium-size macrofungi. Collecting boxes of the type used for fishing tackle or hardware supplies allow for rapid viewing of the specimens and also protect them from being crushed. Placing leaves or pieces of moss or lichens in with the specimens will prevent them from rolling and becoming bruised and will also help small specimens to retain moisture. (Photo by D. J. Lodge)
FIGURE 8.13 Many collectors, especially those working with large fleshy agarics and boletes, wrap their specimens in waxed paper or aluminum foil and carry them in a basket. Both waxed paper and foil provide physical support and help prevent moisture loss without suffocating the fruit bodies. (Photo by D. J. Lodge)

geotropic) and should be carried standing straight up to prevent the twisting and distortion of their stipes.

Fleshy fungi dehydrate and decompose very quickly. Specimens transported in a cooler often hold up better and should remain in the cooler or a refrigerator until they are described and cultured. Small coolers with an ice pack that can be frozen and attached to the lid work well for small specimens, especially in tropical and subtropical climates (Fig. 8.14). If a collector working in a hot or sunny site places the ice pack below rather than above the specimens, a strong temperature gradient develops inside the cooler. That gradient causes the specimens in the upper part of the container to desiccate; condensation that forms in the lower part accelerates decomposition of specimens there. Securing very small specimens inside their containers with a fresh, green leaf
or moss can reduce desiccation and physical damage. Larger specimens are less likely to desiccate, and they can be placed on the bottom of a large cooler lined with frozen ice packs. Separating the ice packs and the specimens with a layer of paper bags or newspaper keeps specimens from freezing.

**DOCUMENTATION**

Fleshy fungi are particularly difficult to work with because sporocarps dry or decompose readily; the characteristics needed for identification are often ephemeral; and many macromorphological features such as size, shape, and color are lost with preservation. Specimen documentation includes obtaining cultures, data on macrochemical tests, photographs, spore prints, and written descriptions. Not all taxa or specimens require equal documentation. When fruiting is at its peak, one must maximize field time. Specimens that do require documentation—that is, those that are fragile, decay rapidly, or otherwise require immediate attention—must be processed quickly. It is, therefore, necessary to organize specimens before working through them. Specimens are put into priority order by taxon; within each taxon they are processed in order according to condition of the specimen (best to poorest), which provides the most accurate and efficient documentation of diversity.

Collectors of biological materials have long recognized the need to catalogue their specimens. Typically, that
includes a label for each specimen and a catalogue that contains the data for all collections. In addition, some biologists keep a field journal wherein they record their travel routes, daily activities, and observations. A field journal can be valuable in relocating a collection site or verifying a location or other information when a question arises later on. General methods and justification for keeping a field journal are described by Herman (1986). Whereas keeping a field journal is an elective activity, consistent labeling and cataloging of collections are mandatory.

Field labels are the first step in tracking collection data. The data should be recorded in pencil or permanent black ink (e.g., India ink) on waterproof paper at the time each collection is made. Minimum data for each label are collection number (if only an interim one), collector’s name, date, and location. One also can include a tentative identification; a plot number; and notes on the associated vegetation, substratum, microhabitat, and ephemeral characters of the sporocarp.

Preprinting labels ensures that important data are not omitted, saves time while collecting, and facilitates efficient recording of ecological and host data. Some collectors use prenumbered labels; this has advantages and disadvantages. If a specimen is discarded, the collection number goes with it, so one must keep track of “discarded” numbers. An alternative to prenumbering the field labels is to use labels that are numbered sequentially beginning at one on each date and later assigning “final” collection numbers to specimens. If that method is used, a separate entry of the daily collection numbers should be included in the catalogue to facilitate tracking of specimens when they are sorted or identified.

A catalogue is a list of collections in numerical order that includes the date, collector, and location. It also can contain site and weather data, photographic data, and other relevant information. Every collector should maintain a catalogue with a unique number for each specimen. Various numbering systems can be used to construct a collection number; the critical factor is that one be consistent in the system used. Some mycologists’ collection numbering systems incorporate the collector’s last name or initials and a unique number (e.g., O’Dell 2401, JLL 1014, or JLL 1995–17); other systems include an institutional acronym or an abbreviation for the country of origin followed by a unique number. Because collection numbers constitute a historical record that others may use someday, the consistent use of a chosen numbering system is imperative.

**Preservation**

Maintaining voucher specimens is a critical aspect of measuring or monitoring diversity (Ammirati 1979). Most agarics and other fleshy fungi require a heat source for drying. For most polypores, some Corticiaceae, and some ascomycetes, air-drying is preferable because the fungus is not killed but merely goes dormant. The specimens subsequently can be used for isolating cultures. To facilitate drying, the specimens should include minimal adhering wood, especially if the wood is particularly wet. Submerging small specimens in repeated changes of silica gel until the sporocarps are dry is also an option, but specimens should not come into direct contact with the silica crystals because adhering crystals can cause problems when microscopic mounts are made.

Dryers with wire screen shelves suspended on a rack over a heat source of 38–55°C are most effective. The source of the heat is not critical; incandescent light bulbs, hot plates, and space heaters all can be used, but a fan to increase air movement is helpful. Commercially made food dehydrators are popular because they are self-contained, reasonably compact, complete units, with no assembly required. An alcohol burner with something to disperse the heat (e.g., aluminum foil or a metal steamer basket) placed between the flame and the specimens is effective and will burn for 8–12 hours without refilling. Small kerosene stoves or white gas catalytic heaters also can be used in the absence of electricity. The advantage of the latter is the lack of flame. Specimens should not be placed too close to any heat source; they can char or catch fire easily.

Large agarics and boletes should be split in half or quartered from top to bottom before being placed in a dryer. That practice speeds drying, prevents the context (the interior of the sporocarp) from decaying, and stops the feeding activities of insect larvae. If drying racks are stacked over the heat source, the largest fleshy fungi should be placed on the lower shelves, and the smaller fungi should be placed on the upper shelves. When many collections of small specimens are placed together on the same drying tray the risk of losing part of some collections or incorrectly matching the specimens to the labels is high. Very small specimens can be placed inside paper packets or small, thin, cloth bags before drying.

**Fixing Material for DNA Analysis**

Subsamples of material collected as part of an inventory can serve as sources of DNA for laboratory-based studies and should be preserved accordingly. Pieces of sporocarp are excised carefully and submerged in a fixative solution in cryovials or microcentrifuge tubes. Several fixative solutions prevent degradation of sporocarp DNA during long-term storage. The two most commonly used solutions are 2X CTAB buffer and a supersaturated solution of DMSO (Wu et al. 2000; Appendix II).
Specimen Storage

Temporary storage of preserved macrofungi can be difficult, especially when the ambient relative humidity is high, because dried specimens are hygroscopic. Self-closing (e.g., zipper), airtight, transparent bags provide excellent temporary storage for dried macrofungi. The bags come in different sizes and accommodate sporecarps of various sizes. Wax paper and plastic sandwich bags are less expensive alternatives. Bags protect the specimens from moisture and insects while allowing for visual inspection or sorting of specimens; spore deposits also may be included. Specimens to be shipped in the mail first should be wrapped in blank newsprint and then put in a plastic bag. The paper will afford the specimens more protection. A label always is included with each specimen.

In tropical regions special care must be taken to prevent rehydration of preserved specimens. Filling and closing the plastic bags over a drier will prevent moisture from entering the bags. Cracker and cookie or biscuit tins are airtight, crush-proof containers suitable for transporting specimens in a completely dry state. When air is extremely humid, tins should be packed and sealed over a heat source, or the packed tins should be exposed to extremely humid, tins should be packed and sealed over a heat source, or the packed tins should be exposed to heat for 30 minutes before the lids are sealed with duct tape. Cloth or paper packets of silica gel, preferably mixed with indicator dye, also can be dried and packed with specimens.

For long-term storage, specimens should be maintained in herbarium boxes or packets according to the methods outlined in Chapter 2 (see “Packeting, Boxing, and Mounting Specimens”).

CULTURING MACROFUNGI

Cultures are sometimes helpful in distinguishing similar taxa and identifying them to species. Cultures are especially important for some groups of ascomycetes (e.g., Xylariaceae; Hypocreaceae; other pyrenomycetes, such as Chaetosphaeria) in which the anamorph (asexual stage) is required for accurate species identification. Because obtaining, maintaining, and studying fungal cultures is time consuming and requires specialized materials and training, such preparations will be beyond the scope of many biodiversity studies. In those cases, only generic identifications will be possible for some ascomycetes.

Terrestrial macrofungi have been cultivated from tissue or from germinated basidiospores or ascospores with varying success. Saprobic fungi generally are cultured more easily than ectomycorrhizal species, but exceptions exist. For example, certain Lepiota have been grown on leaf litter that has been decomposed by Marasmius and then autoclaved (Hedger 1985). Tissue cultures of ectomycorrhizal fungi such as boletes, Laccaria, Tricholoma, Hebeloma, and Amanita, can be obtained using the proper procedure and media; other genera, such as Lactarius, Russula, Entoloma, Cortinarius, and Hygrophorus are difficult or impossible to grow. For general information on the cultivation of macrofungi, see Wading (1980) or Stamets and Chilton (1983).

Many wood-inhabiting fungi grow well on malt extract agar, but potato dextrose agar and oatmeal agar also are used in some laboratories (Appendix II). Special agar containing organic matter from a particular substratum can be used to grow some fungi difficult to culture on standard media; some wood-inhabiting basidiomycetes, for example, will fruit on wood-containing media with sawdust used as a base (Etter 1929). Special media also have been tried with varying success with some hard-to-germinate wood-inhabiting ascomycetes (S. M. Huhndorf, personal communication) and basidiomycetes such as species of Laetiporus, H. H. Burdsall, personal communication) and Pluteus (O. K. Miller, personal communication).

Culturing from Basidiospores and Ascospores

The most common way to obtain macrofungi in culture is to germinate their spores. An easy method for obtaining spore cultures is to let a fungus drop or shoot spores directly onto an agar surface. Ascomata or portions of the hymenial surface of basidiomycetes are suspended near the edge on the lid of a Petri plate of agar (petroleum jelly, small chips of agar, or water drops can be used to attach the fungus to the lid). The wrapped plate can be tilted at a steep angle with the hymenium of the fungus located at the upper end (gills of agarics or pores of polyporaceae are aligned vertically in the direction the plate is to be tilted). Such positioning produces a cascade with spore densities high at the upper end and decreasing toward the lower end and along the side. Both polyspore and single-spore isolates can be obtained from the same spore print. The source specimen should be removed as soon as spores are detected. Alternatively, the lid of a horizontal Petri dish can be rotated periodically (e.g., every 30 seconds to 10 minutes, depending on the specimen) to obtain distantly spaced spores for single-spore isolates. Individual spores can be identified and then removed using a fine sterilized needle while under a dissecting microscope. For single-spore isolations, only germinated spores that are well separated from other germinated and ungerminated spores should be transferred to individual culture plates. The microscope should be equipped with an adjustable mirror below the stage for the light source. The mirror should be adjusted so that the spores appear bright against a dark background; spore deposits and germination can be observed by
turning a plate upside down and focusing through the agar. Usually 8–12 hours (sometimes much less, especially with dark spores) is sufficient time for a spore print to be observable, especially when using a dissecting microscope. For tropical fungi, the hymenium or ascomata should be removed within 12 to 24 hours. Additional incubation will not result in additional spore production but likely will lead to contamination of the plates with spores from opportunistic fungi such as *Fusarium* and *Trichoderma*. In other cases (e.g., temperate and boreal fungi) several days to a week may be required to obtain a spore drop culture. Changes of barometric pressure that are associated with changes in elevation of more than several hundred meters are probably responsible for the failure of some tropical basidiomycetes to drop spores (Dennis 1970; D. J. Lodge, personal observation; R Singer, personal communication); it may be necessary, therefore, to set up spore drops in the field if the investigator’s laboratory is located at a very different elevation. In addition, tropical basidiomycetes that were placed in a refrigerator for more than an hour before culturing failed to drop spores, and their cultures often died when held at 5–10°C (D. J. Lodge, personal observation). The elevational effect has not been observed among temperate species (H. H. Burdsall, personal observation), nor has the cold-temperature effect been observed among temperate basidiomycetes (H. H. Burdsall, personal observation) or tropical ascomycetes (Fig. 8.15; D. J. Lodge, personal observation).

Spore prints for agarics, most aphyllophorales, and some ascomycetes can be obtained in the field on sterile 60-mm-diameter Petri plates from which spores are transferred to tubes or agar. For some fungi, spore prints can be obtained on paper or aluminum foil, air-dried, and used later. Spore prints of some temperate fungi (e.g., morels) can be taken on aluminum foil and kept frozen for a year or more without affecting viability (T. Volk, personal communication). For others, such as *Armillaria*, air-dried spores more than several days old are not usually viable, and air-dried spores of *Marasmius* remain viable for only a couple of hours (D. Desjardin, personal observation). Likewise, the spores of many tropical rain forest fungi are sensitive to desiccation and lose their viability after 1–2 hours. When the period of spore viability is in doubt, it is best to drop the spores directly onto an agar surface.

Spore prints of basidiomycetes obtained in the field can be used to obtain single-spore isolates by suspension plating or streaking. Streaking is less complicated and

![FIGURE 8.15](image.png) Spores can be dropped directly onto agar plates under field conditions. Many fungi have fragile spores that will not germinate after being dried. (Photo of Dr. Karen Nakasone by D. J. Lodge)
involves transferring a drop of spores suspended in sterile water to an agar plate and then streaking the suspension over the agar surface using a piece of agar or a flame-sterilized loop or rod.

To obtain a spore print from air-dried basidiomycetes, the basidiome is refrigerated for 2–3 days in an agar-filled Petri dish (usually this procedure works only with certain temperate-zone aphyllorhizales). When returned to room temperature, the basidiomes will develop new basidia and produce a spore print. Resupinate basidiomycetes are more likely to produce spores if the hymenium is taken from wood rather than from bark. Many basidiomycetes, especially fleshy fungi like mushrooms, fail to drop spores if they have been refrigerated. Tropical fungi usually drop spores within the first 2 hours following setup; suspension of an hymenium over a plate for more than 12 hours leads to contaminated plates rather than spore maturation (D. J. Lodge, personal observation). Immature specimens of tropical fungi are better placed in a moist chamber with the original substratum intact and tagged and placed back in an easily accessible part of the forest until the basidiomes or ascomata mature.

Mature ascomata (moist and turgid, either fresh from the field or air-dried and rehydrated) are used for obtaining ascospores. Mature air-dried ascomata of Xylariaceae usually can be rehydrated by soaking them in water for 30 minutes to 2 hours prior to attempting the spore shoot or drop. For Xylariaceae and other ascomycetes that forcibly discharge their spores, contamination by hyphomycetes (and other fungi that do not forcibly discharge their spores) can be avoided by inverting the Petri dish, placing the ascomata on the lid below the agar surface, and allowing the ascomata to shoot the spores upward (J. D. Rogers, personal communication). It may be necessary to elevate the perithecia with a small piece of clay, small Petri dish lid, or something similar so that the discharged spores can reach the agar surface. The ostioles must not be clogged with spore deposits, which can be picked off with a sterile needle and streaked onto the agar surface after adding a drop of water from the lid. If ascomata fail to discharge spores, the ascus contents can be extracted with a sterile scalpel, flamed needle, dental tool, or insect pin and streaked onto the agar surface. Single spores usually can be obtained, but spore germination within the ascus is common in many species. We do not recommend attempting to culture other tissues, such as the ascomal wall or internal stromatal cells, of small ascomycetes, but that procedure may be used as a last resort for some larger Xylariaceae. However, cultures of Hypoxylon have been obtained from the internal stromatal tissues of Xylaria species (D. J. Lodge, personal observation), which illustrates that it is possible to isolate a contaminant rather than the intended fungus. Consequently, one cannot assume that a culture obtained from stromatal tissue necessarily belongs to the species that produced the ascocarp from which it was isolated.

**Isolating Tissue from Basidiomes or Vegetative Structures**

As mentioned earlier, we do not recommend isolating hyphae or spores from stromatal or ascomal wall tissues of ascomycetes. In contrast, cultures of basidiomycetes can be obtained by isolating sterile hyphae from the tissue of the basidiome. For mushrooms, the basidiome is split down the middle, and a small piece of tissue is removed from the upper stipe or pileus with a sterile scalpel or forceps. Tearing the basidiome apart instead of cutting it reduces the likelihood of contamination and reduces tissue damage. Reflexed or ungulate species that have tough basidiomes also can be isolated from the tissue or context. In that case, we recommend cutting part way through the basidiome and tearing the part from which the tissue will be removed. The extracted tissue then is placed directly on small test tube slants for growth. Media recipes and other procedural details can be found in Molina and Palmer (1982), Wadding (1980), and Stamets and Chilton (1983) (also see Appendix II).

Some thin, resupinate basidiomycetes that are difficult to culture from basidiospores or tissue can be isolated by means of “hyphal bridges” (D. J. Lodge, personal observation). The specimen on its substratum is placed inside a moist chamber, and aerial hyphae or mycelial strands that attempt to bridge air gaps by growing toward the paper toweling or lid can be transferred to agar plates. Vegetative structures of species that produce hyphal strands, rhizomorphs, or cordons can be fastened to the lid of an agar plate. Clean hyphae will grow from the broken ends and make contact with the agar in a day or two. Hyphal tips then may be transferred to fresh agar plates. Some species are difficult to culture from hyphal tips but will grow from larger masses of hyphae, in which case the agar plate with the rhizomorph should be inverted to reduce the risk of contamination once contact has been made. In such cases, large masses of hyphae are transferred on agar chips, some of which are placed on new agar with the hyphal side up (Fig. 8.16).

**Direct Isolation from the Host or Substratum**

Some fungi present in wood produce fruiting bodies infrequently, posing problems for diversity surveys. Fungi can be isolated from reasonably solid wood by placing chips or cores taken from the source on agar media. Recording the location from which a sample was
taken relative to the location of disease symptoms is especially important for many pathogens that can be isolated only from the disease front. Samples can be obtained using a flame-sterilized increment borer. The surface of the substratum to be sampled is cleaned thoroughly with a 70%-ethanol or a 10%-sodium hypochlorite wash, or a clean surface is exposed with a flame-sterilized knife. The increment borer is screwed into the wood to form the core. When the core is removed, it is placed in a sterile container and returned to the laboratory. Plastic drinking straws frequently are used to hold the cores. It is not possible, however, to place a sample from a friable woody substratum into the straw without destroying it. In such cases, the core can be placed in a piece of aluminum foil that then is rolled around it. Cores can be taken from sections of a substratum appropriate to the information desired. For example, if determining the internal distribution of the fungi in a log is important, then the position of the sections in the core must be recorded. Chips are handled in a similar fashion. Before plating, chips or cores are dipped in 95% ethanol and flamed for surface sterilization. Alternatively, the surface can be sterilized by washing for several minutes under a strong stream of distilled water. Surfaces of large pieces of wood or roots can be sterilized by washing with and then submerging in a 0.5% solution of sodium hypochlorite for 10–15 minutes. Chips then are cut using a flamed knife, and fungi are isolated as they grow from the core or chip.

Another method used to extract internal hyphae is to drill into a log and sample a few of the resulting shavings at given depths. The shavings are more difficult to handle than the cores, but they can be collected directly into culture tubes or plates and allowed to grow. The intermediate steps required by the coring method then are avoided.

Cultures of polypores and some other fungi can be identified using the keys and descriptions provided in Nobles (1965) and Stalpers (1978). Appropriate monographs must be used for other taxa.

**FIGURE 8.16** Some rhizomorph-forming corticioid species, such as *Phanaerochete flavida* shown here, can be cultured by inverting an agar plate and placing the fruit body on the lid. Rhizomorphs growing up to the agar surface are generally free of contaminants. (Photo by D. J. Lodge)
Collecting and Describing Macrofungi

FIGURE 8.17 Describing agarics and boletes requires good light so that all of the characters can be observed and the colors can be matched against a standardized color guide. Dr. Orson K. Miller is shown matching colors against a Methuen Color Guide, Hope Miller is describing specimens, and Dr. Clark Ovrebo, in the back, is taking photos against a neutral gray photo card in Belize. (Photo by B. Ortiz Santana)

project and the resources available. If two or more workers are available for descriptive work, it is usually most efficient to assign each worker responsibility for some groups or genera of fungi each day. When several workers are on a project for an extended period, assigning taxa leads to more consistent descriptions. If a lot of culturing is needed, however, one individual might do that instead of descriptive work (Fig. 8.17).

The procedure for describing macrofungi depends on the type of fungus under consideration and also on the investigator. Forms, worksheets, or computer formats should be organized so that a description is recorded in logical sequence (Figs. 8.18, 8.19, and 8.20). Most investigators begin with the pileus and move downward through the lamellae or tubes, stipe, and finally to any veils that are present. Recording information about the context is also important (e.g., color, color changes, consistency, presence or absence of latex), as is the inclusion of data on all available developmental stages. Procedures for studying macrofungi that lack lamellae and/or a stipe (e.g., coral fungi, puffballs) vary slightly, and monographs should be consulted for details. The goal in all cases is to describe the sporocarp well enough for someone else to picture it (Figs. 8.21 to 8.23).

Good lighting is critical for documenting color, vesture, and other features. The type of light used is very important and should be noted (e.g., natural, incandescent, fluorescent). Color notes can be taken in natural daylight, but an appropriate artificial light system is more reliable and consistent. A light system with two Vita-Lite fluorescent tubes works extremely well because it provides illumination that is close to natural light. Portable daylight-corrected light sources, such as Ott-Light True Color Lamps (Appendix IV), also provide high-quality light. Those lights and fixtures are smaller and more easily transported than large Vita-Lite fluorescent tubes. Normal fluorescent light tubes and incandescent light sources, however, distort colors, especially those in the red to purple range.

Experienced mycologists often record descriptions on cards, paper, or directly into a computer file. However, preprinted or computerized forms ensure inclusion of all diagnostic features, and we recommend their use, especially if a team is describing the specimens. Equipment required for describing specimens include a pencil or pen with permanent ink; a pocket knife with a sharp, thin blade and single-edge razor blades for sectioning sporocarps; a hand lens or dissecting microscope for small
FIGURE 8.18  Agaric annotation sheet, in English. These are copied, and one is filled for each collection. (D. J. Lodge)
FIGURE 8.19 Agaric annotation sheet, in Spanish. (D. J. Lodge and S. Cantrell)
FIGURE 8.20 Bolete annotation sheet, in English. (B. Ortiz-Santana)
FIGURE 8.21 Nonagaric macrofungi, such as this *Ramaria cyanocephala*, are difficult to annotate, because the terms used to describe patterns of forking and the shape of the ultimate branches are different from those used to describe other groups. (Photo from Puerto Rico by D. J. Lodge)
FIGURE 8.22 Describing stink-horn fungi (Phallales), such as this *Dictyophora* species, is challenging for individuals who are not experts in this group. Consult Dring (1980) for appropriate descriptors and terminology. (Photo by G. Mueller)
fungi and fine detail; forceps for handling specimens and cleaning particles of dirt and debris from specimens; a millimeter rule; and depending on the goals of the study, a spot plate and chemicals for macrochemical tests (see “Testing for Macrochemical Color Reactions,” later in this chapter). References such as the Dictionary of Fungi (Kirk et al. 2001), How to Identify Mushrooms to Genus: Macroscopic Features (Largent 1986), Modern Genera (Largent and Baroni 1988), and other taxonomic references (see “Taxonomic Resources for Identification,” later in this chapter) are helpful, but not essential, books to have in the field laboratory as sources of names, descriptors, and definitions.

Accurate and consistent notation of sporocarp color, including color changes of mature sporocarps and colors of different developmental stages, is important when describing macrofungi, so we recommend use of standardized color names. Commonly used color guides are those by Ridgway (1912), Kornerup and Wanscher (1978), Munsell (1966), Rayner (1970), Maerz and Paul (1950), and Kelly (1965). Unfortunately, most of those books are out of print, but libraries often have copies of at least some of them. The Munsell system is the most comprehensive; it is still in print, but it is expensive.

**COLOR PHOTOGRAPHS**

Color photographs (slides, prints, or digital images taken with a megapixel digital camera) should be taken whenever possible because they are extremely valuable for documenting macromorphological features and can be used in a variety of ways. Photographs taken in the field are excellent for publications and lectures. Because field photography is time consuming, however, it is not always practical. The alternative is to photograph specimens in the laboratory or studio. Shaw (1987) gives a wealth of valuable information on field macrophotography.

The most important considerations in photographing mushrooms are adequate close-up capability and appropriate film for the light source. Magnification of 1:2 is the minimum, although for very small taxa 1:1 or better is necessary and can be obtained with a bellows or extension tube. The best color generally is obtained with daylight type film in combination with either sunlight or a flash. A ring flash, multiple flashes, or a bounce flash setup eliminates the “shot-at-night” look that often comes from a single directed light source. Some full-feature megapixel digital cameras give excellent results and can serve as an alternative to traditional film-based...
cameras (macro capability and control over depth of field through aperture priority settings are important features to look for when choosing a digital camera).

Specimens should be photographed on a black or neutral gray background in a way that illustrates important diagnostic features of the sporocarps and allows easy comparison of features. All stages of development available, a sporocarp cut lengthwise to reveal the interior, a top view of the pileus, and a view of the underside of the pileus showing the edges of the lamellae should be included. If the sporocarp is large, or if the specimen consists of only one sporocarp, pie-shaped sections of the top and underside are sufficient (Figs. 8.24, 8.25, and 8.26).

**Testing for Macrochemical Color Reactions**

Macrochemical color reactions, or spot tests, have been used for decades in the identification and classification of fungi. Such tests can be helpful in identifying genera or groups of species, especially in the genera *Ramaria* and *Russula* (Romagnesi 1967; Marr et al. 1986; Singer 1986b; but see Thiers 1997 for an opposing view). The tests are performed by placing reagents on pieces of the sporocarp and noting any color changes. Although reactions are not routinely checked in large biodiversity sampling programs because the tests are time consuming, they may prove useful when identification of key fungal taxa require such information. Marr and colleagues (1986) and Singer (1986b) provided recipes for and discussed the application of the most commonly used macrochemicals. Chemicals are applied to detached portions of sporocarps to prevent degradation of the specimen and interference with other chemical tests or extraction of DNA. No more than one reagent or solution should be applied to a piece of sporocarp. Some of the test chemicals are dangerous to handle so appropriate safety measures should be used. Both negative and positive color-change results should be recorded.

**Obtaining Spore Deposits for Identification**

The main reason for taking spore deposits is to determine spore color. Spore deposits should be observed on white paper, clean glass slides, or foil. Using spores from spore deposits during micromorphological analyses eliminates

**FIGURE 8.24** Obtaining the proper lighting (from the side but without hard shadows) for taking photographs of fungi in the field requires significant time. Such photographs are often the most esthetically pleasing, however, and they are best for delicate specimens that are easily damaged by handling. (Photo of *Laccaria amethystina* by G. Mueller)
FIELD photographs are often taken using a fill-flash to provide more light, thereby decreasing the exposure time required for hand-held shots, as well as softening hard shadows. (Photo of Mycena aff. leana in Costa Rica by T. O’Dell)
questions about the maturity of the spores because only mature spores are dropped. Spore deposits can be obtained from some macrofungi (e.g., coral fungi) by placing a piece of white paper directly under the branches of the sporocarp before it is wrapped or put into a container. With agarics, the cap is removed from the stipe and placed on a piece of white paper. Alternatively, a round hole (or a large "X") is cut in a piece of paper through which the stipe is inserted until the lamellae make contact with the paper. Only a portion of the pileus of a large basidiome is needed to obtain a spore deposit.

Mature sporocarps are placed in a relatively humid environment at 10–15°C. A large box (e.g., a cigar box) can be used to raise the humidity and can accommodate several caps. A plastic container also works well, as does wrapping the setup in aluminum foil or wax paper. Providing a moist environment may increase success if it is particularly dry. That can be done by placing a moistened piece of paper towel in the container, by stretching a piece of moistened paper towel tightly over the container before closing, or by taking the spore print in a closed plastic bag. The sporocarp should not be in water, and the collection number should be included with each spore deposit.

**MICROMORPHOLOGICAL FEATURES**

A compound microscope sometimes is used in the field laboratory to study spore characteristics, pileus anatomy, or other features that can help in initially assigning a specimen to a taxonomic group. It also can be used to assess the condition and maturity of small ascomycetes to help determine if a specimen should be kept. A quick look under the microscope can provide information that improves the overall description of the fresh material and allows one to check for features that are lost on drying. Some workers like to do a complete description of micromorphological features in the fresh condition. That is time consuming, however, and often impractical. Well-preserved specimens work as well as fresh material as a source of micromorphological data, so those analyses normally are done between collecting trips and off-season.
and Singer (1986b) have discussed micromorphological characters and the techniques and reagents needed to describe them (Fig. 8.27).

**TAXONOMIC RESOURCES FOR IDENTIFICATION**


**Basidiomycetes**

Dependable publications for identification of nonagaric basidiomycetes from woody substrata include Eriksson and Ryvarden (1973, 1975, 1976), Eriksson and colleagues (1978, 1984), Gilbertson (1974), Jülich and Stalpers (1980), and Gilbertson and Ryvarden (1986). Burt (1914–1926, reprinted as a book in 1966 with an index to new names and synonyms) can be used to identify many resupinate and erect nonagaric basidiomycetes from North America and the Caribbean, but his generic

![FIGURE 8.27 Some micromorphological characters in ascomycetes, such as the presence of a fluorescing ascus ring in Valsaria rubicsosa treated with Calcafluor, are ephemeral and are lost or difficult to see after drying. (Photo by S. Huhndorf)](image)
concepts are outdated and the species descriptions are not always dependable. The keys to genera of temperate fungi do not always work for tropical species, but annotated checklists of tropical and subtropical species facilitate identification once a genus has been determined (e.g., Hjortstam and Larsson 1994). The conspicuous Tremellales can be identified in North America using McNabb (1964a, 1964b, 1965a, 1965b, 1965c, 1965d, 1965e, 1966a, 1966b, 1969, 1973); in Europe using Breitenbach and Kränzlin (1984); and in the Neotropics, at least to genus, using Dennis (1970), Lowy (1952, 1971) or Ryvarden (Keys to Neotropical Polypores, unpublished manuscript) (Fig. 8.28).

Keys to North American and some neotropical polypore species are found in Gilbertson and Ryvarden (1986). A preliminary polypore mycota for East Africa was published by Ryvarden and Johansen (1980). Keys and descriptions for poroid basidiomycetes families, genera, and species also can be found in Domanski (1972) and Domanski and associates (1973). Other useful references for neotropical polypores include Murrill (1915, reprinted in 1973), Fidalgo (1968), Fidalgo and Fidalgo (1968), Furtado (1981), Ryvarden (2000), and Lodge and colleagues (2001). Pleurotoid, lamellate polypores (i.e., *Lentinus*) and agarics (i.e., *Pleurotus* and *Panus* sensu Corner) can be identified using the monographs by Pegler (1983b) and Corner (1981), respectively.

Keys to orders, families, and genera of gasteromycetes can be found in Breitenbach and Kränzlin (1986), Coker and Couch (1928), Miller and Miller (1988), Pegler and colleagues (1995); and Smith and Smith (1973). Keys to the genera and species of Clathraceae are available in Dring (1980), and keys to the genera and species of West Indian Nidulariaceae can be found in Broclie and Dennis (1954).

Wading and Wading (1980) provided an annotated list to the taxonomic literature for Agaricales. However, considerable literature on Agarics has appeared since 1980. Anyone working with those fungi, or any group of fungi for that matter, should visit a major library and conduct computer searches by genus and/or author to access that literature. Older taxonomic literature (1753–1821) is reviewed in Pfister and associates (1990). There are many field guides to temperate agaric fungi, especially for North America and Europe. A key to families and genera was published by Largent and Baroni (1988) as part of a Mad River Press series that also includes guides to morphological features of agarics (Largent 1977; Largent et al. 1977). Useful guides to European fungi include Bas and colleagues (1988–1995), Breitenbach and Kränzlin (1991, 1995, 2000), Courtecuisse and Duhem (1995), and Moser (1978). There is nothing approaching a complete agaric mycota for North America, but popular field guides by Arora (1987), Bessette and associates (1997, 2000), Lincoff (1981),

**FIGURE 8.28** Many identification guides are available for polypore fungi such as this *Bridgidoporus* species. (Photo by T. O’Dell)

**ASCOMYCETES**


**FIGURE 8.29** Many of the 'macro' Ascomycetes on wood, such as *Striatosphaeria codinaeaphora* shown here, are rather small. They must be magnified before they can be described and photographed. (Photo by S. Huhndorf)
FIGURE 8.30 Discomycetes, such as the *Sarcoscypha* shown here, are members of the Ascomycota. (Photo by G. Mueller)

FIGURE 8.31 Species, such as dead man's fingers (*Xylaria*); resupinate or pulvinate ones, such as most species of *Hypoxylon* (shown here); and related genera in the Xylariaceae are included in the Ascomycota. (Photo of *H. haematostroma* from St. John, U.S. Virgin Islands, by D. J. Lodge)