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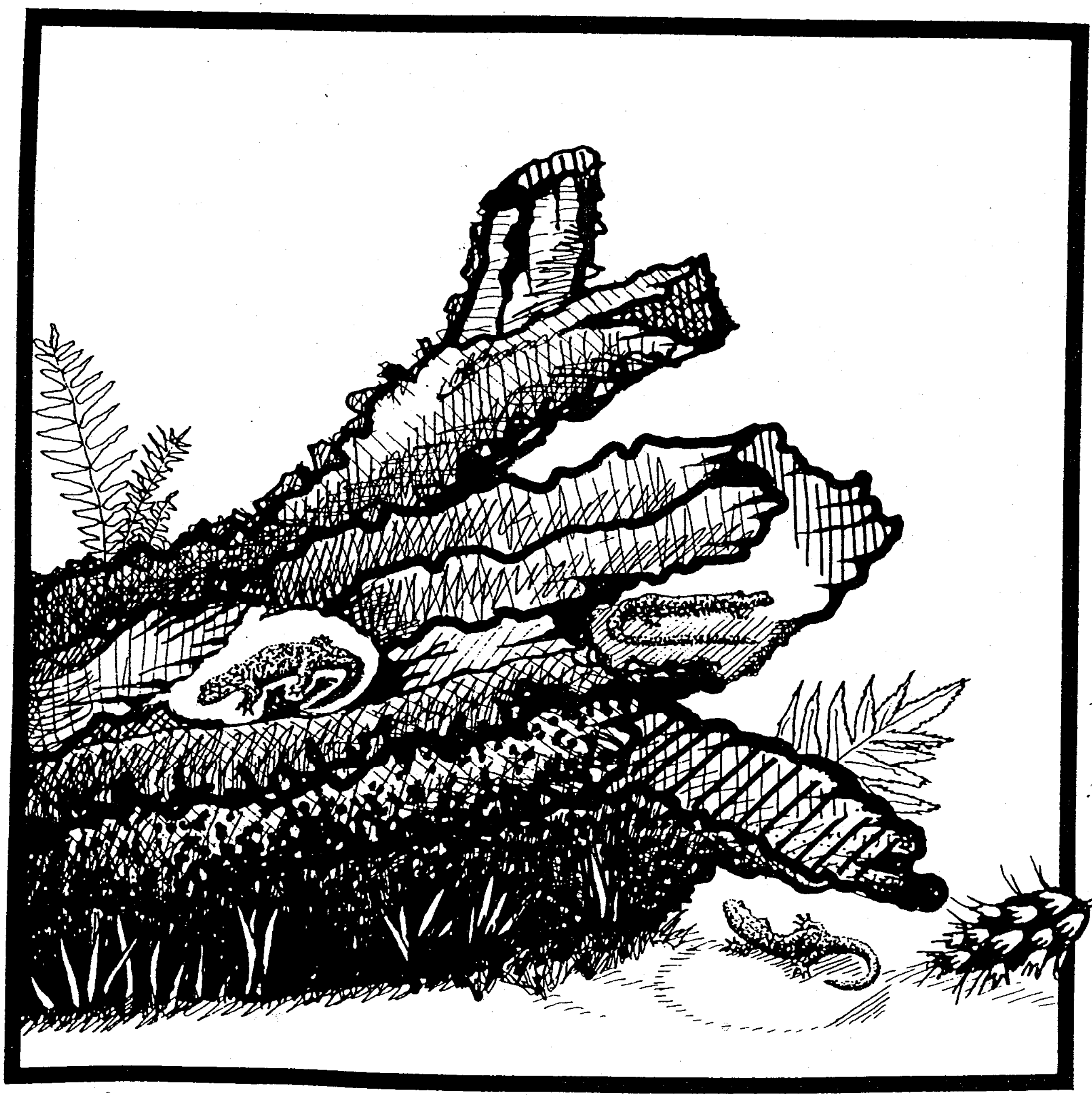

Wildlife-Habitat Relationships: Sampling Procedures for Pacific Northwest Vertebrates

Andrew B. Carey and Leonard F. Ruggiero, Technical Editors

Sampling Methods for Terrestrial Amphibians and Reptiles

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Preface

Concern about the value of old-growth Douglas-fir forests to wildlife in the Pacific Northwest began escalating in the late 1970s. The available information on wildlife-habitat relationships suggested that as many as 75 species including amphibians, birds, and mammals, could be dependent on old-growth forests. The USDA Forest Service chartered the Old-Growth Forest Wildlife Habitat Program to investigate the role old growth plays in maintaining viable populations of wildlife. It was apparent that broad surveys of vertebrate communities would be necessary to determine which species were truly closely associated with old-growth forests. Insufficient guidance on techniques, procedures, and sample sizes was available in the existing literature. We assembled a team of researchers from universities and Federal agencies to conduct pilot studies to develop sampling protocols and to test the basic experimental design for contrasting the wildlife values of young, mature, and old-growth forests. The sampling protocols resulting from the pilot studies were implemented in 1984-86 across broad areas of the Cascade Range in southwestern Washington and in Oregon, the Oregon Coast Ranges, and the Klamath Mountains of southwestern Oregon and northern California. Naturally, improvements were made to the protocols as time passed. A tremendous amount of experience in sampling was gained.

Our goal in this series is to compile the extensive experience of our collaborators into a collection of methodology papers providing biologists with pilot study-type information for planning research or monitoring populations. The series will include papers on sampling bats, aquatic amphibians, terrestrial amphibians, forest-floor mammals, small forest birds, and arboreal rodents, as well as papers on using telemetry for spotted owl studies and a guide to bird calls.

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Abstract

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Methods described for sampling amphibians and reptiles in Douglas-fir forests in the Pacific Northwest include pitfall trapping, time-constrained collecting, and surveys of coarse woody debris. The herpetofauna of this region differ in breeding and non-breeding habitats and vagility, so that no single technique is sufficient for a community study. A combination of pitfall trapping and hand collecting is the most effective approach.

Keywords: Amphibians, reptiles, sampling techniques, pitfall trapping, time-constrained collecting, downed wood.

Contents

Introduction

There is a rich herpetofauna in the Pacific Northwest, with 43 species of amphibians and reptiles present west of the Cascade Range (appendix 1). Depending on the geographical area, 19 to 32 species may be present at a given site (fig. 1). The number of species of amphibians is consistent at 13 to 15 species in most areas in this region, but reptiles range from 5 species in southwestern Washington to 17 species in both southwestern Oregon and northwestern California. The difference is due to increased aridity and higher temperatures in the southern locales, which favor reptiles. Although a diverse reptilian fauna may occur in an area, many species (particularly snakes) are locally rare or restricted to certain habitats; for example, oak-woodland (many snakes) or permanent water (turtles).

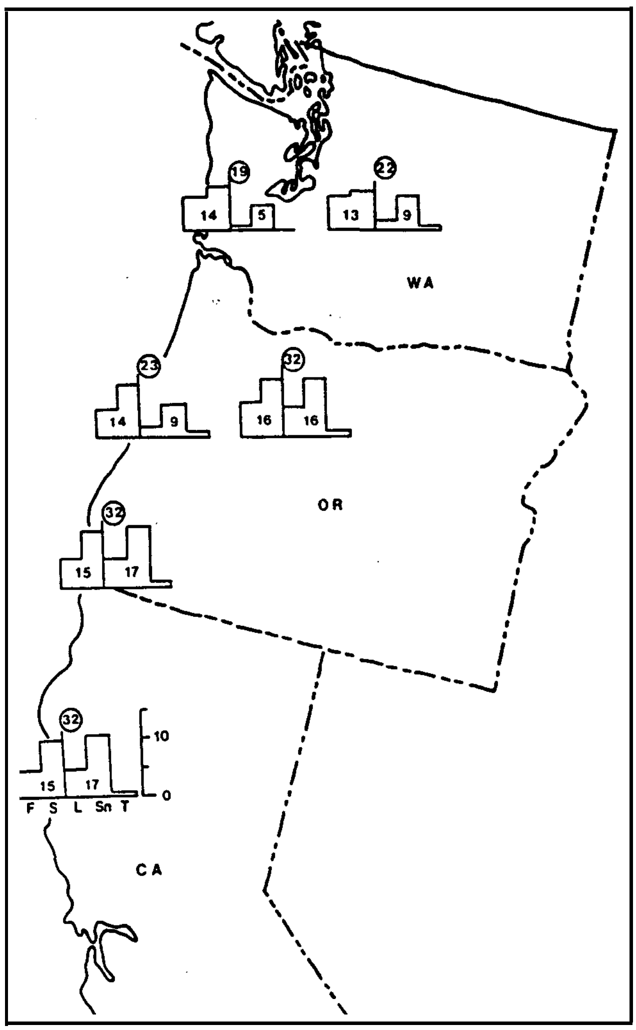

Figure l-Number of amphibians and reptiles potentially present in different regions of the Pacific Northwest west of the crest of the Cascade Range. The histograms are by major taxonomic groups: F = frogs, S = salamanders, L = lizards, Sn = snakes, and T = turtles.

During recent research in western Oregon and Washington, we found few or no reptiles present in closed-canopy Douglas-fir (Pseudotsuga menziesii (Mirb.) France) forests (Bury and Corn 1987, 1988; Corn and Bury, in press). Reptiles are usually encountered in rocky, open areas (for example, cliff faces) or in grasslands and oak woodlands (Herrington 1988, Nussbaum and others 1983); these habitats were rare or absent in the forest stands we studied. Thus, reptiles were a small fraction of the sampled herpetofauna, and they will receive little mention here. Biologists will need to employ special techniques if reptiles are encountered at a study site (see Bury and Raphael 1983, Jones 1986, Scott 1982b).

In the Pacific Northwest, amphibians are often found in terrestrial habitats, particularly in forests, and among terrestrial vertebrates may be the most numerous group. Terrestrial salamanders, for example, can exceed over five individuals/m* in local aggregations (Bury and Raphael 1983, Jaeger 1979). In 1983, Bury estimated that there were over 400 salamanders/ha in old-growth redwood forests in northern California (Bury 1983). In 1984, Raphael reported densities of 10 to 180 salamanders/ha in Douglas-fir forests in northern California (Raphael 1984). We estimated that mean density of plethodontid salamanders associated with downed wood ranged from 364/ha in young Douglas-fir forests to 744/ha in old-growth forests (Corn and Bury, in press). For eastern deciduous forests in New Hampshire, Burton and Likens (1975) estimated about 3,000 salamanders/ha, and Hairston (1987) estimated that energy present in salamanders in southern Appalachian forests exceeds that of all other vertebrate predators combined.

Amphibians are important components of the northwestern fauna in ways other than numbers or biomass. Of 22 amphibian species inhabiting forest habitats in the Pacific Northwest, 14 species (64 percent) are endemic (species whose distributions are restricted to the Pacific Northwest). Many of these habitats are affected increasingly by human activities.

Several species of plethodontid salamanders are more abundant in older forests, or show relations to habitat features that are prominent in old-growth forests. Ensatinas[1] are more abundant in older Douglas-fir forests than in younger stands in northern California (Raphael 1984). Ensatinas, Oregon slender salamanders, and clouded salamanders are often associated with large pieces of downed wood (Aubry and others 1988; Bury and Corn 1988; Corn and Bury, in press). Coarse woody debris (CWD) is a major component of old-growth forests and is severely reduced by modern forestry practices (Harmon and others 1986, Maser and Trappe 1984). The plethodontid salamanders in general are useful for assessing logging impacts because they have completely terrestrial life cycles (the eggs are deposited on land and hatch into miniature individuals), and most species have stable populations (Hairston 1987).

[1] Scientific names of reptiles and amphibians are given in table 6 (appendix 1).

The relations of frogs and aquatic-breeding salamanders to older forests are more difficult to explain than are the relations of plethodontids. Most of these species use terrestrial habitats to a degree, especially for feeding. They also may migrate overland to breeding ponds or streams and, thus, temporarily occur in many habitats during their travels. Tailed frogs previously had been considered to be closely tied to streams (Metter 1967) but we discovered that they are found in forests long distances from flowing water (Bury 1988). Our results also suggest that juvenile tailed frogs disperse into terrestrial habitats away from streams.

Given the diversity of amphibian life histories, habitat preferences, and different means of locomotion, more than one sampling technique is needed to sample adequately all species of amphibians. We used several methods to sample amphibians; methods for sampling aquatic species are discussed separately (Bury and Corn, in press). We sampled the terrestrial herpetofauna in three main ways: (1) time-constrained searches (TCS), (2) searching specified numbers of pieces of downed wood (CWD surveys), and (3) pitfall trapping.

We will discuss the objectives, sampling design, and techniques specific to each method separately. We will then discuss techniques common to all the methods we used and make recommendations for effectively sampling the herpetofauna in the Pacific Northwest.’ The methods described here were used by the Old-Growth Forest Wildlife Habitat Program (Ruggiero and Carey 1984) in field work from 1983 to 1985. This program included studies of vertebrates in Douglas-fir forests in California, Oregon, and Washington west of the Cascade Range (Ruggiero and others, in press). With the exception of experiments to determine the most effective design for pitfall trapping (Bury and Corn 1987), these methods were not rigorously tested against alternatives (field methods, particularly hand-collecting techniques, have rarely been subjected to experimental comparisons). Rather, they reflect our current professional judgment and draw heavily from other recent descriptions of field methods (Campbell and Christman 1982, Jones 1986, Raphael and Barrett 1981, Vogt and Hine 1982).

Objectives

Overview

The primary objective of our study was to identify species associated with old-growth Douglas-fir forests (Ruggiero and Carey 1984), and so the techniques we used were slanted to favor survey methods. Pitfall trapping and CWD surveys will provide some information on populations. These data can be used to analyze habitat use by individual species and the patterns shown by groups of species in different habitats. Coarse woody debris surveys and TCS can also provide detailed information on the use of. microhabitats by various species. Basic ecological data are needed that can be applied to recommendations for management of specific habitats.

There are marked differences in catch between hand collecting (TCS and CWD surveys) and pitfall trapping (table 1). Species such as clouded salamanders and Oregon slender salamanders are closely associated with CWD and were frequently caught by hand but were trapped infrequently. Tailed frogs, newts and other migratory species were trapped effectively in pitfalls but rarely were caught by hand.

The choice of a specific method to achieve stated objectives depends on the species under study as well as the scope of the objectives. If a small-scale study on one or a few species is intended, then only one method may be needed. A survey of community structure over a large geographic area will likely require all three methods.

Table 1-Comparison of captures of amphibians and reptiles by pitfall trapping and time-constrained searches (TCS), H.J. Andrews Experimental Forest, 1983

Species	Number of captures		
		Pitfalls[b]	
	TCS[a]	Summer[c]	Fall[c]
Total	281	206	822
	Percent of captures (rank)		
		Pitfalls	
	TCS	Summer	Fall
Salamanders:			
Northwestern salamander	0 (-)	0 (-)	5 (5)
Pacific giant salamander	0 (-)	1 (13)	2 (6)
Clouded salamander	28 (2)	3 (10)	1 (8)
Oregon slender salamander	22 (3)	4 (7)	1 (10)
Ensatina	43 (1)	24 (1)	25 (2)
Dunn's salamander	1 (5)	1 (14)	1 (9)
Rough-skinned newt	1 (6)	15 (2)	37 (1)
Frogs:			
Tailed frog	0 (-)	9 (5)	19 (3)
Pacific treefrog	2 (4)	4 (8)	1 (7)
Red-legged frog	0 (-)	1 (12)	6 (4)
Lizards:			
Western skink	1 (8)	11 (4)	1 (12)
Northern alligator lizard	1 (9)	13 (3)	1 (11)
Western fence lizard	1 (7)	3 (11)	1 (15)
Snakes:			
Rubber boa	0 (-)	1 (15)	0 (-)
Northwestern garter snake	0 (-)	7 (6)	1 (14)
Common garter snake	0 (-)	4 (9)	1 (13)

[a] TCS were done for 8 staff hours in 18 study areas in April.
[b] Arrays of pitfall traps with drift fences (Bury and Corn 1987) were operated in the same areas for 180 days from late May to November.
[c] The results of pitfall trapping are divided into the first 90 days of trapping (summer) and the second 90 days (fall).

Throughout this paper, we will use the terms study site and stand interchangably. This is due to the bias of working in forests, where study sites tend to encompass areas of more or less uniform habitat, which are referred to as stands. Stands in the old-growth studies were patches of forest of uniform age with a minimum area of 10 ha (Carey and Spies, in press).

Time-Constrained Searches

Time-constrained searches involve searching study areas for amphibians and reptiles, which are immediately collected by hand (Bury and Raphael 1983, Campbell and Christman 1982). Equal effort is expended in each area searched, as measured by the number of staff hours spent searching. Thus, each search will have a specific time limit, dependent on the prescribed effort and the crew size. Time-constrained searches are most useful for determining presence or absence of species and for providing initial data on the types of microhabitats occupied by individual species.

Time-constrained searches are not suitable for providing population data beyond presence or absence. Because this is a "plotless" technique, the same amount of potential habitat tends to be searched in each study area: however, amounts of suitable habitat differ among study areas. Results from some TCS may show habitat-poor areas yielding similar numbers of animals as habitat-rich areas, even though the population sizes may be quite different. Indeed, evidence is that salamanders are more clumped in areas with less habitat, which will increase the bias in favor of these areas. In the Coast Ranges of Oregon, we found the density in downed wood (number per m^3) of ensatinas was significantly higher in young and mature stands compared to old growth (fig. 2) (Corn and Bury, in press). In this case, TCS could possibly result in an inverse relation of numbers caught to' actual population size.

If population estimates are an objective, then other techniques need to be applied. We used CWD surveys effectively (explained below), but another common method is complete removal of all residents of a predetermined area (Bury 1983; Campbell and Christman 1982; Jaeger 1979; Raphael 1984; Scott 1976, 1982a). Plot searches are labor intensive: Bury (1983) required 20 to 44 staff hours to search 0.125-ha plots in old-growth redwood forests in northern California. For surveys of several study areas, plot searches may require too much effort to produce sample sizes large enough for statistical analysis.

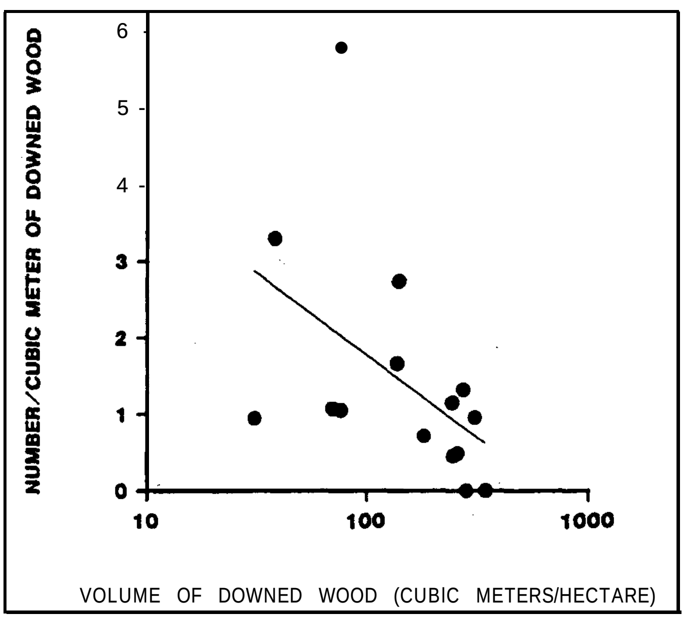

Figure 2-Salamander density in downed wood. The density of salamanders (number/m^3) in downed logs is inversely related to the amount of downed wood present in the study areas. Salamanders appear to be less clumped as more habitat is available.

For initial surveys of presence or absence, TCS are more effective than plot searches because collectors are free to examine large objects over a wide area, and usually more amphibians are found in large objects than in the leaf litter, at least in the Pacific Northwest. This method is efficient because the objects searched are most likely to yield animals. In northwestern forests, TCS may produce as much as a 10 times greater yield than will area-constrained collecting (Bury and Raphael 1983). In recent studies, capture rates of TCS have ranged from one to two animals per staff hour in the Cascade Range in Oregon and Washington (Aubry and others 1988, Bury and Corn 1988), to over eight animals per staff hour in northern California (Welsh and Lind 1988). Time-constrained searches are best employed when several study areas need to be surveyed in a short time.

Surveys of Coarse Woody Debris

In 1985, we were confronted with the choice of initiating TCS in the Oregon Coast Ranges or developing a technique to quantify habitat use and estimate density of selected species of salamanders. We chose the latter and developed a technique involving searches of predetermined numbers of pieces of downed wood. Numbers of animals caught were then related to the amounts of CWD in the stand, and minimum-density estimates were calculated.

Surveys of CWD are operationally similar to TCS; but to estimate animal densities, the density of CWD must be known. Knowing the amount of CWD present also allows for quantifying microhabitat use and drawing meaningful comparisons of microhabitat use among species.

The primary drawback of surveys of CWD is that density estimates apply to only one feature of the habitat. Surveys of CWD underestimate density of species using downed wood only occasionally; for example, most species of woodland salamanders *(Plethodon* spp.) frequent rocky soils, but an unknown fraction of a population may occur in CWD. For species strongly associated with CWD (for example, the clouded salamander or the Oregon slender salamander), surveys of CWD should provide general estimates of population sizes.

Pitfall Trapping

Pitfall trapping is a flexible technique that can be used to achieve several objectives; for example, drift fences with pitfall traps have been used to encircle specialized habitats such as amphibian breeding ponds (Gibbons and Semlitsch 1981, Shoop 1968, Storm and Pimentel 1954). This technique can be used for complete enumeration of breeding populations. Pitfall trapping also has been employed widely for surveys of amphibian and reptile diversity and abundance in different habitat types (Bury and Corn 1987; Campbell and Christman 1982; Friend 1984; Jones 1981, 1986; Raphael 1984; Vogt and Hine 1982; also see selected papers in Ruggiero and others, in press; and Szaro and others 1988). The main drawback of pitfall trapping is that trapability differs widely among species (Bury and Corn 1987, Campbell and Christman 1982, Gibbons and Semlitsch 1981). A survey of ail species of herpetofauna in an area therefore requires more than one technique.

Pitfall trapping provides data on the presence or absence of species, and because the trapping effort can be quantified and standardized across study areas, relative abundances can be calculated. Estimates of actual population size may be possible, though probably only for abundant species. Pitfalls may be used as live traps if checked frequently, and mark and recapture techniques also may be used. If pitfalls are used as a removal method to estimate density, then the area being trapped must be known. This is extremely difficult to determine for most herpetofauna and is something we have not done in any of our studies.

Pitfall trapping is also useful for investigating seasonal activity patterns. Traps can be operated continuously, so that variation in activity due to weather can be detected (Bury and Corn 1987). Pitfall traps are permanent structures, so long-term monitoring can be accomplished by operating the same trap array or grid periodically over several years. Trapping has unknown effects, however, on population structure due to the removal of resident individuals.

Experimental Design

Time-Constrained Searches

This technique is a quick survey method requiring few restrictions on the approach. Three points need to be considered: (1) collecting should be done away from forest edges; (2) aquatic habitats, such as breeding ponds or creeks should be avoided-these are covered by a separate protocol (Bury and Corn, in press); and (3) collecting should cover as much of the stand as possible. There are two ways to accomplish this last point. One is to devote enough time to the search to be able to collect across the entire study area. The second is to restrict the search to a fairly small area (for example, a circle with a radius of 25 m) and restrict the amount of time spent collecting. The number of smaller areas that can be searched in each study area depends on the amount of time devoted to the TCS. We found that 6 or 8 staff hours of collecting were sufficient; few additional species were detected by collecting for longer than that. If 1 hour is spent in each of the subsamples, then six to eight areas can be searched in each study area.

Surveys of Coarse Woody Debris

This technique is somewhat more complicated than TCS in that it involves systematicaily searching a predetermined number of logs in each study area. Several questions must be addressed when a study is designed, including how many logs to sample, how to apportion the sample among the different decay states of downed wood, and how to select the logs sampled.

in 1985 in the Oregon Coast Ranges, we conducted CWD surveys in 18 study areas. Each survey included 30 logs greater than 10 cm in diameter. We selected 10 logs in decay classes 1 and 2, 10 in decay class 3, and 10 in decay classes 4 and 5 (see Franklin and others [1981] or Maser and Trappe [1985] for methods of classifying CWD). The three decay categories that we used reflected natural divisions of the five-class scale. Class 1 and 2 logs are intact with more or less complete bark cover. Class 3 logs have decaying sapwood, and the bark is beginning to slough off. Class 4 and 5 logs are thoroughly decayed, have little bark, and are disintegrating. We recommend sampling equal numbers of logs in each of these decay categories. We searched each log for a maximum of 20 staff minutes.

We found salamanders in only 37 percent of the logs (198 of 536) that we examined, so a sample size of 30 logs per stand should probably be considered the minimum. If few logs are sampled and salamanders occupy a small percentage of these, then the estimates of salamander density will be based on minimal information.

Logs to be sampled are best selected by a systematic sampling scheme (Mendenhall and others 1971). If the study area has not been mapped and the locations of all logs determined, it will not be possible to draw a random sample. A systematic sample involves selecting logs in a specified order as they are encountered while the crew moves through the stand.

Pitfall Trapping

Planning pitfall trapping mainly involves selecting the appropriate trap design. We used two different pitfall designs in our old-growth studies (fig. 3). In 1983, we used arrays of pitfall traps with aluminum drift fences (Bury and Corn 1987). In 1984 and 1985, we used grids of single pitfall traps without fences. There were quantitative and qualitative differences in the yield of each technique that must be considered in planning a project.

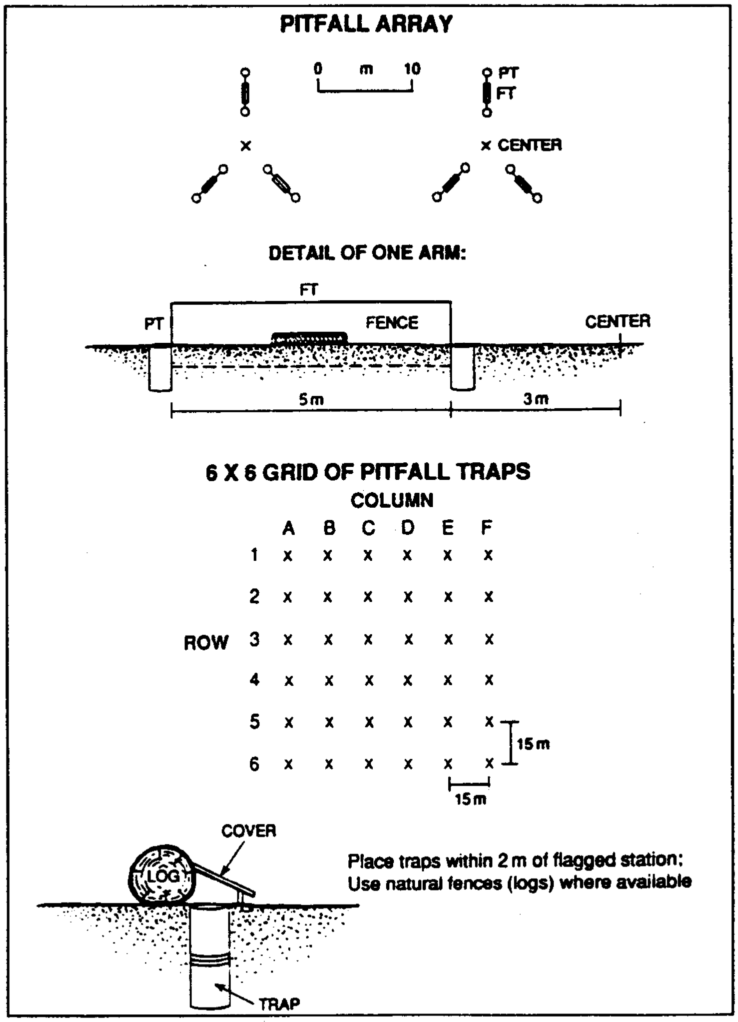

Figure 3—Designs for arrangements of pitfall traps either with or without drift fences. PT = pitfall trap, FT = funnel trap.

Trap rates for salamanders are similar for both arrays and grids, but arrays caught considerably more frogs and reptiles than grids did (table 2). The differences were due, in part, to the absence of drift fences in pitfall grids and the season when trapping was done. Pitfall arrays were open continuously for 180 days in 1983, from May to November. Grids were open for 30-50 days, beginning in October 1984. The grids were operated too late in the year to capture reptiles and large numbers of postmetamorphic juvenile frogs, which disperse from breeding sites in late summer or early fall. These frogs composed the majority of frogs caught by the arrays.

Although arrays catch more animals than grids do, arrays are not necessarily better for determining presence or absence of amphibians. Grids caught few reptiles but were able to detect amphibians, including frogs, as well as or better than arrays (table 3).

Table 2—Capture totals and trap rates (captures/100 trap nights) for major groups of amphibians and reptiles for 30 pitfall arrays in the Cascade Range of Oregon and Washington, 1983 (180 days), and for 48 pitfall grids in the Oregon Coast Ranges, 1984 and 1985 (80 days)

	Pitfall arrays			Pitfall grids		
Group	Species	Captures	Trap rate	Species	Captures	Trap rate
Salamanders	10	1145	1.77	8	1762	1.27
Frogs	3	915	1.41	2	103	.07
Lizards	3	79	.12	1	1	<.01
Snakes	3	41	.06	0	0	0

Table 3—Species of amphibians and reptiles inhabiting Douglas-fir forests at Old-Growth Wildlife Habitat Program study areas in Washington and Oregon

	Pitfall arrays[a]		Pitfall grids[a]
Species	Central Oregon Cascade Range	Southern Washington Cascade Range	Oregon Coast Ranges
Amphibians:			
Northwestern salamander	C	C	C
Long-toed salamander	P?[b]	P?	—
Cope's giant salamander	—	P	—
Pacific giant salamander	C	C	C
Olympic salamander	P	C	C
Clouded salamander	C	—	C
Oregon slender salamander	C	—	—
Ensatina	C	C	C
Dunn's salamander	C	—	C
Larch Mountain salamander	—	C	—
Van Dyke's salamander	—	—	P?
Western red-backed salamander	P?	C	C
Rough-skinned newt	C	C	C

Table 3—continued

Species	Pitfall arrays[a] Central Oregon Cascade Range	Pitfall arrays[a] Southern Washington Cascade Range	Pitfall grids[a] Oregon Coast Ranges
Tailed frog	C	C	C
Boreal toad	P	P	P
Pacific tree frog	C	C	P
Red-legged frog	C	C	C
Yellow-legged frog	P?	—	P?
Cascades frog	P?	P?	—
Spotted frog	P	P	—
Detection Efficiency[c]	59-77	62-77	77-83
Reptiles:			
Western skink	C	—	P
Northern alligator lizard	C	C	C
Southern alligator lizard	—	P?	P
Western fence lizard	C	P	P
Rubber boa	C	P	P
Sharp-tailed snake	—	—	P
Ring-necked snake	P	—	P
Gopher snake	—	—	P
Sierra water snake	—	—	P?
Terrestrial garter snake	P?	P?	P
Northwestern garter snake	C	C	P
Common garter snake	C	C	P
Western rattlesnake	—	—	P
Detection Efficiency	75-86	43-60	8

[a] P= potential occurrence, C = captured in pitfall traps, — = species does not occur in the area.
[b] Potential occurrence of a species in our study areas was uncertain.
[c] Species captured + species potentially present × 100.

The choice of whether to install arrays or grids ultimately depends on the needs of the study. Arrays are superior for catching reptiles, but reptiles may not be abundant in forest habitats or of interest to the goals of a study. Arrays can provide large sample sizes in relatively short periods. Grids remove fewer animals than arrays and may be more suitable for long-term monitoring. Both techniques are effective for catching small mammals as well as amphibians.

Arrays may be placed in pairs, as we operated them in 1983 (fig. 3), or single arrays may be placed at more than one location within a stand. Three or four single arrays scattered throughout the stand may better assess the variation within study areas, but this approach requires significantly more time for checking the traps in each area.

Cost may be part of the decision on whether to install arrays or grids. Grids are not substantially cheaper in cost of materials, because more pitfall traps can supplant the cost of fencing. Grids took only about one-half the effort to install as the arrays did. When personnel costs are high, this can result in a large difference in cost between the two methods. The cost involved in checking the traps is similar and depends mainly on the number of stands and the travel time between them.

Field Methods

This section provides instructions for carrying out TCS, surveys of CWD, and pitfall trapping. We will not discuss selection of study areas. If the study is an integrated wildlife survey, then study areas for mammals or birds can be used just as well for studying the herpetofauna. All the techniques discussed here require small areas as compared to bird or mammal studies.

Crew Sizes

Optimal crew sizes depend on the technique being used. Time-constrained searches and surveys of CWD use the same collecting techniques, and three to four persons are suitable for both. In both crews, one person is the data recorder, and the remaining people do the collecting. A 6-staff-hour TCS, done with a two-person crew plus a recorder who does not collect, requires 3 hours, plus the time for breaks.

For pitfall trapping, a large crew is generally necessary to install traps, but only one or two people are needed to check the traps once they are open. Installation of either arrays or grids is relatively fast with a crew of six. Crews of this size can install two arrays or grids per day. Two people can check a grid of 36 traps in an hour or less. Several sites can be checked in one day, depending on the travel time between study areas.

Time Frame and Weather

Hand collecting (TCS and surveys of CWD) should be done when amphibians are most likely to be active; that is, in the rain. In the Pacific Northwest, this is either in spring or fall (it rains in winter also, but low temperatures inhibit surface activity by amphibians). If there are several study areas, then the primary consideration is that the weather be as consistent as possible throughout the collecting period. Activity of amphibians is highly dependent on weather, and comparisons between areas of collection under radically different weather conditions may not be valid. Collecting therefore should begin as early as possible in spring or as late as possible in fall, but still avoiding lengthy periods of cold and snow. Collecting should not be done in heavy snow; light snowfall in a period of wet weather probably will not seriously affect amphibian activity. Two TCS can usually be done in one day, but one survey of CWD requires most of a day. It is possible, but not recommended, to split a survey between two days.

Pitfall trapping has more flexibility, because all traps are open at the same time; thereby reducing variability among study areas due to weather. The best season for operating pitfalls depends on the animals being trapped. For amphibians, spring and fall are again the periods of highest activity and will result in the largest catch. If reptiles are being sampled, then early summer is the best time to open pitfall traps. Pitfall installation can be done at any time, but data (Bury and Corn 1987) suggest that pitfalls should be in the ground at least 1 month before trapping begins.

Operating Guidelines

Time-constrained searches-Determine the number of 1-staff-hour searches that can be done in the amount of time allotted to each study area. On a topographic map or aerial photo of the study area, distribute the 1-hour searches for maximum coverage of the study area. The crew should enter each TCS with a map of the study area that shows the approximate location of each l-hour search and the path to follow between searches, with compass headings and approximate distances. Each 1-hour search should be confined to an area with a radius of about 25 m, and the center of each 1-hour search should be at least 75 m from any forest edge.

Each TCS is a survey of as much habitat as possible within each study area. Move from one object to the next after a few minutes. It is possible to spend over an hour at one large log, but a maximum of 10 minutes per object should suffice. Assuming a crew of two collectors and one recorder, each staff hour of search takes 30 minutes of actual time. When an animal is found, time is spent by the collector in assisting the recorder. The recorder should keep track of this time, and the total amount of data recording time is added to the end of the search, so that 1 full hour of collecting is achieved. This becomes more important in searches yielding many animals, because data recording will require more time.

Surveys of CWD-The techniques involved here are more precise than those used in TCS. Logs are chosen by a systematic sampling scheme. Specifically, a choice is made to sample one log out of a certain number of logs encountered. In most habitats, choosing one out of every three logs will produce a survey covering a large proportion of the study area. Further, logs are divided into subsamples based on the decay state of the log. We compressed the standard five-point decay classification into three categories: category A-decay classes 1 and 2, category B-decay class 3, and category C-decay classes 4 and 5. Sample 10 logs in each category (one of every three logs encountered in each category) for a total of 30 in each study area.

Plot a path through the study area that will cover a large portion of the area but will not intersect itself. For each decay category, choose a random number from one to three. Begin following the designated path. At every downed log, determine the decay category and whether the log should be sampled. The recorder keeps a running tally of the number of logs encountered in each category. Each category of log accumulates at its own pace, and whether a log is sampled depends on the number of logs encountered in that decay category. The decision may be, for example, to sample every second category-A log, every third category-B log, and every first category-C log. For this example, table 4 shows which logs will be selected from the first 20 logs encountered.

When a log is selected, measure the total dimensions (see appendix 2 for data forms and a description of the data to be recorded). Determine the tree species, if possible, and the slope and aspect of the site where the log occurs. Search the log for a maximum of 20 staff minutes. Carefully remove any bark and tear into the decayed wood layer by layer. If the entire log cannot be sampled within the time limit, search a portion of the log as completely as possible. This is very important, because salamander densities are based on the volume of wood actually searched.

Table 4—A hypothetical example of log selection in surveys of CWD

Log number	Decay category	Number encountered in decay category A	B	C	Sample log?[a]
1	A[b]	1			No
2	A	2			Yes
3	C			1	Yes
4	B		1		No
5	B		2		No
6	A	3			No
7	A	4			No
8	B		3		Yes
9	C			2	No
10	A	5			Yes
11	C			3	No
12	A	6			No
13	B		4		No
14	A	7			No
15	B		5		No
16	B		6		Yes
17	B		7		No
18	C			4	Yes
19	A	8			Yes
20	B		8		No
and so forth[c]					

[a] Assume that 1 out of every 3 logs is to be sampled, and the following sampling scheme is to be followed: category A, log number 2 of 3, category B, log number 3 of 3, and category C, log number 1 of 3.
[b] Decay categories: A = decay classes 1 and 2; B = decay class 3; and C = decay classes 4 and 5.
[c] Continue selecting logs until 10 logs in each decay category have been sampled.

Collecting tips—We have several pointers for more effective collecting for both TCS and surveys of CWD. Tools needed for both techniques include potato rakes and crowbars. It is necessary to purchase high-quality potato rakes; the less expensive ones cannot withstand extensive use. Crowbars are handy for peeling bark and breaking up the less-decayed logs. (See appendix 3 for a complete list of materials and tools needed to take samples.)

Large logs and bark piles adjacent to these or large, well-decayed snags are the most productive sites for TCS. Follow the instructions above for sampling logs. Other habitats should not be ignored during TCS, however. Moderate-sized debris (10 cm or more in diameter) on the forest floor should be turned over; often two people are needed to roll logs. In general, avoid raking through leaf litter or turning very small objects, but search piles of bark, slash, or mounds, because these often house amphibians. Rocks or boulders, if present, should be turned. Exercise caution when turning rocks on steep slopes. Be alert; searches often occur on rainy days when visibility is poor, especially under closed canopies. Salamanders can flee rapidly down a crevice, so grab them by cupping your hand on top of them. Frogs are elusive, and to catch them you may need the cooperation of two or three people to surround the quarry. Collectors should scrutinize the area under turned objects. Salamanders often freeze and most are cryptically colored.

Some species have special traits. Ensatinas are commonly found, and they rarely move once exposed. They are easily captured but must be picked up carefully or else they will autotomize (spontaneously amputate) their tail. Newts are slow moving but possess a highly toxic skin poison. This poison typically is released only if the animal is under attack but may show up during rough handling (for example, if the newt is hit by a rake tine). All terrestrial salamanders have some toxic secretions, but they rarely exude these substances when being handled.

The Oregon slender salamander and the Larch Mountain salamander often coil up, an apparent mimicry of distasteful millipedes that also curt up. Check any coiled animal closely. Clouded salamanders and western redback salamanders can move rapidly and need to be grabbed quickly. At least one hand should be bare to capture animals; gloves are usually too awkward for collecting agile species.

Snakes might be encountered during searches. Rattlesnakes occur at low elevations in Oregon and California, especially around rock outcrops. We recommend no collecting of rattlesnakes. Other snakes or lizards can be grabbed or, if fleeing, stepped on gently. Reptiles should be sluggish in cool, wet weather.

Habitat destruction can be minimized by returning cover items to their original positions. Roll small logs and rocks back and replace large pieces of bark slabs. Rake decayed logs back together and replace as much bark as possible. Some habitat destruction is unavoidable, but the organic material remains, and at least a portion of the log-soil interface can be restored by careful replacement of disturbed objects.

Installation of pitfalls-Place pitfall arrays and grids in spots representative of the study area. If single arrays are to be placed around the study area, the locations should be preselected from maps or aerial photos. The array or grid location should be at least 75 m from any forest edges (the farther, the better). For arrays, establish the center point of the first array at random. If a pair of arrays is used, measure 25 m from this point in a random direction for the center of the second array. For a grid, select one corner at random for the location of the grid. The grid is then laid out by using handheld compasses and 15-m tapes or measured ropes (necessary in dense brush). Installation of grids is generally fast with a six-person crew; four people lay out the grid, and two people begin installing traps. Two-person teams are best for grid layout. One person pulls the tape or rope until stopped by the second person, who remains at the previous station. Flag the new point and continue.

Pitfalls are constructed by fastening the open ends of two number 10 tin cans together with duct tape and then cutting the bottom out of one end (fig. 4). Traps are installed flush with the ground and have a plastic collar inserted at the top. This collar functions to keep animals from crawling out of the trap and is constructed by cutting the bottom out of a l-lb plastic margarine tub. When not being used for trapping, the traps should be closed; use the plastic lids from the margarine tubs. In grids, place the trap within 2 m of the station flag. If possible, place the trap next to a cover object, such as a rock or downed log. Traps next to logs should be placed on the downhill side of the log. The hole for the trap is dug most easily with a posthole digger, which creates a hole with the correct diameter. A tile spade can also be used. Traps have an additional optional wood cover. When the trap is open, the cover is suspended above the opening. This functions in part as a rain cover and partly to attract animals.

If an array design is being used, drift fences are constructed from 50-cm-tall aluminum valley roofing metal. This comes in 15.2-m rolls, which should be cut into 5-m sections before it is taken to the study area. We placed fences pointing away from the center of the array at equal (120^{o}) intervals. The interior end of each fence began 3 m from the center of the array (fig. 3). There are many other possible arrangements for placing pitfall arrays; see figures in Campbell and Christman (1982), Jones (1981, 1986), and Vogt and Hine (1982).

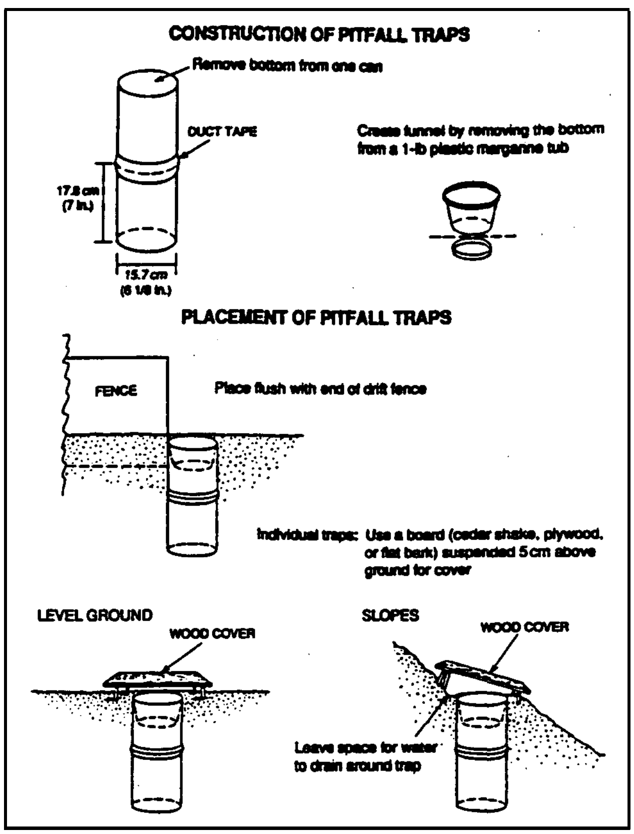

Figure 4-Construction and placement of a pitfall trap.

Use a mattock or hoe to dig a trench 20 cm deep and 5 m long, stand the fence into the trench, and back fill with soil. Occasionally an axe is needed to cut large roots. Tamp down the loose dirt so that the fence is self-supporting (stakes are not necessary for these relatively short fences), and smooth the dirt alongside the fence to create a runway. Move small obstacles (twigs, rocks) away from the fence. Traps are placed at the ends of the fence so that no gaps occur between the fence and the rim of the trap.

There are two important safety rules to follow when installing arrays. First, always wear gloves to handle the aluminum. The sharp edges can inflict serious cuts on unprotected hands. Second, exercise extreme caution in wet weather. The tools quickly become coated with slick mud, and a mattock or axe flying out of someone's hands is a lethal weapon.

Funnel traps will need to be constructed if reptiles are a target of the study (see Jones 1986, Vogt and Hine 1982). Funnel traps are constructed from window screen, which comes in rolls 76 cm wide. Cut a piece 90 cm long, and staple the ends together along the cut edge. Fold back the stapled edge so that you have a tube 25 cm in diameter by 76 cm long. Construct funnels by rolling square pieces of screen into a cone and stapling. Fold back the edge and attach to the tube. One end is fastened permanently with staples, and paper clips are used at the other end so that animals may be easily removed. Funnel traps are placed midway on both sides of each drift fence. Shape the trap and fill in with dirt so that no gap occurs between the fence and the trap. Shade the trap by placing loose bark or litter over the trap.

Pitfall operation-operating pitfall traps is a simple task. Techniques do not differ between arrays and grids. The primary decision is how frequently the traps should be checked. Check the traps every other day, if possible, but if there are a large number of study areas, then traps may have to be checked lessfrequently. Intervals of more than 5 days between checks should be avoided. Checking traps more frequently produces better specimens, particularly among the mammals that will be caught. If the number of study sites is such that all traps cannot be opened on the same day, care must be taken that all traps are closed in the same order they were opened in. This ensures the same trapping effort for each area.

Each time a trap is checked, remove debris that has fallen into the trap, and bail out excess water. A small amount of water should be placed in traps when they are opened, but in wet weather, most traps will accumulate more water than is desired. It has previously been recommended that water be placed in pitfall traps (Raphael and Barrett 1981, Williams and Braun 1983), and this is probably the quickest, most humane way to kill small mammals. Current guidelines for using pitfall traps to kill trap small mammals (American Society of Mammalogists 1987) specify drowning as the only acceptable method. But drowning is a slow and inhumane way to kill amphibians, and it has been prohibited in the current guidelines for field methods for herps (ASIH and others 1987). A generally acceptable compromise between these apparently incompatible recommendations is to keep a small amount of water (2 to 5 cm) in traps and check them frequently. Small mammals, particularly shrews, will become hypothermic and drown in this amount of water, but most amphibians should be able to survive.

All animals trapped in pitfalls are to be returned to the laboratory for processing. Separate mammals, live herps, and dead herps, but otherwise place all animals from the same trap in one plastic bag. Carry a field notebook with waterproof paper to record the number of individuals, species, and trap number of all animals caught. This record is important and should become a permanent part of the data set. It provides critical information during the initial processing of specimens and is a valuable reference for the questions that inevitably arise even after the data have been processed. Record the study area, date, and trap number in pencil on a small piece of waterproof paper and place in each bag of specimens. Bag all the specimens from a single study area together in a large plastic bag. Keep the specimens in a cooler with reusable ice containers while in the field. On returning to the lab, place dead specimens in a freezer and live herps in a cool space or refrigerator.

Identification

Accurate identification of specimens in the field is critical for TCS and surveys of CWD. Field identification is less important for pitfalls, because all specimens are examined later in the laboratory. The field notes listing the specimens caught in each trap are more valuable, however, if they are accurate. To increase accuracy, it is helpful for team members to examine series of specimens at a museum before field work begins. An additional field practice session is recommended to catch animals alive and to practice field identification. Most forms have distinct shapes or colors, but some species present problems. Most people have difficulty with woodland salamanders (Plethodon spp.), ranid frogs *(Rana* spp.), and juvenile salamanders. References for identification of northwestern herps are Nussbaum and others (1983) and Stebbins (1985). Other useful regional references are by Green and Campbell (1984) and Gregory and Campbell (1984).

Disposition of Specimens

All animals captured in pitfall traps are routinely euthanized and preserved (special consideration will need to be given to species with special status, such as those listed by the Federal or State governments as threatened or endangered). Specimens from TCS or CWD surveys may be treated in the same manner, or they may be released after the surveys near points where they were captured. If specimens are released, then positive identification is absolutely necessary (see above). Also, if animals are released, a representative series of voucher specimens should be retained from each study area and preserved. Capturing animals and retaining specimens requires valid scientific collecting permits from the appropriate State wildlife agency, and arrangements should be made before the study begins to deposit the specimens in an appropriate museum.

Process all specimens from a given survey, or all specimens collected from a pitfall site on a given day, together. This will provide for the most accurate recordkeeping, and it helps in solving the mystery of the occasional unlabeled specimen. Thaw any frozen specimens, and kill the live ones. Be sure to keep the label identifying the specimen closely associated with each specimen. Kill by relaxing amphibians in a dilute solution of Chloretone and by injecting reptiles with aqueous sodium pentobarbital. Chloretone is a saturated solution of hydrous chlorobutanol in 95 percent ethanol. An effective dilution is 2 ml per 570 ml of water. Sodium pentobarbital (Nembuto1[2] is one trade name) is a restricted drug and may be difficult to obtain. Reptiles may also be killed by injecting 95 percent ethanol into the heart region.

After the animal is dead, weigh and measure it (see appendix 2), tie a numbered tag to the right hind leg, and preserve in formalin. Create a lo-percent solution of buffered formalin by diluting commercial formalin to 10 percent and adding 4 g of baking soda or sodium carbonate per 400 ml of solution. Amphibians that appeared dead may begin to move when placed in the formalin. These should immediately be rinsed in water and returned to the Chloretone until dead. Amphibians and lizards should be laid out ventral side down in a shallow pan with a tight-fitting lid; for example, a plastic freezer container. Line the bottom of the pan with commercial paper towels (household towels have "dimples" that become imprinted on the skins of the animals), and pour a small amount of formalin into the pan. Snakes should be folded into an oblong coil with the head on the inside. The coil should be short enough to fit in the storage jars. Reptiles also must have formalin injected into the body cavity, limbs, and tail. Do not inject so much that a balloonlike specimen is created. If injection is not possible, then the body cavity, limbs, and tail must be slit to allow the formalin to enter the body. Body cavities of large Pacific giant salamanders should also be slit for thorough preservation. Pisani (1973) provides a thorough discussion of preservation techniques. Let the specimens fix in the formalin for at least 24 hours, then store in 50 percent isopropyl alcohol.

If specimens are released, then reasonably accurate measures of snout-vent and total lengths can still be made. Place the animal in a plastic bag and restrain it against the bottom of the bag. When the animal is quiet and relatively straight, measure to the nearest millimeter with a ruler. Mass can also be measured in the field with spring scales available in forestry supply catalogs.

The investigator should be aware that in northwestern forests, twice as many small mammals as herps generally are captured in pitfall traps. If a study is planned that uses pitfall traps, provision should be made for preserving the mammals. Neglecting this would be a criminal waste of valuable data.

Data Analysis

Numerous analyses can be done on the types of data collected from surveys of amphibian occurrence and abundance (see papers in Szaro and others [1988] and Ruggiero and others [in press] for examples). We will give a couple examples of the types of analyses that can be done, and we will discuss any special analyses that need to be performed.

All the techniques are excellent at providing data on presence or absence of species, and two or more techniques can be combined to provide a complete assessment of all the species potentially present. One example is provided by considering amphibians and reptiles detected by pitfall trapping with arrays and TCS at 18 study areas

[2] The use of trade, firm, or corporation names in this publication is for the information and convenience of the reader. Such use does not constitute an official endorsement or approval by the U.S. Department of Agriculture of any product or service to the exclusion of others that may be suitable.

in the Oregon Cascade Range in 1983 (table 5). Presence-absence data can be analyzed by calculating measures of similarity and then using a clustering procedure to look for patterns among groups of study areas (Pielou 1984). From the data matrix in table 5, similarities were calculated for every pair of stands by using Jaccard's index (Pielou 1984), which is the percentage of species both areas have in common compared to the total number of species present at either area. Clustering was accomplished by using the nearest-neighbor technique. One group of five old-growth and mature stands cluster together above the 60-percent level of similarity, but in general, there are few recognizable patterns related to habitat type (fig. 5). Pielou (1984) and Gauch (1982) are valuable sources of techniques for analyzing the structure and organization of communities.

Table 5-List of species of amphibians and reptiles present (P) at 18 study areas in and near the H.J. Andrews Experimental Forest, Lane County, Oregon, 1983

	Stand number																	
	Old-growth stands								Mature stands				Young stands			Clearcut stands		
Species	2	3	15	17	24	25	29	33	11	35	39	42	47	40	75	55	92	93
Amphibians:																		
Northwestern salamander						P	P					P	P		P		P	P
Pacific giant salamander	P						P					P		P	P	P		
Clouded salamander	P	P		P	P	P	P		P	P	P		P		P	P	P	P
Oregon slender salamander	P	P	P	P	P				P	P		P	P	P	P	P	P	
Ensatina	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P
Dunn's salamander	P					P	P											
Rough-skinned newt	P		P	P	P	P	P		P			P	P	P	P	P	P	P
Tailed frog	P	P		P	P		P	P	P		P	P		P			P	
Pacific trecfrog			P				P			P	P			P	P		P	P
Red-legged frog							P			P					P		P	
Reptiles:																		
Western skink							P									P	P	
Northern alligator lizard				P		P	P									P	P	P
Western **fence** lizard						P										P	P	
Rubber boa														P				
Northwestern garter snake	P		P	P												P	P	
Common garter snake		P	P		P			P		P				P	P		P	
Number of species	8	5	6	7	6	7	11	3	5	6	4	6	5	8	9	9	13	6

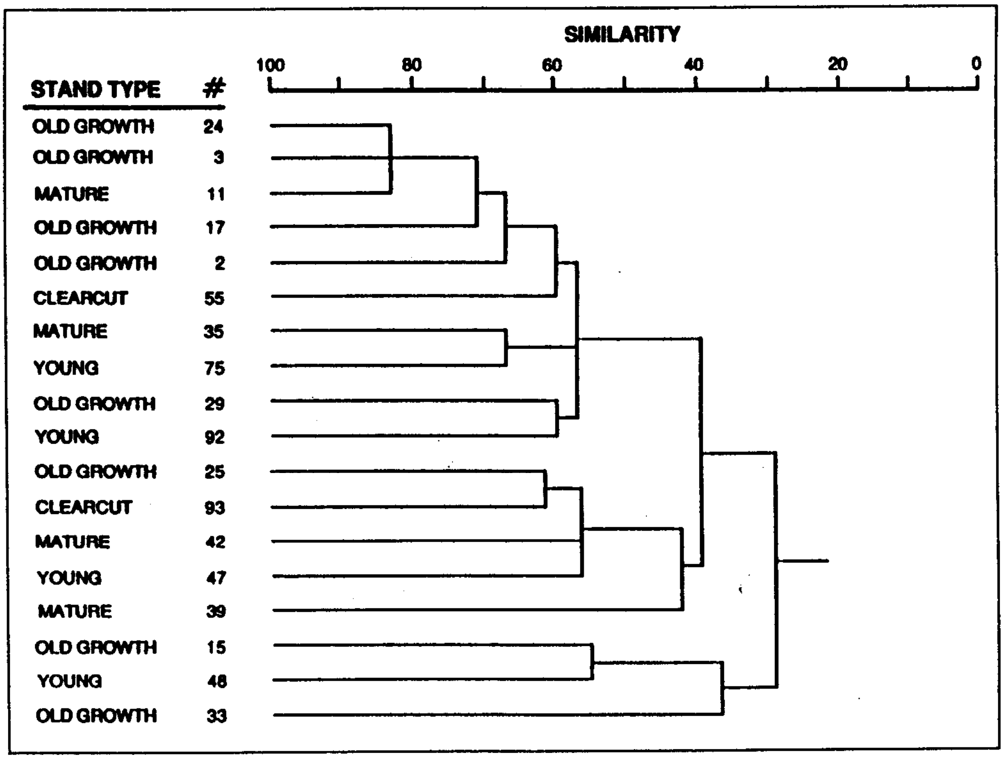

Figure 5—Nearest-neighbor clustering of herpetofauna at 18 study areas in the Oregon Cascade Range in 1983. Similarities were calculated by using presence-absence combined data from pitfall trapping and TCS.

Surveys of CWD can provide initial estimates of population density. The density in downed wood of each species of salamander (number per cubic meter) is calculated as the number caught in each log, divided by the volume of wood sampled in each log. Mean densities in downed wood in each stand were calculated for each of the three decay categories (decay classes 1 and 2, class 3, and classes 4 and 5). Use a nested analysis of variance (stands within forest age classes) to test whether density (log transformed) in downed wood of any species varies among decay categories or age class (old growth, mature, and young growth).

We calculated predicted densities of plethodontid salamanders in 45 forest stands from the following formula:

$$D = \sum_{i=1}^{3} (d_i \cdot V_i) ,$$

where D = number of salamanders per ha, d_i = density in downed wood in decay category i, and V_i = m^3 of downed wood per ha in category i. See Spies and others (1988) for techniques to determine the amount of downed wood present in a stand. Where d varied among age classes, D was calculated by using the mean density in downed wood for each age class.

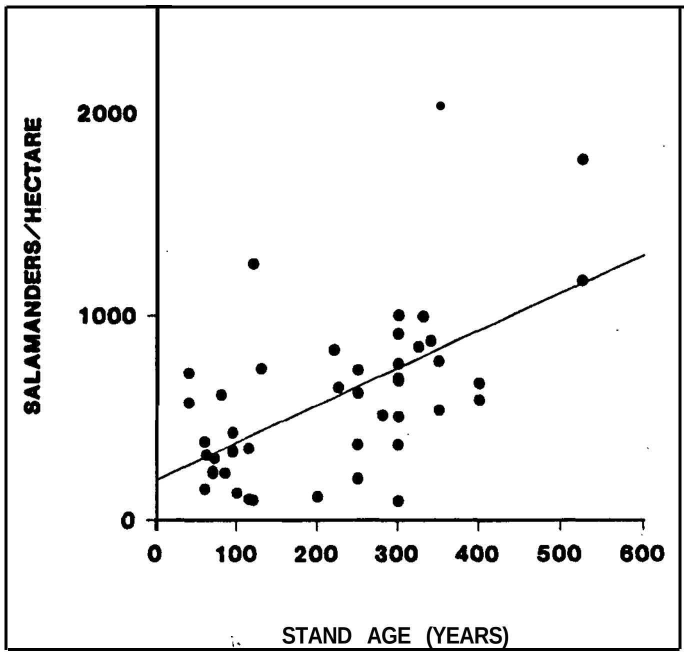

Figure B-Estimated densities of salamanders relative to forest age in the Oregon Coast Ranges in 1985. Data are based on surveys of CWD at 15 study areas. Density was estimated for an additional 30 stands, for which data existed on the volume of downed wood present in the study area.

Estimated density of plethodontid salamanders was related to stand age for 45 study areas in the Oregon Coast Ranges in 1985 (fig. 6). There were 15 study areas with surveys of CWD. Densities in the remaining 36 areas were estimated by using the average values of d for each habitat type and the measured value of V for each area.

Conclusions

There is a vast literature on techniques for sampling and analyzing vertebrate populations, but it was not our intention to provide a complete overview. Rather, we have described the specialized methods for sampling herpetofauna that we have used and refined in 3 years of field work in the forests of Oregon and Washington. Comprehensive references on sampling techniques include Cooperrider and others (1986) and Schemnitz (1980).

The methods we have described are most appropriate for surveys of forest-dwelling amphibians. Because these species use several habitats for breeding, feeding, and cover and differ widely in vagility, no single method is adequate to sample the entire community. Pitfall trapping needs to be combined with either time-constrained collecting or surveys of coarse woody debris in any comprehensive survey.

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Equivalents

When you know:	Multiply by:	To find:
Centimeters (cm)	0.394	Inches
Meters (m)	3.281	Feet
Square meters (m^2)	10.764	Square feet
Hectares (ha)	2.471	Acres
Cubic meters (m^3)	35.315	Cubic feet
Grams (g)	0.035	Ounces
Milliliters (ml)	0.035	Fluid ounces

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Appendix 1

Table 6-Scientific and common names of amphibians and reptiles found in the Pacific Northwest west of the Cascade Range from northern California to British Columbia

Scientific name[a]	4-letter code	Common name[a]
Amphibia, order Urodela (salamanders):		
Family Ambystomatidae-		
Ambystoma gracile	AMGR	Northwestern salamander
A. macrodactylum	AMMA	Long-toed salamander
Family Dicamptodontidae-		
Dicamptodon copei	DICO	Cope's giant salamander
D. ensatus	DIEN	California giant salamander
D. tenebrosus	DITE	Pacific giant salamander
Rhyacotriton olmpicus	RHOL	Olympic salamander
Family Plethodontidae-		
Aneides ferreus	ANFE	Clouded salamander
A. flavipunctatus	ANFL	Black salamander
A. lugubris	ANLU	Arboreal salamander
Batrachoseps *attenuatus*	BAAT	California slender salamander
B. wrighti	BAWR	Oregon slender salamander
Plethodon dunni	PLDU	Dunn's salamander
P. elongatus	PLEL	Del Norte salamander (includes *P, stormi)*
P, larselli	PLLA	Larch Mountain salamander
P. vandykei	PLVA	Van Dyke's Salamander
P. ***vehiculum***	PLVE	Western red-backed salamander
Family Salamandridae-		
Taricha granulosa	TAGR	Rough-skinned newt
T. rivularis	TARI	Red-bellied newt
T. torosa	TATO	California newt
Amphibia, order Anura (frogs and toads):		
Family Leiopelmatidae, *Ascaphus truei*	ASTR	Tailed frog
Family Bufonidae, *Bufo boreas*	BUBO	Western toad
Family Hylidae, *Hyla regilla*	HYRE	Pacific treefrog
Family Ranidae-		
Rana aurora	RAAU	Red-legged frog
R. boylii	RABO	Foothill yellow-legged frog
R. cascadae	RACA	Cascades frog
R. catesbeiana	RACT	Bullfrog (introduced)
R. clamitans	RACL	Green frog (introduced)
R. pretiosa	RAPR	Spotted frog
Reptilia, order Chelonia (turtles):		
Family Emydidae-		
Chrysemys picta	CHPI	Painted turtle
Clemmys marmorata	CLMA	Western pond turtle
Reptilia, order Squamata (lizards and snakes):		
Family Anguidae-		
Gerrhonotus coeruleus	GECO	Northern alligator lizard
G. multicarinatus	GEMU	Southern alligator lizard
Family Iguanidae-		
Phrynosoma douglassii	PHDO	Short-horned lizard
Sceloporus graciosus	SCGR	Sagebrush lizard
S. occidentalis	SCOC	Western fence lizard
Family Scincidae, *Eumeces skiltonianus*	EUSK	Western skink
Family Boidae, *Charina* bottae	CHBO	Rubber boa

Table 6-continued

Scientific name[a]	4-letter code	Common name'
Family Colubridae-		
Coluber constrictor	*c o c a*	Racer
Contia tenuis	COTE	Sharptail snake
Diadophis punctatus	DIPU	Ringneck snake
Lampropeltis getulus	LAGE	Common king snake
L. zonata	LAZO	California mountain kingsnake
Masticophis taeniatus	MATE	Striped whipsnake
Pituophis melanoleucus	PIME	Gopher snake
Thamnophis couchi	THCO	Sierra garter snake
T. elegans	THEL	Western terrestrial garter snake
T. ordinoides	THOR	Northwestern garter snake
T. sirtalis	THSI	Common garter snake
Family Crotalidae, *Crotalus viridis*	CRVI	Western rattlesnake

[a] Scientific and common names follow Banks and others (1987).
Sources: Nussbaum and others (1983) and Stebbins (1985).

Appendix 2

Data Sheets

Data sheet for TCS-This data sheet (fig. 7) needs to be on waterproof paper. The number of data sheets needed will depend on the number of animals captured. Note that each area search (1 staff hour) is listed separately and there is room for five animals per search. If more than five animals are captured in one area, then continue the data in the space for the next area, but if fewer than five animals are captured, then skip to the space for the next area before recording data from the new area. Data categories are explained below:

1. Standard header. This will differ by study. We illustrate the information we recorded in the old-growth study.
2. Weather (WR). Use the codes listed at the bottom of the data sheet.
3. Temperature (°C).
4. Start time, end time. Use 24-hour notation.
5. Crew. List the initials of the other crew members. The recorder should be the same person for each stand. Note whether or not the recorder participated in the collecting.
6. Catalog initials. Initials of the collector in whose catalog the specimens will be recorded.
7. Area. Each-l -staff hour search should be numbered sequentially.
8. Aspect (degrees). Record for each area searched.
9. Slope (percent). Record for each area searched.
10. Specimen number. Each herptile encountered is given a unique number, either sequentially for the entire stand (1, 2, 3,...n), or sequentially for each area searched (l-l, 1-2, l-3,...n; 2-1, 2-2, and so forth). Whichever method is used, the data collected in the laboratory (see below) must be matched to the data collected in the field.
11. Catalog number. This is the number given to preserved specimens. We use small, rectangular tags, preprinted with the catalog number.
12. Species. This is the four-digit code for each species (see appendix 1).

TIME-CONSTRAINED COLLECTING DATA

technique	province	habitat	stand #	day	month	year	wr	temp	start time	end time	crew	recorder
T C S												

area	aspect	% slope

spec #	catalog number	species	sex	snout-vent length (mm)	total length (mm)	mass (g)	vertical pos	tree species	decay class	cover object (cm) length	cover object (cm) width
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area	aspect	% slope

spec #	catalog number	species	sex	snout-vent length (mm)	total length (mm)	mass (g)	vertical pos	tree species	decay class	cover object (cm) length	cover object (cm) width
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VERTICAL POSITION:

1 - On Surface
2 - Under Rock (not in talus)
3 - Under Rock in Talus
4 - Under Log
5 - In Log
6 - Under Bark on Snag
7 - Under Bark on Log
8 - Under Bark on Ground < 1 m from Snag
9 - Under Bark on Ground < 1 m from Log
10 - Under Bark on Ground (> 1 m from log or snag)
11 - Other

WEATHER:

1 - Clear
2 - Partly Cloudy
3 - Overcast
4 - Fog Only
5 - Drizzle
6 - Showers
7 - Rain
8 - Snow

Figure 7—Data sheet for recording information from time-constrained surveys. This sheet should be on waterproof paper.

The following data items (13-17) are recorded most accurately from anesthetized animals in the laboratory. Animals should be placed individually in plastic bags with the specimen number (item 9) so that the data can be properly recorded. If animals are released after collecting, these data can still be recorded; they will have slightly lower accuracy.

13. Sex. M = male; F = female; if unknown, leave blank.
14. Age. A = adult, J = juvenile.
15. Snout-vent length. Record to the nearest 0.1 millimeter.
16. Total length. Record to the nearest 0.1 millimeter. If the tail has been broken or is otherwise incomplete, leave this blank.
17. Mass. Record to the nearest 0.1 gram.

Items 18-21 are recorded in the field.

18. Vertical position. Use the codes at the bottom of the data sheet.
19. Tree species. The four-digit code for the species (if known) of the snag or log.
20. Decay class. Use the code for either snags or logs, as appropriate.
21. Cover-object dimensions. Record length and width to the nearest centimeter.

Data sheet for surveys of CWD-This data sheet (fig. 8) also needs to be on waterproof paper. Data for the specimens collected at each log are recorded directly below the data for the log. At least 15 data sheets will be needed per study area. As with the data for TCS, there is room for five animals per log. If the number of animals captured exceeds the space available, then follow the same procedures as for TCS. Data items are explained below:

1. Standard header. The first two lines at the top of the page are the same as for TCS. The following items (2-13) are data collected on each log before it is searched for animals.
2. Log number. Number logs sequentially from the start of each survey.
3. Time. Record the number of minutes required to search the log (20 staff minutes, maximum).
4. Decay class. Use the five-class scale. Other decay categories can be assigned during data analysis.
5. Tree species.
6. Aspect.
7. Slope. Record the percent slope over a 10-m run, with the log at the midpoint.
8. Total log: length. Record to the nearest meter.
9. Total log: maximum diameter (cm).
10. Total log: minimum diameter (cm).
11. Portion sampled: length (m). Record the amount of the log that was actually searched.
12. Portion sampled: maximum diameter (cm).
13. Portion sampled: minimum diameter (cm).

LOG SURVEY DATA

technique	province	habitat	stand #	day	month	year	wr	temp	start time	end time	crew	recorder
L G S												

log #	time	decay class	tree species	aspect	% slope	total log: length (m)	total log: max diam (cm)	total log: min diam (cm)	portion sampled: length (m)	portion sampled: max diam (cm)	portion sampled: min diam (cm)
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specimen number	catalog number	species	position	depth in log	sex	snout-vent length (mm)	total length (mm)	mass (g)
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log #	time	decay class	tree species	aspect	% slope	total log: length (m)	total log: max diam (cm)	total log: min diam (cm)	portion sampled: length (m)	portion sampled: max diam (cm)	portion sampled: min diam (cm)
						.			.		

specimen number	catalog number	species	position	depth in log	sex	snout-vent length (mm)	total length (mm)	mass (g)
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POSITION: 1 - Under Bark (on log); 2 - Under Bark (on ground); 3 - Under Log; 4 - In Log; 5 - Other

Figure 8—Data sheet for recording information from surveys of coarse woody debris. This sheet should be on waterproof paper.

The following items are collected for each animal encountered. Most are the same as for TCS and may be recorded in the field or in the lab, if all the animals are retained. Data unique to log surveys that are recorded in the field are:

14. Position (POS). Use the codes at the bottom of the data sheet.
15. Depth in log (cm). Record the distance to the exterior surface of the log.

Pitfall trapping data sheet-These data (fig. 9) are recorded in the lab when the animals are processed. A waterproof sheet is not necessary. Use a new data sheet for each time the traps are checked. Most of the data are the same as those collected for TCS and surveys of CWD. Unique elements are:

1. Trap night. Record the number of nights since the traps were opened; for example, if the traps were opened October 1, and these are data for animals picked up when the traps were checked on October 18, then trap nights are 17.
2. Trap number. Record the trap position (column and row) for each animal.

PITFALL DATA

technique	province	stand #	habitat	day	month	year	trap night	catalog initials
P I T								

catalog number	species	trap col	trap row	snout-vent length (mm)	total length (mm)	mass (g)	sex	age
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TRAP NIGHT: Number of nights since the traps were opened.
SEX: Male, Female (if unknown, leave blank)
AGE: Adult, Subadult, Juvenile

Figure 9—Data sheet for recording information on animals collected in pitfall traps. This sheet does not need to be on waterproof paper.

Appendix 3

Materials Needed for TCS or Surveys of CWD

Item	Number
Potato rakes (a backup rake is not a bad idea)	2
Crowbar	1
Stopwatch	1
Clipboard	1
Thermometer	1
Plastic bags	several
Cloth bags or pillowcases	1 or2
Pencils	2+
Compass	1
Clinometer	1
Short (15 cm) plastic ruler	1
Long (30 cm) plastic ruler	1
10-m measuring tape	1

Materials Needed for Pitfall Installation and Operation

Item	Number
Installation	
Posthole digger (l/person)	l+
15-m tape or measured nylon rope	2+
Plastic flagging (1 roll/pair of people)	l+
Waterproof ink marker (l/person)	l+
Number 10 tin cans	72/grid, or 24/array
l-lb margarine tubs	36/grid, of 12/array
Wood covers	36/grid, or 12/array
Operation	
Waterproof notebook and paper (l/person)	l+
6- by 10-inch plastic bags	Many
12- by 16-inch plastic bags	Many
Plastic cup or long handled spoon (l/person)	l+
Small cooler with reusable refrigerant	l+

Materials Needed in the Lab

Calipers
Plastic ruler (30 cm)
Spring scales (10 g, 50 g, 100 g)
Scissors
Forceps
Tags with preprinted catalog numbers
Paper towels (industrial type)
Plastic trays with lids
Cloretone
Nembutol
Formalin (40 percent formaldehyde solution)
95 percent ethanol
Isopropyl alcohol (dilute to 50 percent)
Jars for specimen storage