Field methods for rodent studies in Asia and the Indo-Pacific

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ACIAR MONOGRAPH SERIES

This peer-reviewed series contains results of original research supported by ACIAR, or deemed relevant to ACIAR's research objectives. The series is distributed internationally, with an emphasis on developing countries.

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CHAPTER 1

Why study rodent populations?

Introduction

Rodents are a dominant group of mammals. There are more than 2700 species of rodents worldwide; in fact, 42% of all the mammal species on Earth are rodents. Two-thirds of living rodent species belong to just one family, the Muridae, and most of the rodents found in Asia, both pests and non-pests, also belong to this family.

Rodents occupy a wide range of natural habitats, including forests and grasslands, as well as the human world of agricultural landscapes, villages and townships. Most rodents are prolific breeders and they often represent a significant amount of the animal biomass in forests and other natural ecosystems. As such, they play an important role in the food web, both as consumers of plants and fungi, and as a food resource for many of the larger predators. They are also important environmental engineers, helping to spread pollen and seed, aerating the soil through their digging and burrowing activities, and in extreme cases (e.g. beavers), changing the whole nature of the landscape. These ecological benefits are sometimes called 'ecosystem services'.

A relatively small number of rodent species have adapted successfully to the human environment of gardens, fields, villages and towns. Unfortunately, the people who created this environment generally view the successful rodents in a different light. Indeed, in almost all societies, the rodent species found around houses and in fields are viewed as 'pests' or even as 'vermin'. And often with just cause—the rodents consume and spoil crops in the field and in storage bins, they damage household possessions and even buildings and roads, and they play an often overlooked but highly significant role in the transmission of various diseases.

Rodents as pest species

Rodents affect rural families in three main ways: they eat agricultural crops in the field; they eat, spoil and contaminate stored food; and they carry diseases of humans and their livestock. In the Asia–Pacific region, rodents are one of the most important constraints to agricultural production. This region contains two-thirds of the World's poor—approximately 800 million people in 2001—and the majority of these people live in rural areas. Management of rodent pests in agricultural regions is therefore a high priority for reducing poverty.

The losses caused by rodents to rice crops in Asia provide a graphic example of their impact. Rodents typically cause annual preharvest losses to rice of between 5% and 10% of production. However, in some areas, episodic outbreaks of rodents cause heavier losses or even the complete destruction of crops. Postharvest losses in some areas may match or exceed the preharvest damage, and reports of 20% losses caused by rodents to grain after harvest are not unusual. Some 90% of the world's rice is grown and consumed in Asia. If we were able to reduce rodent losses by only 5%, then there would be enough rice to feed the population of Indonesia for one year (210 million people who rely on rice providing 65% of their daily calories)!

Rodents as beneficial species

For decades, the literature on integrated pest management of insects has emphasised that not all insects are pests. Indeed, there has been much scientific effort in identifying non-pest species and those that are described as 'beneficial' insects because they provide benefit through preying upon, or competing with, pest species of insects, or play a significant role in the pollination of crop and other plant species. We have reviewed the available literature on rodents and found that for any particular region, only 5–10% of rodent species are major agricultural pests (Table 1.1). Hence, rather than developing general methods that will control

most rodent populations, we should try to minimise the effect of control on species of rodents that are not pests. Indeed, the conservation of non-pest species of rodents should always be of concern in any control program. To illustrate this issue, a rare species of tree rat (*Chiromyscus chiropus*; Fea's tree rat) is sometimes captured at the edge of upland rice fields in Laos (Lao People's Democratic Republic). If farmers conduct non-specific rodent control around the rice fields, then these animals may be affected.

The importance of conserving non-pest species of rodents is not an easy concept to promote in developing countries. Many farmers have a long cultural tradition of battling the depredations of

rodents; it is understandable if from their perspective 'the only good rat is a dead rat'. We may be able to change this perspective, but to do so will require some very clear examples of the benefits that non-pest rodent species provide.

The high diversity of rodent species in many agro-ecosystems may also provide an opportunity to identify species that can indicate whether the ecosystem is in poor condition (degraded landscape) or in good condition (sustainable production is likely). Such species are known as 'indicator species'. The indicator species concept has been widely adopted using certain bird species as a measure of the health of a landscape. In agricultural landscapes,

Table 1.1 The number of species belonging to the Order Rodentia in various geographical regions that are considered significant pests of agriculture, and those whose conservation status is of concern (endangered, critical or vulnerable) or insufficient is known to assess the risk. The conservation status data are from the International Union for Conservation of Nature and Natural Resources (IUCN) website (<http://www.redlist.org/>). (Based on Singleton et al. 2003a.)

Continent or country	Number of rodent species	No. of rodent species that damage crops	No. of significant pest species in cropping systems	Conservation status	
				No. of species at risk	Little known
Africa	381	77	12–20	60	11
Australia	67	7	4	14	1
Europe	61	16	5	4	
India	128	18	12 (5 wide distribution 7 restricted distribution)	21	1
Indonesia (not incl. Papua)	164	25 +	13	11 +	28 +
Laos	53	12 +	4–8	4	14 +
New Guinea (PNG + Papua; not incl. Island Melanesia)	73	10 +	6	0	9 +

rodents and other more sedentary animals may be better indicators of environmental health at a local to regional scale.

Ecologically based rodent management

Ecologically based management of rodent pests is a concept that has developed a strong following in developing countries since the late 1990s. The concept aims to combine basic and applied research on rodents through focusing on the population ecology of rodents and developing management directed at the agro-ecosystem level. The concept is appealing because it promotes actions that facilitate sustainable agriculture and have minimum environmental impact. However, developing an effective integrated management plan requires a good understanding of the basic ecology of individual rodent pest species. This in turn is dependent on access to field methodologies that enable us to understand the population dynamics and field ecology of rodents.

In our experience, the process of developing effective, ecologically based rodent management is a learning cycle that involves phases of observation, formulation and testing of hypotheses, and further observation or experimentation, with each round of activities leading to better understanding. This flexible and responsive process is appropriate to the complex nature of the ecological problems that we face in dealing with rodent pests, and to the equally complex socioeconomic context presented by the diverse political and cultural systems of the Asia–Pacific region.

Despite the cyclic nature of the learning process, we believe that it is useful to distinguish three distinct phases in any investigation of rodent problems. These phases, described below, can provide a useful framework for designing a long-term rodent management study, or as a means of assessing the current state of knowledge for any given region. Indeed, a good way to begin is to ask the question, Where do we currently fall in relation to the three phases?

Phase 1: problem definition

Although rodents are frequently mentioned as a major cause of damage to both field crops and stored foodstuffs, there is often little in the way of hard data on crop losses or on other economic or social impacts. Rodent control activities always cost money and time, so before launching into any kind of control activity, it is a good idea to first define the scale of the problem. This usually involves the following steps:
* confirming that rodents are genuinely the cause of the problem
* identifying the species of rodents involved
* estimating the amount of damage to field crops and stored food.

Identifying the major rodent pest species is a useful part of problem definition because it allows the researcher to make use of the results of prior ecological studies and to learn from previous attempts to control the same species. For example, finding that *Rattus rattus* is the major field pest in an area would immediately alert the fieldworker to the likelihood that this highly adaptable species will need to be controlled in all local habitats, including around human habitation.

A preliminary assessment of health issues, perhaps based on local clinic or hospital records and some focus group meetings, might also be informative at this stage.

The problem definition phase might also be called the 'question definition' phase, for it is during this period that we should be trying to identify the key factors that influence rodent numbers and activity, and the level of risk that they pose to crops, stored food and human health. Such questions might be, Are we dealing with a localised problem or one that occurs over large areas? Do rodents cause substantial losses every year (chronic problem) or is the damage much heavier in some years than others (episodic acute problem)? Are periods of high crop damage due to increases in rodent numbers or due to a shift in the focus of their activities? If the former, is the

population increase due to rapid breeding within the fields at certain times of year, or is it due to migration of rodents from other habitats? Issues of this kind are fundamental to the design and implementation of ecologically based rodent management—where the goal is to manipulate the ecological system in ways that reduce the opportunities for rodents and thus improve human livelihoods.

Other important questions might relate to the history of rodent problems for a particular region: Have rodents always damaged crops in the area, or have their impacts increased in recent years? What changes in land use or cropping systems might have taken place at the same time?

Local knowledge is, of course, fundamental to framing many of these questions. Although some information might be contained in reports or other documentary sources, the richest and most direct source of information on the scale and extent of the problem invariably comes from members of the farming community itself. Various methods can be used to gain access to this wealth of information, many of them drawn from the realm of farmer participatory research (see Chapter 10).

Phase 2: ecological and historical studies

During this phase, we try to find answers to particular questions or test particular hypotheses that we identified during phase 1. In many cases, this means carrying out basic ecological studies on: changes in population size; the timing and location of breeding activity; patterns of habitat use and movement; and the timing and pattern of damage within both the cropping systems and the habitation areas.

An important part of ecological research is to decide upon an appropriate spatial and temporal scale for the studies (see Chapter 2). How large an area do we need to study and how long does our study need to last? These are particularly important questions where the primary objective is to develop options for ecologically based rodent pest management. This is because rodent management actions generally will need to be implemented over large areas and in a coordinated and sustained fashion if they are to be effective.

Before starting any ecological studies, it is sensible to learn as much as possible from any previous studies of the same species or similar cropping systems. Much of the information currently available is summarised in Chapter 11 for the major pest species, with the relevant literature sources provided at the end of each species account. Where basic biological

information is known for a particular species from earlier studies (e.g. average litter size, preferred location of nesting sites), it may be sufficient to do a small study only—just enough to test whether the species has a similar basic biology in your local population. This book contains information on many of the basic field techniques required to carry out ecological studies of this kind.

To answer historical questions, it is sometimes possible to obtain information from written sources such as agricultural records of crop production or pest problems. In some countries, these records are detailed and extensive, and span many decades. These can provide valuable insights into the history of rodent problems and it is usually worthwhile investing some time and effort into extracting the useful information. For many areas, records of this kind do not exist. In such situations, it may be possible to piece together a history of the rodent problem by conducting interviews with farmers and extension personnel. While gathering this information, we would also recommend asking questions about changes in cropping patterns and rodent management methods (e.g. poison use), and in general lifestyle factors such as the size and location of villages. By building up an overall picture of the historical changes, it may be possible to identify some of the key factors that have led to increased rodent problems—and hopefully then use these insights to reverse the trend.

Phase 3: designing and testing management options

Options for the management of rodent pests in any particular agro-ecosystem should develop in the first instance out of the improved ecological knowledge of the system. However, this knowledge in itself may not be a sufficient basis for designing management options. The other essential component is an understanding of what we might term 'the human factor'.

The human factor has many dimensions, including diverse cultural beliefs relating to rodents and the wider environment, variable systems of social organisation that influence the willingness or ability of people to work together in particular ways, and complex economic considerations that determine local priorities for allocating money and labour. It is also expressed at a variety of scales, from individual differences between members of one community, to more structured variations based on factors such as gender and wealth.

The complex interaction of ecological, cultural, social and economic factors needs to be given careful consideration when designing rodent management options. This is particularly so in areas where the agricultural community consists of smallholder farmers who are perhaps more used to making individual decisions and less familiar with the concept of broad-scale and coordinated actions.

The issue of sustainability is also vitally important. Because it is rarely, if ever, possible to completely eradicate a rodent pest (except perhaps from small islands), a lapse in management actions, even for a short period, may lead to a rapid resurgence of rodent populations and associated problems. In most situations, a high level of ongoing community commitment and involvement is therefore fundamental to effective pest rodent management.

The most direct way to find management options that may be appropriate for any given location is to adopt a participatory approach at all stages of project design and implementation. This involves working closely with communities that are representative of the potential long-term users of the management options. Once we have identified some management options that are ecologically appropriate, culturally acceptable, and both socially and economically sustainable, we then need to perform further tests to see how well they will perform in the real world. In many cases, their performance will need to be judged against a range of criteria, including their immediate economic benefit, their social implications, and their longer-term environmental impact. Some of these parameters may be difficult to measure; hence wide community consultation may be needed to gain a comprehensive and balanced view of how a particular management strategy is likely to perform in the longer term.

Despite these complexities, whenever we test a management option, we need to keep in mind that we are conducting an experiment. This is a critically important point. Field or village-level trials that are not conducted according to the principles of experimental design very often fail to deliver any truly interpretable results. This is not to say that an experimental approach will automatically guarantee good management options. Rather, good experimental design should allow a researcher or manager to understand why a particular management option has failed, and to design new trials or experiments accordingly, thus continuing the cycle of learning.

Purpose and scope of this book

We have written this book as a resource for anyone who is intending to conduct field studies of rodents in Asia or the Pacific. However, given the current, strong interest in reducing the impact of rodent pests on rural livelihoods across the region, we expect that the majority of users of this book will be agricultural scientists, extension personnel and students working in the context of management projects. For this reason, we will focus on methods that are appropriate for the study of 'pest' rodents and of the damage to crops that they cause. Nevertheless, many of the same methods would be appropriate for

the study of forest rodents (and with some minor adaptation, other small mammals) and in different geographical regions.

Wherever possible, we have avoided the use of specialised ecological and anatomical terminology; a glossary is provided at the end of the book to explain the technical terms that are used. Throughout the text we use scientific names rather than 'common' names for the main rodent pests. The reasons for this are explained in Chapter 4, and we encourage all users to become familiar with the scientific names of at least the main pest species in their area.

The methods that we describe in this book are ones that we have found especially useful in studies of pest rodents in Australia, Bangladesh, Indonesia, Laos and Vietnam. The coverage is by no means exhaustive and we freely acknowledge that there are many alternatives to the methods presented here. While we do not wish to be prescriptive, we do believe that there are advantages to be gained by other researchers adopting the methods

recommended here, at least as a basic set. Most importantly, the use of common methods will facilitate the rapid growth of ecological data for the main pest rodents of the Asia–Pacific region. This will hopefully reduce the need to acquire basic ecological data in each new study area, and will also allow everyone involved in ecologically based rodent management to learn directly from each other's experiences. Rapid advances in this field will depend to a large degree upon the free sharing of information, experiences and ideas.

Further reading

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Design of field studies

Introduction

Field studies of rodents can be highly rewarding but also immensely time-consuming. Many species are difficult to catch and it is often necessary to set large numbers of traps over many months before any general pattern will emerge. Environmental data, such as measurements of crop damage caused by rodents, can be obtained much more easily, but fieldwork and subsequent analysis of the large datasets are also very time-consuming. Before we start any field activities, we need to be sure that our work will not only be done accurately and precisely, but also that the activities fit into a framework with a good experimental design. The aim of this chapter is to explain and illustrate some of the general principles of ecological experimental design for field studies on rodents.

General principles of experimental design

Experimental design is a term describing the logical structure of an experiment. An **experiment** is an attempt to test a **hypothesis**—an explanation for one or more observations made in the field or laboratory (see below). Rodent ecologists typically make many different kinds of observations and they frame many different kinds of hypotheses. Throughout this chapter, we use two hypotheses to illustrate our key points. These are:

+ hypothesis 1—rice-field rats are more abundant in fields near refuge habitat, such as a large canal
+ hypothesis 2—providing barn owl nest boxes will reduce rat damage to paddy rice.

These examples illustrate that there are two broad types of experiments—mensurative and manipulative.

+ **Mensurative experiments** involve making some measurements of rodents and their habitat. The ecologist does not take any specific action against the rodents but measures what currently happens under current conditions. For example, to test hypothesis 1, we could measure the abundance of rats in fields near canals and in fields more distant from canals.

+ **Manipulative experiments** involve taking some action either directly against the rodents or that somehow modifies their habitat. At least two sets of plots or manipulations are required. For example, to test hypothesis 2, we might 'treat' four fields by installing barn owl nest boxes and leave four similar fields without nest boxes as 'controls' (see below).

Both kinds of experiments share many properties and require that certain essential design features are met. The most important of these are:

- identification of the key factors under investigation
- use of experimental units of an appropriate size and duration
- inclusion of a baseline or control to distinguish non-random from random events
- replication to estimate causal linkages and experimental error
- randomisation and interspersion to avoid bias.

Identification of hypotheses and key factors

As a field biologist, you will start making observations from the very first day of a new project. These observations will lead to ideas about how the various rodent species are distributed across the various local habitats, how the rodent populations are likely to respond to the changes in food availability through the natural and agricultural cycles, and how the different species will respond to possible management options. As the body of observations and information grows, each of these ideas will develop in substance and sophistication. At an early stage in a new project, it is a good idea to write out a number of general hypotheses about the position and role of rodents in the local environment.

Each of these hypotheses will probably lead to a number of more specific hypotheses that can serve as the basis for an experimental design.

A **hypothesis** is distinguished from a simple observation in various ways. One distinguishing feature is that a hypothesis can be tested by further observations or by an experiment. This means that it is capable of either being supported or proven incorrect by further observation. Testing of a hypothesis often leads to a refinement of ideas and a new hypothesis that incorporates the new evidence and insights.

A clearly stated hypothesis will include mention of one or more **key factors**. Using the two examples introduced above, hypothesis 1—rats are more abundant in fields near canals—identifies *distance to a canal* as a potential key factor in determining the local abundance of rats in any given field. As indicated above, an obvious way to test this is to compare rat numbers in fields located at different distances from a canal.

Hypothesis 2—owls reduce rat damage—identifies *the presence of owls* as a potential key factor in controlling rat damage in rice fields, although in this case, it does not specify whether this is because owls will reduce rat numbers or because they will modify rat behaviour in some way that makes them less likely to damage rice. This hypothesis might also be made more explicit by specifying that the number

of owls might be important, rather than just their presence or absence.

In general, the more explicit we can make our hypotheses, the more likely we are to have good experimental design and ultimately come up with satisfactory answers.

Size of experimental units

The concept of an **experimental unit** is critical for understanding the design of all ecological experiments because it determines the scale of the study. An experimental unit is defined as *the smallest division of the experimental material such that any two units may receive different treatments.*

Before defining the experimental unit for your study, it is necessary to think very carefully about the biology of the situation. In the case of the owl example, if our hypothesis is that the *presence* of an owl will reduce crop damage, then clearly the experimental unit cannot be any smaller than the area hunted over by an individual owl. However, if our hypothesis is that the *abundance* of owls will influence the *intensity* of crop damage, then the experimental unit for a mensurative experiment could be smaller than one owl's hunting range, assuming that the ranges overlap and that we can measure differences in owl abundance between locations. For experiments that involve agricultural damage, the size

of the experimental unit will often be determined by the size of the average crop field or plot.

If the owl experiment is manipulative, as suggested by the example of installing nest boxes in some fields but not others, then the experimental unit will be the area influenced by the installation of nest boxes. If the nest boxes are spread evenly through an entire 10 ha area of rice paddy, bounded by non-paddy habitat, then the experimental unit will be the 10 ha area. However, if the 10 ha area of paddy is surrounded by other paddy fields, the experimental unit will extend beyond the 10 ha in which nest boxes are installed, out to some point where the influence of the increased number of owls is no longer felt. Judgment is very important in deciding on the size of the experimental units and, wherever possible, this judgment should be based on sound biological knowledge or, in the absence of biological information, on conservative estimates of critical parameters (such as how far owls might fly). Many ecological experiments have suffered from using too small experimental units. In particular, rodent management experiments will often need to use large experimental units if they are to demonstrate differences in crop protection. Rats, like owls, often move much larger distances than you might think when they are searching for food or a mate.

Experimental units can also be too large or, more commonly perhaps, they can be located too far apart. The key problem here is that the experimental units should be as similar to each other as possible. Typical problems that might come from having overly large or widely spaced experimental units might be differences in soil types or hydrology, or differences in the variety of crops planted or in their time of planting. Uncontrolled sources of variation in an experiment may seriously reduce our ability to identify the role of the key factor or factors.

Duration of an experiment

Experiments need to be run over appropriate time periods. In testing hypothesis 1, measurements of rat abundance at various distances from a canal should probably be taken over an entire 12-month period. Most rodent populations undergo marked seasonal fluctuations in abundance and it is likely that any differences in abundance would be expressed at certain times of year but not at others. In almost any study of rodent ecology, once-off measurements may produce a result but they are unlikely to produce any real, meaningful insights.

Rodent researchers involved in management studies often attempt to determine the impact of a specific 'treatment' applied to a population. A simple illustration of why it is important to think about the duration of such an experiment before you begin is shown in Figure 2.1. Suppose that you are the manager of a rice farm and you wish to determine if adding barn owl nest boxes on the farm will reduce the abundance of rats. If you do a single measurement before and after the addition of nest boxes, you might observe the data shown in Figure 2.1a. These results by themselves might encourage
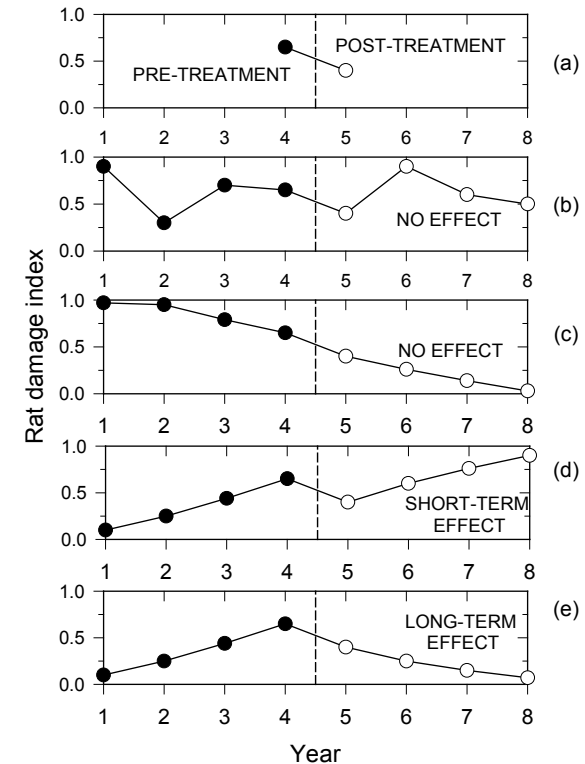
Figure 2.1 Why ecologists need to think about experimental design in field experiments. A manipulation such as putting up owl nest boxes is carried out between years 4 and 5 (dashed line): (a) a single observation before and after with no control—this result is impossible to interpret; (b) to (e) illustrate four possible scenarios if additional data before and after the manipulation are available.

you to jump to the conclusion that the treatment reduces rat damage. But by collecting data for a longer period, both before and after the addition of nest boxes, you would be in a much stronger position to draw the correct inference. As illustrated in Figure 2.1b–e, you might observe no effect, a temporary effect, or a long-term effect of the manipulation.

Inclusion of controls

The need for a 'control' is a general rule of all scientific experimentation. Quite simply, if a control is not present, it is impossible to conclude anything definite about an experiment.[1] For manipulative experiments, such as the owl experiment, a **control** is defined as *an experimental unit that has been given no treatment* (an unmanipulated site). For mensurative experiments, a control is defined *as the baseline against which the other situations are to be compared*. For the canal experiment, the baseline situation would come from fields that are so distant from a canal that the canal has no influence on the rats. Again, sound judgment is needed in such cases as to what distance from the key factor is far enough away. In this case, the relevant biological parameters are the distance that individual rats might move from the canal, the total distance that one season's progeny from canal-

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[1] In some experiments, two or more treatments (like fertilisers) are applied to determine which one is best. Unless an unfertilised control is included, this experiment will not allow you to say whether either treatment would give a better outcome than using no fertiliser at all.

dwelling rats might disperse, and the distance away from the canal that any 'knock-on' or 'ripple' effect might be felt (e.g. through displacement of other individuals).

For the owl nest box experiment, the control would be a nearby farm that is similar to the treated one but does not have any owl nest boxes added. If the treatment site showed a long-term effect of the kind shown in Figure 2.1e but the control site showed either no change in rat damage or only random change through the experimental period (e.g. Figure 2.1b), then the case for adding nest boxes would be even more compelling. However, in the event that both treated and control areas showed similar long-term patterns of change, then you would have to conclude that some other, entirely different factor was responsible for the observed changes. Changes in climatic conditions would be worth considering or perhaps changes in the abundance of some other predator.

Although the exact nature of the controls will depend on the hypothesis being tested, a general principle is that the control and the treatments should differ in only the key factor being studied. For example, if you wish to measure rat damage in paddies near to a canal and distant from a canal, you should use experimental units that are planted with the same variety of rice and that were planted at the same time. In ecological field experiments, there is often so much year-to-year variation in communities

and ecosystems that you should always do the entire experiment at the same time. You should not measure the controls in 2003 and the treatments in 2004, for example.

Replication

Replication means the *repetition* of the basic experiment. There are two reasons why experiments **must** be repeated and one other reason why it should be. The most important reason for replication is that any experimental outcome might be due to chance. Repeating the experiment will allow us to distinguish a chance or random outcome from a genuine or non-random outcome. The more times we repeat an experiment and observe the same or similar outcomes, the more certain we can be that our hypothesis has identified a genuine causal factor.

The second essential reason for repeating experiments is that replication provides an estimate of **experimental error**. This is a fundamental unit of measurement in all statistical analysis, including the assessment of statistical significance and the calculation of confidence limits. Increased replication is one way of increasing the precision of any experimental result in ecology.

In addition, replication is a type of insurance against the intrusion of unexpected events on ecological experiments. Such events are one of the major sources

of interference or 'noise' in field ecology. They are most troublesome when they impinge on one experimental unit and not on the others. As an example, let us assume in our study of rat numbers close to and distant from canals that we have three **replicates** (i.e. three fields close to the canal, three distant from the canal). During the course of our study, one of the plots close to the canal is accidentally flooded. The flooded site would be omitted from the final analysis, but because we have sufficient replication, we can still obtain meaningful results from the other sites.

These considerations mean that every experiment should be repeated at least once, giving two replicates. When this requirement is added to the need for a control or baseline, it is clear that field experiments should include at least two treatment areas and two control or baseline units. However, two is a minimum number of replicates and statistical power will increase if you have three replicates or more. Each additional replicate gives more statistical power to the experiment, but each replicate also represents an additional cost in terms of labour, resources etc.

The decision about how many replicates are needed is a fundamental one in experimental design. In essence, it can be seen as a trade-off between benefit and cost—the benefit of additional statistical power and confidence in the results, but gained at the cost of extra fieldwork, and extra data processing and analysis. Statisticians can advise you on optimal number of replicates for any given experiment, but

they will need to know many details concerning the cost of obtaining data, the likely sources of variation, and the risk of chance events (e.g. the flood example) intruding on your experiments.

Randomisation and interspersion

There are three main sources of variability that can cloud the interpretation of experimental results (Table 2.1). Some of these sources of confusion can be reduced by the use of controls, and by replication, as discussed already. However, two other important methods remain—these are called **randomisation** and **interspersion**.

Randomisation

One kind of randomisation involves the random selection of individuals from within a population of animals or of field plots from large areas of uniform habitat (e.g. for measurement of crop damage). A second kind involves the random allocation of experimental units to treatment or control categories. This second type is an important consideration in experimental design. Randomisation by categories insures against bias that can inadvertently invade an experiment if some subjective procedure is used to assign treatments and controls. Randomisation of treatments and controls also helps to ensure that

observations are independent—that what happens in any one of the experimental units does not affect what happens in the others. This is especially important where the data will be subject to statistical significance testing, because most such tests are invalid unless experimental units are independent.

In many ecological situations, complete randomisation is not possible. Study sites cannot be selected at random if not all land areas are available for ecological research. Within areas that are available, patterns of land ownership or access will often dictate the location of study sites. The rule of thumb to use is simply to *randomise whenever possible*. Where this is not possible, statistical tests should be applied with caution.

Table 2.1　Potential sources of error in an ecological experiment and features for minimising their effect.

Source of error	Features of an experimental design that reduce or eliminate error
Temporal changes	Treatments with a control or baseline
	'Before and after' experimental designs
Experimenter bias	Randomised assignment of experimental units to treatments
	'Blind' procedures[a]
Initial or inherent variability among experimental units	Replication of treatments
	Interspersion of treatments

[a]　A 'blind' procedure is one where the researcher is unaware of whether a particular test animal or site is part of a 'treatment' group or a 'control' group. This removes any possibility of bias in the experimental procedure. However, it is usually only possible in laboratory studies, such as in feeding trials.

Interspersion

Where should experimental and control plots be placed in relation to one another? This is a critical problem in field experiments, and the general principle is to avoid spatial segregation of treatment plots. Randomisation does not always ensure that experimental units are well interspersed; there is still a chance that all the treatments will be 'bunched'. Hence, after randomly assigning treatments, you should check that they have not been grouped by chance—for example, with all treatment plots north of a village and all control plots south of a village. Such a design would not be desirable if there is some kind of systematic differences between the sites, such as a soil nutrient or moisture gradient. **Interspersion** means getting a good spatial mixture of treatment and control sites. Avoiding bias of any kind is one of the main goals of good experimental design.

Summary

The general principles of experimental design are often overlooked in the rush to set up ecological experiments. The first step in designing a good experiment is to develop one or more testable *hypotheses*. Each hypothesis should clearly identify the *key processes* or *factors* under investigation and should also include a definition of appropriate *experimental units*. *Baselines* or *controls* need to be established for any measurement or treatment plot. *Replication* is needed to estimate experimental 'error', the measure of statistical significance. The experimental units must be sampled *randomly* to satisfy the assumption that all observations are independent and to reduce bias. Treatments and controls should be *interspersed* in space and in time to minimise the possibility that chance events will affect the results of the experiment. If interspersion is not used, replicates may not be independent and statistical tests will be invalid.

Checklist for experimental design
1. What is your hypothesis?
2. What are the experimental units?
3. What measurements or treatments will you undertake?
4. Have you established appropriate baselines or controls?
5. How many replicates of these units do you need?
6. Have you randomised your measurements or treatments?
7. Are your measurements or treatments segregated or interspersed?

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CHAPTER 3

Capture and handling of rodents

Introduction

Rodents are generally difficult to observe directly in the field. Most species are nocturnal in habit and they are often extremely wary of all potential predators, including humans. Under some circumstances, indirect signs of rodent activity, such as footprints, faeces or burrows, may provide a good measure of rodent numbers and activity patterns. However, methods of this kind will first need to be calibrated against more conventional measures of abundance and activity. All field studies of rodents thus begin with a phase of trapping, sampling and identification of the rodents themselves. In this chapter, we describe some basic methods for the capture and handling of rodents. Chapter 4 is devoted to the process of identifying captured rodents.

It is important to be aware that some countries have laws governing the capture and handling of wild animals. In some cases, these laws even cover introduced or pest animals. Depending on the country where the study is being undertaken, you may need to obtain permits before you start to trap animals. Furthermore, in some countries, you may need to obtain animal ethics approval for any study involving the capture and handling of live animals.

Capture methods

Human ingenuity has come up with many different ways of catching rodents. Many groups of people have developed specific traps and snares that either kill or capture any rodent that ventures too close. These are usually either set in a place that shows signs of regular rodent activity, such as across a

rodent pathway, or are baited with a substance that acts to attract rodents from the surrounding area. Sometimes traps are used in combination with low fences that guide the rodents towards the trap (e.g. Figure 3.1).

Figure 3.1   A traditional dead-fall trap set in a low fence in the uplands of Laos.

In many places, rodents are actively hunted. This is either done at night while the rodents are active, or during the day by digging into their burrow systems or flushing them from their hiding places. Dogs are often used to help locate rodents in their daytime retreats.

Poisoned baits are used extensively in many parts of the world. Use of baits is not considered here as a capture method because there is no certainty that any animals killed by poisons will be recovered. Nevertheless, rodents killed through the application of poisons should not be neglected as a possible source of biological information, especially during the early part of a study, when even the most basic questions may need to be answered (e.g. Which species are found in my study area? When do they breed?).

Major types of trap

The four main kinds of traps are:
* single-capture live-traps
* single-capture kill-traps and snares
* multiple-capture live-traps
* pitfall traps.

Any of these trap types can be used in combination with a drift fence that directs the rodents towards the trap. However, this method is most commonly used with multiple-capture live-traps and pitfall traps and is discussed under those headings.

Care should be taken to ensure that all traps are well maintained and set to optimum sensitivity. A poorly set trap is a waste of precious time and resources—and it will bias your trapping results. Whenever a trap is set for the first time in a trapping period, it should be test-fired to ensure that all parts are functioning correctly. If a trap fails to fire or seems insufficiently sensitive, it should be fixed on the spot if possible, or taken back to a workshop for repair.

Single-capture live-traps

There are two main types of single-capture live-traps: **cage-traps** made of open material such as wire mesh (Figure 3.2) or perforated sheet metal, and **box-traps** with fully enclosed sides. Box-traps offer protection for the captured animals and are favoured in many parts of the world, especially where overnight conditions are very cold or wet. Some box-trap designs are covered by patents—Longworth and Sherman traps are perhaps the best-known examples. Cage-traps are used more often in Asia. They are cheaper and simpler to make than box-traps, and they are often manufactured locally and sold in markets.

All single-capture live-traps work on the principle that an animal enters the trap and then releases a trigger which allows the door to close behind it. In some cases, the trigger is released when the animal pulls on a bait. In other variants, the trigger is released when the animal steps on a treadle.

Single-capture live-traps must be made of strong material and have reliable functioning components. The captured animal must not be able to break through the sides of the trap or push open the door once it has closed. The trap must be large enough and strong enough to comfortably hold the largest rodent that is likely to be caught. In most parts of South and Southeast Asia, this is probably an adult *Bandicota indica* (body weight of approximately 500–1000 g). We have captured this species in Vietnam in traps measuring approximately 400 × 150 × 150 m.

Figure 3.2 Metal, single-capture live-traps (cage-traps). Each trap has a door at one end with hinges at the top of the trap. The door can be locked open with a pin that connects to a trigger device holding some bait. When a rodent touches the bait, the pin holding the door open is released and a spring mechanism is used to close the door firmly.

Single-capture live-traps are always baited. The bait is either attached to the trigger device or placed behind the treadle. In either case, the bait should be firmly attached so that it cannot be easily stolen. Ideally, only one type of bait should be used in all traps. However, where the rodent community contains a range of species with different preferences, it may be necessary to use several different baits. These might be alternated between traps, or placed together in the same trap. The most important point is that the type of bait or combination of baits should not be altered during the course of a study, or it will be difficult to assess whether changes in capture rates are due to bait preference or to other factors. An experimental design for selecting suitable baits is discussed below.

Certain kinds of bait play a second role in that they provide food for captured animals to protect them from starvation or dehydration. This is particularly important in population studies where we must be careful that the period spent in the trap does not have any serious impact on the health of the individual. Where the primary bait will not satisfy the basic food and water requirements of the target species, you should consider whether or not to add some other moist food, such a block of cassava or sweet potato.

Traps are often set under cover, such as low vegetation or under a house. Where cage-traps are set in exposed positions, it may be necessary to provide some shade so that the animals do not become heat-stressed. This can be as simple as placing rice straw or large leaves on top of the trap.

Single-capture kill-traps or snares

These traps also work on a trigger mechanism, but they are designed to kill the rodent rather than catch it alive. Kill-traps offer a number of advantages, including the fact that they are often very cheap and readily available, allowing very large numbers to be set. In some circumstances, they also are more effective than live-traps. In many parts of Asia, locally produced snares made of bamboo or wire are highly effective in catching rodents, having been perfected over many generations of use.

Kill-traps are obviously only useful where the experimental design specifies that all captured animals will be sacrificed, such as for studies of diet and breeding activity. This is not the case in many ecological studies, where animals will be marked and released as a way of estimating population density or to study patterns of survival, habitat use and movement. Another disadvantage of using kill-traps is that the specimens are often damaged by the trap's mechanism or by ants.

Multiple-capture live-traps

A disadvantage of all single-capture live-traps is that once triggered (either with or without a successful capture), they are no longer effective. This can be a serious issue where rodent numbers are high relative to the number of traps, such that all available traps have caught a rodent early in the evening, or in situations where heavy rain or interference by other animals causes the triggers of many traps to be fired without capturing a rodent.

Multiple-capture live-traps are similar in general design to the single-capture models, but instead of having a trigger mechanism, they have a 'one-way' entrance that allows rodents in, but not out. The most common entrance of this kind is a funnel, as shown in Figure 3.3. However, a doorway that is opened by a treadle mechanism is also effective.

Figure 3.3   Multiple-capture live-trap with a cone-shaped funnel leading from the entrance of the trap.

There are several variations on the standard multiple-capture live-trap. One type, developed in Vietnam, is divided into two compartments by an internal partition, but joined by a second funnel. Captured

rats tend to move into the second compartment in their bid to escape. The rationale for this design is that rats may be deterred from entering the trap if any prior captives are moving around too close to the fence. Experimental results show a higher capture rate for the two-funnel version compared with the standard trap. Another variant on this concept includes a 'false wall' that stops rats from huddling against the fence.

As with single-capture live-traps, each multiple-capture live-trap should be provided with moist food, such as blocks of cassava or sweet potato. Provision of food will maintain captured animals in better health and may also provide further incentive for rats to enter the traps. Traps should be covered with rice straw or other loose vegetation to protect captured animals from the sun. In addition, a small amount of rice straw or similar material should be placed inside the traps. This will allow animals to hide and may reduce the chance of fighting between adults or between different species.

Trap–barrier systems

Multiple-capture live-traps are generally set at openings along a fence or 'barrier system' (Figure 3.4). When rodents encounter a barrier, instead of jumping or climbing over, most will run along it until they find a way through. Traps are usually placed opposite regularly spaced holes in the fence. The linear trap–barrier system (LTBS) has been used to

good effect in several field sites in Southeast Asia. Here, we describe the method as used in lowland rice fields in Java, Indonesia.

The LTBS was implemented in Indonesia after initial studies using single-capture live-traps, break-back traps and various designs of multiple-capture live-trap gave poor capture rates for the major rodent species, *Rattus argentiventer*. This species is often extremely abundant, but notoriously 'trap-shy'. Studies on the use of different bait types showed that choice of bait could increase the success of trapping, but only before the booting stage of the rice and after the harvest of rice crops. The reduced capture rate between these two stages was probably due to the general availability of high-quality food in the fields.

The LTBS has proven to be a successful alternative to conventional trapping for population studies in

Figure 3.4   A linear trap–barrier system with a multiple-capture live-trap, set through dense streamside vegetation in the uplands of Laos.

Indonesia. Placing a LTBS across the path of regular movements of rodents, such as between burrow sites and feeding areas, often leads to large numbers of rats being captured. Importantly, because the system does not depend on bait to lure rats into the trap, the effectiveness of LTBS is not influenced by the availability of alternative foods in the field.

The system used in Indonesia comprises eight multiple-capture live-traps set along a plastic barrier fence which is 180 m long (Figure 3.5). Alternate traps are set facing opposite directions and are spaced 20 m apart. The traps are checked early every morning. Other animals caught in the traps, such as lizards, frogs and snails, are either released or destroyed (e.g. pests such as the golden apple snail).
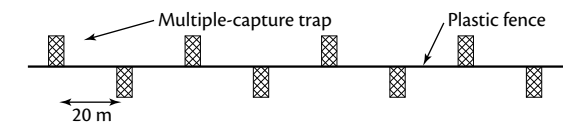
Figure 3.5   Layout of a linear trap–barrier system for trapping rats for population studies.

The multiple-capture live-traps used in Indonesia measure 600 × 240 × 240 mm. The funnel attached to the opening of the trap allows rats to enter but not to exit. A door at the other end of the trap allows access to captured rats. This door is held closed by a pin or wire. All components of the trap are checked to be in working order before each trap is set. After installation, the traps are loosely covered with rice straw to provide shelter from the sun for captured animals.

The fences are made from heavy-duty (woven) plastic sheeting approximately 500 mm high (Figure 3.6). The fence is supported by bamboo or wooden stakes every 1 m, and tension is provided by thick string running along the top of the fence. Holes are made in the fence at the appropriate spacing. Each trap is held tightly against its hole, so that rats cannot squeeze between the fence and the trap. Each trap is held in place with a stick or small piece of bamboo. The bottom of the fence is anchored by burying the base of the plastic in mud or soil, to stop the rats from digging underneath (Figure 3.7). This is easy to do in mud, but more difficult in dry ground.

LTBSs are particularly effective when set up in shallow water, such as in a flooded rice field. In this situation, the rats will be swimming along the fence in search of a way through. They can be encouraged to enter the traps by placing an entry ramp that leads up to the hole (Figure 3.8).

Regular maintenance of the fence is important for the success of the LTBS. Any holes chewed by rats should be quickly repaired or countered by the addition of another trap at the place of the hole. The fence must be kept vertical and taut, and grass or other vegetation must be kept clear of the fence. Construction of a LTBS represents a significant investment of time and resources, so it is important to keep it working at peak efficiency.

Pitfall traps

Pitfall traps work on the principle that animals will either fall or jump into a hole in the ground. Although this sounds unlikely, many animals have no concept of being unable to climb or jump out of a hole. If the pit is very deep, or if it has smooth

Figure 3.6   Plastic fence supported by bamboo posts.

Figure 3.7   Anchoring the fence by burying the bottom of the plastic in the mud.

or overhanging sides, captured animals will be unable to escape. A traditional variety of pitfall trap, with steeply overhanging sides, is used to catch rats in several regions of Southeast Asia. These traps are sometimes covered by a framework of interlaced sticks and a layer of straw. This apparently encourages rats to enter the structure and drop into the underlying pit. In other cases, the pit has sloping margins with a covering of loose sand or gravel that is said to cause the rats to slide into the pit.

Pitfall traps are used to great effect in ecological studies in many parts of the world. In most cases, they are used in combination with a plastic barrier of the kind already described for the trap–barrier system that leads the animals towards the pits. The pits most often consist of plastic buckets or short sections of polyvinyl chloride (PVC) piping set into the ground (Figure 3.9).

Figure 3.8   A small mound of mud at the entrance to a trap functions as an entry ramp.

Pitfall trapping is easiest in dry areas with sand or soft soil, as illustrated in Figure 3.9. It is less practical where the ground is waterlogged, because plastic buckets or piping either tend to float out of the ground or fill up with water. It is important to ensure that the captured animals do not drown, so it might be necessary to place a small piece of wood or polystyrene foam that will float if the trap is partially filled with water.

Figure 3.9   A polyvinyl chloride (PVC) pitfall trap used in combination with a fence made of light fly wire or plastic can be an effective way of catching small rodents.

Checking and cleaning traps

All types of traps should be checked and cleared of rats as early as possible each morning. This is most critical for population studies where it is assumed that the trapping method does not affect the animal's normal behaviour (see Chapter 5). However, it is important also for reasons of animal welfare and to ensure that

no harm comes to non-target captures, such as other small mammals, frogs or snakes.

In studies involving rodents, it is important to use water only when cleaning traps. Avoid using detergents because the odour may deter a rodent that might otherwise have entered the trap. After washing traps, mix them up with unwashed traps (interspersion!) so that any odours can be masked and to ensure that any impact is randomly distributed across the trapping grids.

Comparing trap and bait efficacy

Just about any combination of trap and bait will eventually catch some rodents. However, it is in your interest to maximise the capture rate. **Trap success**, defined as the number of rodents caught divided by the total number of traps set, is influenced by many factors. It will differ between trap types, depending on the behaviour of the rodent species in your area. It will also vary according to how well the traps were set, where they were set, the reliability of the trap mechanism, the age and sex composition of the population, and the weather conditions during the trapping period. Where baits are used as an attractant, trap success also will reflect the general availability of food in the vicinity of the traps. As noted earlier, single-capture traps may have an overall low success rate at times when abundant food is available in surrounding fields.

At the start of a new study, we recommend that you carry out a small trial to test the effectiveness of the available trap types to see which works best at your particular location. You might also test different baits at the same time, but it is important to remember not to make the experiment so complex that adequate replication is not achieved. You can try almost anything as bait, provided that it is attractive to the rodents, and it is also possible to use a combination of bait types. Some types of bait for rodents that have been used successfully in Southeast Asia are:

* vegetables (e.g. sweet potato, cassava)
* fruits (e.g. apple, banana)
* dried or cooked meat (e.g. crab, fish, snail)
* grain (e.g. wheat, rice), usually wrapped in a small piece of cloth or netting
* vegetable oil (coconut, peanut), soaked into cloth.

In studies where the captured animals will be marked and released, the bait will need to sustain the animal in good condition. Cloth soaked in vegetable oil would not be suitable in this case, but could still be used in combination with something less attractive.

Habitat surveys

During the problem definition phase of a study (see Chapter 1), you should set traps in positions that will maximise the chances of sampling the full local diversity of species and habitats in the study area. Set the traps directly alongside burrows or on obvious rodent pathways to maximise the capture rate.

After you have developed some preliminary ecological hypotheses, you need to carry out trapping in a systematic way to ensure that data are comparable between habitats and trapping periods. Systematic trapping is usually carried out on **trap-lines** or **trapping grids**. In both methods, the traps are set at equal spacing as a way of standardising the trapping effort per unit distance or area. The spacing of traps should reflect the expected size and abundance of the target species. Under most conditions, with expected rodent densities of tens to hundreds of animals per hectare, you would probably want to place your traps about 10–20 m apart. However, in some situations, you may want to place the traps closer together or further apart—an example would be if you are trapping specifically for a large, highly mobile species where each animal may occupy a territory of several hectares.

Trap-lines are usually set by walking through a habitat and placing traps after a standard distance (e.g. every 10 or 20 m). It is important to determine the number of paces per standard distance for each person involved in setting traps; for example, some people take 10 paces for 10 m, others take up to 15 paces. The course taken may be a straight line but it can also be a loop that ends back at the point of origin. Trapping grids are more structured arrangements, with traps set in parallel lines that ensure an even density of traps per unit area. Trapping grids also allow the population density to

be calculated, by multiplying the number of animals caught by the area trapped (see Chapter 5 for details).

The choice of whether to use trap-lines or trapping grids will be influenced by the diversity of habitat types available. If there is a uniform habitat type (e.g. large wheat fields), then grids may be appropriate. If a range of crops and other habitats are present (e.g. a mixture of rice fields with vegetable crops and villages—as found in many parts of Southeast Asia), then trap-lines are usually more appropriate. A combination of trap-lines and grids can be used, provided, of course, that the same method is used for each habitat and trapping period.

In village habitat, it may be impractical to set either trap-lines or trapping grids. An alternative is to set one or more traps per house, most often taking a random selection of houses.

Trapping effort and frequency

After deciding on whether to use trap-lines or trapping grids for a habitat survey, the next issues to think about are how many lines or grids should be set up per site, how many traps should be allocated to each unit, and for how many nights each trapping period should run. A good way to think about this is in terms of **trapping effort**.

Trapping effort is usually expressed as the number of effective **trap-nights**. In the simplest case, this is

calculated by multiplying the number of traps by the number of nights of trapping (e.g. 100 traps set for 4 consecutive nights = 400 trap-nights). However, traps that have been triggered without making a capture (sometimes called 'null traps') should really be subtracted from the total. In this case, total trapping effort is calculated as the sum of non-null traps for each night (e.g. 95 + 92 + 99 + 97 = 383 trap-nights).

Trapping effort can be increased either by increasing the number of traps or by trapping over a longer period. In theory, this means that a large number of traps could be set for only one night. However, there are good reasons to spread the trapping effort over a *minimum trapping period of three consecutive nights*. One reason is that variable weather conditions may mean that rodents are far more active on some nights than on others—an extended trapping period is obviously less likely to be affected by this kind of variation. But an even more important reason is that many rodents are **neophobic**, which means that they are naturally wary of any new object in their environment. Neophobia often results in low capture rates on the first night, followed by better results on the subsequent nights as animals lose their initial fear of the traps. In most of our studies, we have found a trapping period of 3–4 nights to be adequate. A good way to decide on the most cost-effective trapping period is to plot the capture rate for each day. If you see the capture rate start to decline then the trapping should be stopped. This will happen most often

where the captured animals are being killed, but it can also be due to learned avoidance of the traps by animals that have been captured once and released. Long periods of continuous trapping should also be avoided in some population studies because multiple captures can have an impact on the health of the animals (e.g. captures of pregnant or lactating females may affect survival).

To decide how many traps to set per line or grid, you should first work out how many traps can be set in total per site and how many consecutive nights of trapping can be done. These are often limited by very practical considerations including the budget available to purchase traps and the availability of people to check the traps. The total number of effective trap-nights should then be allocated across the different habitats selected for trapping. For example, with a total of 100 traps set over 5 nights (500 effective trap-nights), you could set up 10 trap-lines of 10 traps in each of 10 habitats (giving 50 trap-nights per habitat), or 5 trap-lines of 20 traps in a subset of five habitats (e.g. the most important ones; giving 100 trap-nights per habitat). The decision is obviously a trade-off between numbers of habitats and the intensity of sampling, i.e. more habitats but fewer traps in each, or fewer habitats but each with more traps. This is never an easy decision but a good way to start is to think about whether you are interested primarily in statistical testing of particular hypotheses or in getting a general overview of the ecological system.

Another factor that you should take into account when thinking about trapping effort is the abundance of the target animals. If they are likely to be very abundant and easily captured, such that almost every trap can be expected to catch a rat, then 10 traps per habitat, set over 3–4 nights, may be quite enough. However, because our experience in agricultural contexts in Asia suggests that capture rates of around 5–10% are more typical, we would recommend a minimum number of 20 traps per trap-line or grid, giving a trapping effort of 60–80 trap-nights per habitat. Other issues to do with the allocation of trapping effort are discussed in Chapter 5.

Where statistical power is critical, another factor to take into account is the need for replication of habitats. In particular, you should ask whether it is sufficient to replicate the most common habitats between two or more different localities (e.g. rice fields in each of two treatments and two control sites). Perhaps the habitat also should be replicated within each village? Remember, as a general rule of thumb, you should replicate the sampling of all experimental units (in this case, a specific habitat).

Trapping **frequency** will depend on the aims of the study. In many studies, trapping is carried out at regular intervals (e.g. every two weeks or once a month). More frequent trapping sessions will provide better data on population dynamics (e.g. survival of marked animals, changes in breeding condition) and may be especially valuable during the initial phase

of a new study, when basic ecological research is needed. However, as the dynamics of the ecosystem become better known, it may be appropriate to trap at specific periods in relation to the ecological cycles which, in agricultural landscapes, are often linked to the cropping cycles. For example, trapping may be timed for the period immediately before planting, before the reproductive phase of crop growth, just before harvest, and then during a fallow period when food is limited.

Handling a captive rodent

Safe handling methods are important both for captured rodents and for fieldworkers. Whatever methods are used, they should minimise stress to the animals and should also minimise the risk of injury or disease transmission to the handler. Handling live animals is normally only required for population studies where captured animals need to be examined closely to allow taxonomic identification, determination of age, sex and reproductive status, the taking of measurements, and the marking or tagging of an animal before release. Even very competent handlers should not handle captive animals any more than is absolutely necessary.

The first step in handling a captured rodent is to extract it from a trap. This is usually done by placing a cloth bag around the opening of the trap and waiting for the animal to move into the bag. Be patient:

shaking the trap usually just causes the animal to panic and generally does not speed up the process. Once the animal is in the bag, gently move it to a bottom corner and wait until its nose is in the corner of the bag. You can then hold the bag around the body with one hand, while your other hand enters the bag to take hold of the body. Alternatively, hold the animal within the bag, then peel away the bag to expose parts of the animal for marking, measuring or assessment.

There are various techniques that can be used to hold an animal directly. Different methods are appropriate for smaller or larger animals. Whatever technique you use, take care not to hold the animal too tightly and to allow the animal to breathe easily. For a rat-sized animal, we recommend the following technique: place your first and second fingers on either side of the animal's head, creating a firm hold of the head (Figure 3.10). Ensure that there is no undue pressure from your fingers on the skull and that your fingers are not on the animal's neck, as this will cause suffocation. Hold the body gently with your thumb and remaining two fingers. An alternative method for rat-sized animals is to place your first finger on top of the animal's skull, between the ears and position your second finger and thumb on either side of the head. Hold the body with your third and fourth fingers.

For smaller animals (juvenile rats and mouse-sized rodents), it is usually possible to 'scruff' the animal by gently pinching the loose skin along the back of the

neck and upper back between the thumb and first finger.

The grip shown in Figure 3.10 is still suitable for a very large rodent, such as an adult *Bandicota indica*,

Figure 3.10   The recommended method of holding a rat.

but it may be necessary for a second person to control the hind-limbs (and their claws).

Whatever method you are using, take the initial grip inside the confines of the bag. When your hold is comfortable, peel the cloth bag away to expose the animal. If it struggles and your hold is no longer secure, put the animal back in the bag, have a short break and start again.

An alternative to free handling methods is to use a specially designed, funnel-shaped observation bag. This has straps along the length of the bag that can be tightened to restrict the animal's movements. Mesh along the underside of the bag allows the researcher to sex the animal and take basic external measurements such as body and tail lengths.

Methods of euthanasia

Some studies require the humane killing or **euthanasia** of captured animals. This may be necessary to obtain reference specimens for taxonomic studies, to obtain detailed information on breeding activity or diet, or for parasite and other disease studies. Our general objective when euthanasing animals should be to deliver a rapid death with minimal distress and a rapid loss of consciousness before death. A number of standard techniques are available but their appropriateness depends on the experience of the field personnel

and the equipment available. See Further reading for sources of information on a variety of methods.

Asphyxiation

Asphyxiation methods have many advantages. They generally result in rapid death and do not require any direct handling of the animals. Provided a large enough container or bag is available, multiple animals can be killed simultaneously. The two most commonly used methods involve carbon dioxide or carbon monoxide.

Using carbon dioxide

This is probably the best method for euthanasia as it leads to rapid death and poses no threat to people. Carbon dioxide (CO_2) gas cylinders are typically

fitted with valves and a pressure gauge (Figure 3.11). The gas is fed by hose into a sealed chamber such as a plastic bucket with a close-fitting lid. Two small holes should be cut in the lid, one for the gas hose and the other to release excess air. Because CO_2 gas is heavier than air, once the chamber is filled, excess gas will spill onto the ground and disperse. Do not use this method in a tightly closed room.

Procedure

+ Before putting the animal in, pre-charge the chamber with CO_2 for 30 seconds. The pressure dial on the regulator should read no higher than 138 kPa (20 psi). Close the adjustment valve.
+ Place the animal in the chamber and close the lid. The animal can be still inside a bag or even in a cage.
+ After 1–2 minutes, check the animal briefly. At this stage, it should be losing balance or becoming sleepy. Open the adjustment valve again for 1 minute to replenish the CO_2.
+ After approximately 3–5 minutes, check the animal again for any signs of life. The eyes should be fixed and dilated.

The animal is not dead if:
+ its heart is still beating—check this by feeling the chest between your thumb and forefinger
+ it blinks when you touch its eyeball.

Pressurised CO_2 gas is available in most countries. However, the large size of most CO_2 cylinders makes this method most useful in a laboratory setting and generally impractical in the field.

Using carbon monoxide

Vehicle exhaust fumes contain carbon monoxide (CO) and this can be used to euthanase animals where CO_2 is not available. (However, for safety reasons, we strongly recommend the use of carbon dioxide wherever possible.) The basic method is similar to that described above for CO_2 gas, except that the source is a running vehicle (car or motorbike) that runs on petrol. A diesel-powered vehicle is *not* suitable.
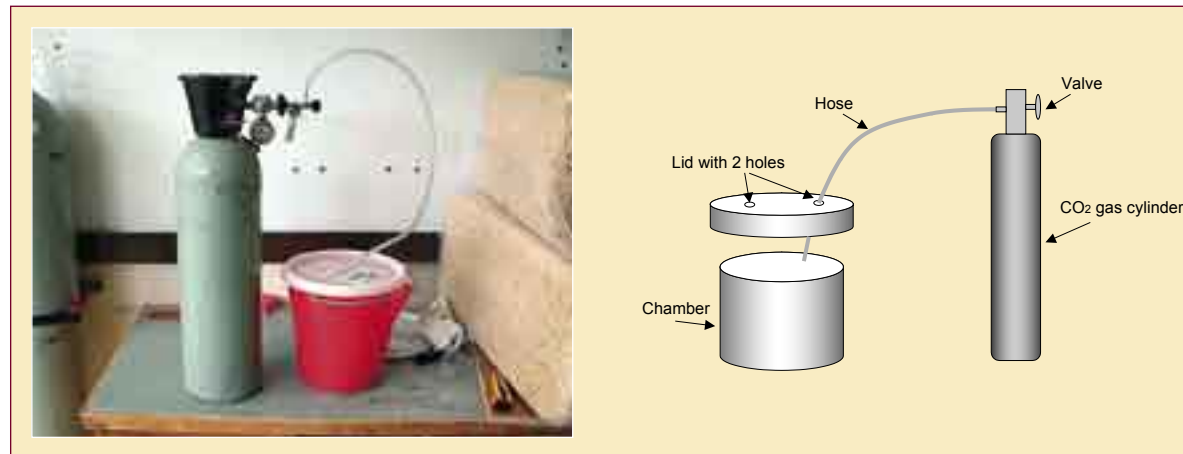
Figure 3.11   Equipment needed for euthanasia with carbon dioxide (CO_2) gas.

Procedure

- Cut a small hole into the corner of a large plastic bag. This allows excess air and fumes to escape.
- Place the animals into the large plastic bag (inside cloth bags or cage).
- Place a collar (rubber tubing or cloth) around the exhaust pipe of the vehicle, then wrap the plastic bag around the rubber collar—so that the plastic bag does not melt onto the exhaust pipe, and so that the person holding the bag does not get burnt. Once the bag is in place, start the vehicle engine. The whole operation should be performed in a well-ventilated place so that the person holding the plastic bag does not get exposed to the vehicle fumes.
- It should take approximately 1.5–2 minutes for an adult rat to die using this method.

Cervical dislocation

This technique is useful for small (mouse-size) animals only. It requires experience to conduct this method quickly and effectively. The technique involves grasping the head and the body in each hand and pulling quickly and firmly so that you feel the neck dislocate. This severs the spinal cord and death occurs very rapidly. This technique is not recommended if the animal will be used for taxonomic assessment as it may cause damage to the cranium.

Safety issues

Anyone working with wild rodents should be aware that many species carry diseases and parasites that can be transmitted to people. However, the risk of transmission can be minimised by following some simple guidelines:

- avoid being bitten—handle animals as little as possible, use secure methods, and avoid causing them distress or injury
- cover open wounds, scratches or cracked skin on hands or wrists before handling rodents—apply bandaids (adhesive dressings) or bandages to affected areas (applying a barrier cream to hands during field work may help prevent cracked skin and therefore lessen the chance of infection)
- avoid placing your hands near your eyes, mouth or nose while handling rodents
- wash your hands thoroughly as soon as possible after handling rodents or traps etc., using soap, nail brush and hot water and then an alcohol lotion, if available
- wear surgical gloves when conducting dissections/autopsies.

Diseases transmitted to humans by rats and mice

There are more than 200 pathogenic micro-organisms, helminths and arthropods described from the three main commensal rodents—*Mus domesticus*, *Rattus rattus* and *Rattus norvegicus*. Some of these micro-organisms may be pathogenic to humans. We have a good knowledge from our recent studies of the range of helminths and arthropods that occur in *Mus domesticus* in Australia and this species also has been screened for antibodies to various micro-organisms. In contrast, our knowledge of pathogens carried by *Rattus* species both in Australia and Southeast Asia is poor.

Some human pathogens that can be transmitted by rodents are *Leptospira* (reactions vary from asymptomatic to fatal disease; responds rapidly to antibiotic treatment), the arbovirus family (arthropod-borne viruses such as Ross River virus), the reovirus family (associated with the respiratory and enteric tract of humans), Hantaan virus, plague (again, responds well to antibiotic treatment), rat typhus and lymphocytic choriomeningitis virus (LCMV; symptoms vary from influenza-like to severe meningitis). The plague (225 cases detected in rodents in Java in 2001), leptospirosis (more than 14,000 human cases with 365 deaths in Thailand in 2000), Hantaan virus (sero-positive rodents reported in Indonesia and Thailand) and rat typhus (5000 human cases and 9 deaths in Thailand in 2001) are present in Asia. Further information on the importance and impact of rodent-borne diseases is given in Chapter 8.

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Rodent taxonomy and identification

Introduction

South and Southeast Asia and the main island of New Guinea support some of the richest rodent communities of anywhere in the world. Even in heavily modified agricultural land, it is not uncommon to find six or seven different species living together in one community. In upland regions, with their complex mosaic of forest, gardens and regrowth, this number may reach 15 or 20, with the addition of a suite of primarily forest-dwelling species that make occasional forays into adjoining cropping areas.

Naturally, it is important to be able to accurately identify the various rodent species in these complex communities. To properly understand the ecology, information on abundances, breeding activity or movements will need to be collected separately for each species, and any misidentifications may result in a confused picture. Good species identification is also necessary to ensure that rodent control activities do not have an adverse effect on any non-target species that may be either neutral or beneficial to agriculture, or rare and of conservation concern.

Unfortunately, rodents are often quite difficult to identify to species level. This is especially true of members of the family Muridae, the group that includes nearly two-thirds of living rodents, and almost all of the major pest species. Three factors contribute to this situation. The first is the remarkable ability of murid rodents to undergo major shifts in ecological adaptation with only minor changes in morphology. For example, *Rattus rattus* (the house rat) and *R. argentiventer* (the rice-field rat) are so similar in appearance that a trained eye is needed to tell the two apart, even when they are lain side by side. However, *R. argentiventer* is entirely terrestrial and lives in burrows, while *R. rattus* is an excellent climber and often occupies arboreal nests. The second complicating factor is that all murid species go through quite pronounced changes through life in body proportions, fur texture and colouration. This means that juveniles, subadults and adults of one species often differ more from each other than do the same growth stage of different species. And finally, some rodent species are highly polymorphic—that is, they show a lot of morphological variation within populations. For example, many populations of *R. rattus* contain adult individuals with pure-white, brown or grey belly fur and these variants are often mistaken for separate species. High levels of variation in turn provide prime material for natural selection—with the result that many murid populations can undergo rapid

morphological changes, over only a few generations, to suit local conditions.

The science of taxonomy tries to make sense of all of this variation and to identify the basic species that exist in nature. It also attempts to provide diagnostic criteria whereby the species can be distinguished from each other. In this chapter, we will start by introducing some of the basic concepts and principles that underpin the 'science' of taxonomy and the 'art' of rodent identification. We then review some of the more important morphological features that are useful in distinguishing between different rodent species and provide instructions on how to collect voucher specimens and tissues for genetic analysis. A key to the pest rodents of Southeast Asia and the Pacific region is given in Chapter 11.

Basic taxonomic concepts

The meaning of scientific and common names

All species have two-part scientific names that should be written in italics (e.g. *Mus musculus*) or may be underlined instead (e.g. Mus musculus). The first part always begins with a capital letter and signifies the name of the **genus**. The second part always begins with a lower case letter and is the **specific epithet** (specific name). Together, the two parts

denote the proper species name. If the name includes a third part (also all lower case), this denotes a subspecies (e.g. *Mus musculus castaneus*). A scientific name is sometimes followed by a name and a date, e.g. *Rattus argentiventer* (Robinson and Kloss, 1916). This is the name of the person or people who first described the species and the date of the publication in which the name was first used; the combination of name and date is known as the **authority**.

The application of scientific names is governed by a set of very precise rules set down by the International Commission for Zoological Nomenclature. One of these rules states that the earliest available published name must be used for each currently recognised species or subspecies. Various checklists of rodent names are available but it can still be difficult to navigate through the plethora of different names and combinations (see Box 4.1). In Chapter 11 we list some of the more commonly used alternative scientific names for each of the major rodent pests.

Common or 'vernacular' names are not bound by any equivalent set of rules. This means that there is no such thing as a 'correct' common name and each person can use whatever term they prefer. For example, the wild progenitor of the domesticated laboratory rat, *Rattus norvegicus*, is variably called the brown rat, sewer rat or Norway rat in English, and it is *chuot cong* ('tunnel rat') in Vietnamese. None of these names is any more correct than the others. Indeed, almost all common names are sometimes

misleading if they are taken as genuinely descriptive terms. *Rattus norvegicus* is not always brown, it does not always inhabit sewers, and the species most certainly did not originate in Norway!

Box 4.1   Why taxonomic names sometimes change

Many species of rodents have been known by a variety of different scientific names and this can make it difficult to use some of the earlier literature. These name changes can reflect a variety of past taxonomic actions and decisions, including:

- the lumping of various geographical populations into a single, more widespread species
- the splitting of one species into two or more individual species, based on new studies
- the movement of a species from one genus to another; e.g. *Gunomys bengalensis* became *Bandicota bengalensis* when the genus *Gunomys* was placed under *Bandicota*
- the discovery of an earlier name in a previously obscure publication.

Units of classification

The basic biological unit of the natural world is a **population**—a group of individuals that occupy a single locality and among which all members of one sex could potentially interbreed with all members of the opposite sex (however breeding is often

constrained by social structures). In theory, genetic and morphological variation should be more or less randomly distributed among individuals within a single population (although preferential mating systems may cause some non-random effects, as may very strong local selection).

A **species** (plural also 'species') is a more abstract concept. It is a group of populations from different geographical areas that would be able to interbreed freely if they were all placed together. These populations are thought to share their reproductive compatibility because of a shared ancestry—a common point of origin from whence they spread to occupy their present geographical range. Members of different species are generally unable to breed with each other. This is usually on account of genetic incompatibilities. However, in some cases, the separateness of the species is maintained by behavioural differences and this may break down when individuals of different species are placed together in captivity or in an unnatural environment such as around human habitation. Interbreeding between members of two different species is called **hybridisation**.

Where a species has come to occupy a large geographical area, different local populations often differ from each other in subtle ways. This may have occurred through random genetic changes in isolated populations (e.g. on islands) or through natural selection to better suit local environmental conditions. These morphologically distinct local populations are sometimes identified as different **subspecies**. Subspecies names are also sometimes used for different variants within a single population (e.g. white-bellied *Rattus rattus* are sometimes called *Rattus rattus arboreus*). However, this is an incorrect use of the category and should be discouraged. Another undesirable practice is the use of subspecies names to distinguish geographically isolated populations that do not otherwise differ in morphology (e.g. many island populations).

The **genus** and **family** categories are even more abstract than the species. In the past, a genus (plural 'genera') was most often used to draw together a group of species that were basically similar to each other in appearance and habits. Likewise, a family pulled together a group of similar genera. More recently, both of these categories have been given an evolutionary meaning—a genus is group of species that are believed to have evolved from a common ancestral species; and a family is a still larger group of related genera.

Morphological and genetic approaches to distinguishing species

Rodent species are most often distinguished on the basis of morphological characteristics, including differences in body size and shape, fur texture and colour, and details of the teeth and skull. This has sometimes included the statistical analysis of large numbers of measurements, making the taxonomy somewhat more repeatable and hence more 'scientific'.

In recent years, the application of genetic methods has produced a revolution in taxonomy (see Box 4.2 for notes on collecting samples for genetic analysis). At the species level, genetic analysis can be used to directly quantify the amount of interbreeding that is occurring within and between populations. Hybridisation between species is easily detected genetically and its potential impact on each species can be estimated. Genetic methods can also be used to recover the history of dispersal of species across a landscape and to estimate the relative (and to some extent, the absolute) timing of key events such as water-crossings or other causes of range fragmentation. At the genus and family levels, the evolutionary history of groups of species also can be reconstructed with increasing levels of precision, thereby removing much of the guesswork that previously surrounded these categories.

Genetic studies are currently under way for several groups of Asian rodents. The results of this work will almost certainly require some changes in the taxonomy of several groups including some of the major pest species. However, in the long term, the application of these methods will result in a more stable and scientifically based classification, as well as many valuable insights into the evolutionary history of the group.

Collecting voucher specimens

You can preserve voucher (reference) specimens either as dry or wet specimens. In either case, it is essential that you label these with details of the place and date of collection, the collector's name and any specimen number or code that links the voucher back to tissue samples. The label should be durable, securely tied to the specimen and written in pencil if the specimen and label are to be placed in ethanol (as most inks are alcohol soluble will thus disappear). Wherever possible, you should also preserve a piece of soft tissue (ideally, liver) for future DNA analysis (see Box 4.2).

Wet specimens

Wet specimens first need to be **fixed** in an appropriate solution. Formalin or ethanol are the two most commonly used fixatives. Each has advantages and disadvantages and you should think carefully before deciding which one to use.

Formalin is the best fixative if you intend to use specimens for detailed anatomical or histological studies (examining the tissues microscopically). Formalin is usually purchased as formaldehyde, mixed as a 37% solution. You will need to dilute this with water to 10% of its original concentration to give a **10% formalin** solution. If the specimens are going to remain in this solution, the formalin should be buffered to a pH of 7 (one option is

to use 4 g monobasic sodium phosphate and 6 g dibasic sodium phosphate per litre of 10% formalin). Without buffering, the bones of specimens stored in 10% formalin will soon decalcify and the flesh will harden.

The two main disadvantages of using formalin as a fixative are:

- it causes extensive damage to the deoxyribonucleic acid (DNA), making specimens fixed in this way unsuitable for genetic studies
- it is a severe irritant (especially to eyes and the respiratory tract) and a poison. Formalin should only be mixed and used in well-ventilated spaces.

Ethanol is not recommended as a fixative for anatomical studies. However, it is much safer to use than formalin and has the extra advantage that it gives good preservation of DNA. Ethanol is also more readily available than formaldehyde in many countries. If ethanol is not available, methanol (another alcohol, usually sold as methylated spirit) can be used instead, but only as a last resort. Ethanol is usually diluted to 70% concentration. Higher concentrations are good for DNA preservation but will dehydrate tissues and make a specimen very hard and inflexible.

Regardless of whether you use alcohol or formalin for fixation, it is always best to slit open the belly (taking care not to cut into the intestine or damage any embryos) to allow the fixative to enter the body

cavity. If a needle and syringe are available, you should also inject the fixative into each of the major muscle masses (shoulders, thighs, neck) and into the chest cavity. Injection is especially important when using ethanol as a fixative. After injection, place the specimen in at least five times its own volume of fixative. It is usually necessary to leave the specimen for at least 3 days, or until the big muscle masses feel firm (but not hard) to the touch.

Specimens that are fixed in formalin are usually transferred to alcohol for long-term storage. Before placing a formalin specimen into alcohol it must be rinsed thoroughly in water. If you have used ethanol or methanol for fixation, replace the fluid after 5–7 days. Small specimens will take less time to fix than larger ones.

Keep specimens stored in ethanol/methanol in airtight containers, out of direct sunlight. Check the fluid level occasionally and top up if necessary. Specimens stored in ethanol/methanol can remain essentially intact for many decades, or even centuries. They can be rinsed and stored for one or two days in water for use in training sessions, but they should be returned to ethanol as soon as possible after use.

Wet voucher specimens can take up a lot of storage space and consume large quantities of fixative. A good way to conserve space and materials is to fix and preserve only the skin. This involves carefully removing the skin from the body, leaving only the

head, hands, feet and tail inside the skin. A skin will be well fixed after only 1 day in formalin or 3–4 days in ethanol. A skin fixed in this way can be made up into a dry specimen at a later date (see below).

Dry specimens

If it is not possible to preserve and store wet specimens, the next best option is to prepare the skull as a voucher specimen. If possible, before you do this, photograph the living or freshly killed specimen as this provides a valuable source of supplementary information to accompany a cleaned skull. Carefully label the photograph with the same details as the skull.

To clean the skull, remove the skin and then boil the head until the muscles and other tissues are soft enough to be picked away without damaging the bones. You can also soften the flesh using a weak solution of sodium perborate. Alternatively, place the skulls in a location where ants can consume the flesh (but away from the attention of dogs or chickens) or put them in a fine mesh bag (wire or nylon) and submerge them in a pond or paddy field where aquatic organisms will do the job.

After cleaning, label the skulls individually and tie or wire the lower jaw to the cranium. Skulls should be stored in plastic or glass vials, or in sturdy cardboard boxes to protect them from damage.

The preparation of dry 'museum-style' skins is a specialist task that is not recommended unless a permanent reference collection is needed. In this case, a special collection area has to be established— somewhere that can be kept dry and free of insect pests. Insects will rapidly destroy any dry specimens left unprotected. Unless moisture is excluded, fungus will also invade and eventually destroy dry specimens. Long-term storage of dry specimens requires similar conditions as storage of dry insect or plant collections.

BOX 4.2 Collection of tissues for DNA analysis

Good DNA sequences can be obtained from small pieces of animal tissue preserved in ethanol. Almost any tissue can be used, but some suggestions are given below for the tissues that give the best results.

If an animal is to be sacrificed, take a tissue sample as soon as possible after death. The most widely used tissue is the liver, but other organs such as lung, kidney, spleen etc can also be used. Muscle either from the heart or from the chest or thigh can also give good results. The most important thing in all cases is to fix the tissue soon after death. This is particularly critical in the case of organs such as liver and kidney that contain many destructive enzymes. If an animal has been dead for some time (e.g. from a kill-trap or a road kill), it is best to collect a sample of muscle tissue from the part of the body that shows the least obvious decomposition. Also, pluck some hairs from the body and include them with the muscle sample.

Place a 5 mm cube of the chosen tissue immediately into a 3–5 mL tube of 70–90% ethanol, then cut the tissue into smaller pieces (approx. 1 mm cubes) with a new scalpel blade or clean fine-pointed scissors. This assists with penetration of the ethanol and improves fixation of the DNA.

Label the tube clearly and carefully. If the tube is likely to leak, write the labels in pencil or scratch them into the tube (as most inks are soluble). The information on the tube must be sufficient to allow the collector to determine the date and place of collection, and the identity of the sampled animal. This latter information might be a numbered voucher specimen or it might be a reference to measurements in a field notebook or to a photograph. The most useful samples are those associated with a voucher specimen, because this allows the DNA results to be linked back to the physical characteristics of the sampled animal.

Keep the samples stored in ethanol out of direct sunlight in as cool a place as possible. Storing them at 5–7°C in a refrigerator is ideal, but not essential for good results.

Major groups of Asian rodents

Four major groups of rodents are represented in Southeast Asia and the Pacific region (Figure 4.1). The major attributes of each are listed below. Here we will be concerned primarily with the Muridae, the group that includes all of the major pest species. Some good general sources on squirrels and other groups of rodents are indicated under Further reading.

Family **Hystricidae** (porcupines)—chunky build; very long, stiff, sharp spines project through fur

Family **Rhizomyidae** (bamboo rats)—chunky build; tail is short, unscaled and almost hairless

Families **Sciuridae** (ground squirrels) and **Petromyidae** (flying squirrels)—variable build; tail is heavily furred to tip

Family **Muridae** (rats, mice etc.)—mostly slender build; tail is generally sparsely furred and has distinct scales arranged in concentric rings.

The family Muridae includes more than 1350 species, the majority of which are found in Eurasia, Africa and Australia. It includes many of the world's most familiar rodents, such as the house rats and house mice, and some of the most destructive of all agricultural pests. However, it also includes many hundreds of other species that play important roles in landscape ecology at all scales and that should be protected and conserved.

Identifying murid rodents

The process of identifying unknown rodent specimens can be made simpler and more reliable if the following basic steps are followed:

- determine the age and sex of the specimens (see below)
- set any juveniles aside and **work first with adults**
- work each through the key provided in Chapter 11 to obtain a provisional identification
- check the notes on geographical distribution and morphological features given in Chapter 11
- if the specimen does not fall within or close to the known geographical range or does not match the description, try working through the key again
- if a convincing identification cannot be obtained, consider taking a voucher specimen and a DNA sample (see Box 4.2).

The reason why determining age and sex is so important is that rodents change greatly in appearance through their growth and development. This is most notable in the texture of the fur but it also affects their body proportions (e.g. relative tail length). Age and sex can be determined by examining the external reproductive condition, as described below. Young rodents are often very difficult to identify. This is best done by first identifying some
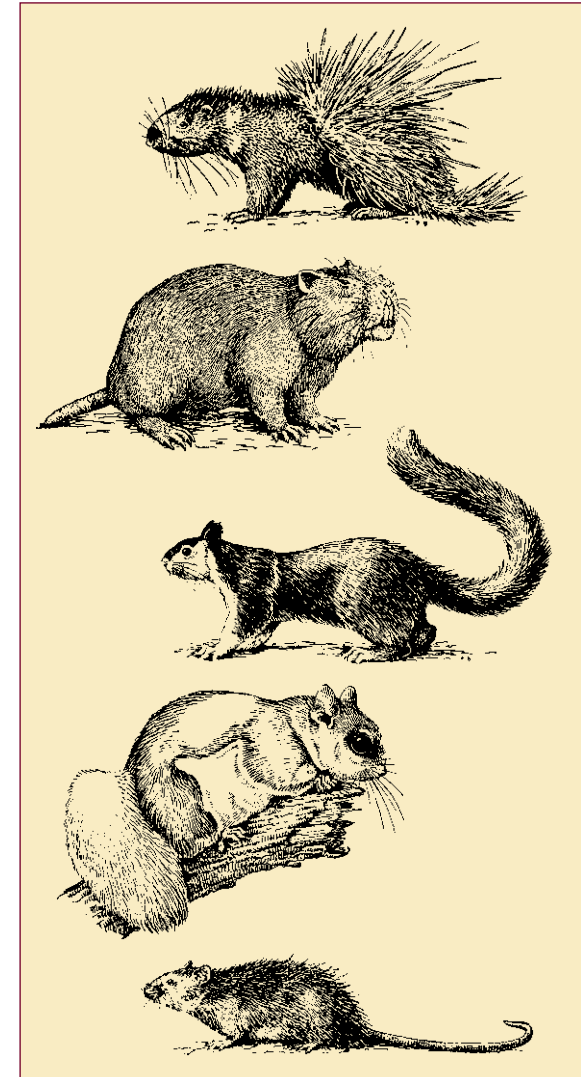
Figure 4.1   Examples of each major group of Asian rodents. From top to bottom: a porcupine; a bamboo rat; a ground squirrel; a flying squirrel; and a rat (after Grassé and Dekeyser, 1955).

sexually mature specimens and then attempting to match juveniles with adults by comparing their features directly. Digging of breeding burrows can result in young animals sometimes being captured together with their parents; these can be used as reference specimens.

Determining the age and sex of a rodent

To determine the age and sex of a captured rodent, hold the animal so that the belly faces you and the head is pointed away from you. The opening at the base of the tail is the **anus**. Both sexes have a **genital papilla** that covers the penis in males and the clitoris in females.

In juvenile male rodents, the **testes** are initially located inside the body, in an **abdominal** position. As the animal matures, the testes enlarge and descend

Figure 4.2   Comparison between juvenile (abdominal testes; left) and adult (descended testes; right) males.

to adopt a scrotal position, inside a hairy **scrotal sac**. In a fully adult rodent, the scrotal sac often projects behind, and hence obscures, the anus (Figure 4.2). The skin at the back of the scrotal sac is often hairless and darker than the surrounding skin. This houses a sperm storage organ called the **epididymis**.

In females, the anus and genital papilla are close together and the skin between them is bare or thinly furred. The vagina should be visible just behind the genital papilla. In juvenile rodents, the vagina is sealed off by a thin, shiny layer of skin, the hymen. This condition is known as an **imperforate vagina** (Figure 4.3). As the animal reaches sexual maturity, the vaginal covering breaks down and the vagina is open or **perforate** from then on. The vagina will be widely open if the animal has recently mated or given birth. It is smaller (but never fully closed off) if the animal is mature but has never mated, or not recently mated.

Figure 4.3   Comparison between juvenile (closed or imperforate vagina; left) and adult (open or perforate vagina; right) females.

Female rodents also have teats associated with subcutaneous mammary glands. These are arranged down either side of the body (Figure 4.4). The teats are prominent and should be easy to locate in sexually mature females, especially in those that have had young. However, they can be very difficult to locate in juveniles and the presence or absence of teats should not be used as a means of determining the sex of an individual. For classification purposes, pairs of teats, or **mammae**, are counted in three groups; pectoral, postaxillary and inguinal. For the rodent shown in Figure 4.4, for example, the number of teats would be given as 1+2+2.
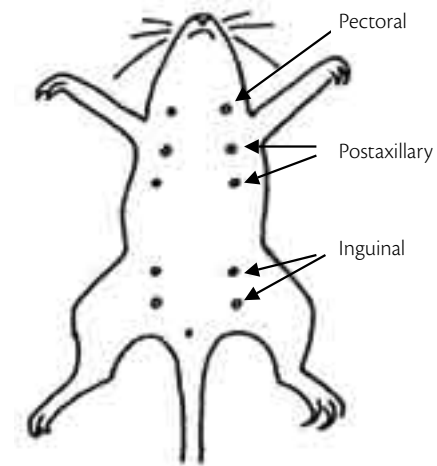
Figure 4.4   Arrangement of mammae on an adult female *Mus domesticus*. In this species, there is one pair of pectoral teats, two pairs of postaxillary teats, and two pairs of inguinal teats (denoted as 1+2+2).

It is more difficult to determine the sex of very young individuals, such as thinly furred pups or recently weaned juveniles. The best way is to compare the distance between the anus and the genital papilla. This will be much greater in a juvenile male than in a juvenile female. In addition, there should be a distinct unfurred line between the vagina and the anus in juvenile females.

Taking measurements

Body measurements are quite useful in limiting the range of possible identifications for an unknown rodent. For example, a fully adult rodent weighing only 10 to 20 g is almost certainly a species of *Mus*, while a body weight of 400 g eliminates all but handful of possibilities. Routine recording of body measurements also provides a good check on field identifications, and may help to highlight any records that might be in error.

Fieldworkers usually take a standard set of external measurements. These are explained and illustrated below. The linear measurements should be taken to the nearest millimetre; any greater precision is probably not repeatable, especially if the measurement is taken on a squirming live rodent! It is best to use a good-quality plastic ruler which has the end trimmed to set the zero mark at the very edge of the ruler.

If you are working in a group, ensure that everyone in the group takes measurements in exactly the same way. This minimises variation occurring between researchers. If anyone is inexperienced, they should practice by taking measurements on an individual already measured by another person; the external measurements should be repeatable to $\pm$ 1 mm for the ear and hind-foot lengths, and $\pm$ 3 mm for the head+body and tail lengths.

Head+body length

The combined length of the rodent's head and body is known as the 'head+body' length. Take the head+body measurement in a straight line along the animal's vertebral column, from the tip of the nose to the distal end of the anus (with the animal lying on its back) (Figure 4.5). Live rodents rarely cooperate in this exercise, hence the head+body measurement is often less precise than those taken of the tail, foot and ear.
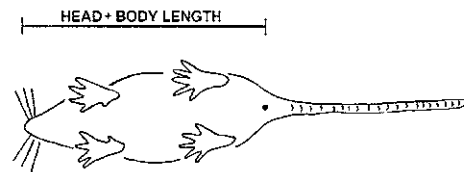
Figure 4.5 Measure the head+body length along the spine of the rodent from the tip of the nose to the middle of the anus.

Tail length

Measure the tail along a straight line from the middle of the anus to the tip of the tail (Figure 4.6). Do not suspend the animal by its tail to take this measurement—the tail will stretch and the measurement will be too long.

Only take the tail measurement on complete, undamaged tails. A damaged tail will terminate in a short, pale section that lacks hairs and scales. If the tail is incomplete, note this on your data sheet.
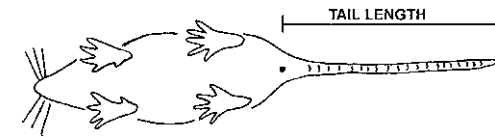
Figure 4.6 Measure tail length from the middle of the anus to the tip of the tail.

Pes length

Measure the **pes** (hind-foot) from the heel to the tip of the central (longest) toe, but without including the claw (Figure 4.7). For live animals, the end of the ruler can usually be hooked under the claw, allowing the foot to be gently flattened against the ruler.
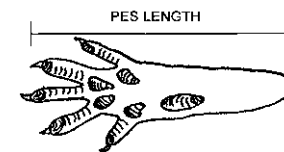
Figure 4.7 Measure the pes length from the base of the heel to the end of the toe pad on the longest toe (not including the claw).

Ear length

Measure the ear from the bottom of the notch of the ear to the furthest point along the rim (Figure 4.8). Do not take the measurement if the margin of the ear is damaged as a result of fighting.
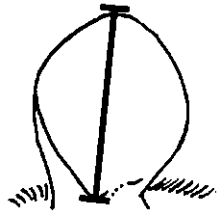
Figure 4.8   Measure the length of the ear from the bottom of the ear notch to the furthest point along the rim.

Body weight

Rodents and other small mammals are usually weighed using a calibrated spring balance (such as a Pesola spring balance; Figure 4.9). Such balances are available in various sizes. Be sure to use an appropriately sized balance for the individual rodent and hold the balance by the swivel ring at the top. Suspend dead animals by a foot or the tail.

Check balances before each session to make sure that they are calibrated to zero (or to the correct mark if it has been adjusted to allow for error).

Live animals are generally weighed inside a cloth bag. Tie a knot in the top of the bag and take the weight of the rodent plus the bag. After the animal is removed, weigh the bag by itself. The weight of the rodent will be the difference between the two measurements. Make sure that you use an appropriately sized bag—do not weigh a 5 g mouse in a rice sack!

Figure 4.9   Weighing a rodent using a Pesola spring balance. If possible, two people should read the balance to avoid any misreading.

Diagnostic characteristics

Only a few species of rodents possess uniquely diagnostic features, such that they are instantly recognisable. More typically, rodents are distinguished from each other by unique *combinations* of features.

The following list indicates the kinds of external characteristics that will be useful for identification:

+ general body proportions
+ colour and texture of the fur on the belly, flanks and back
+ size, shape and hairiness of the external ears
+ colour and length of the vibrissae (whiskers) on the face
+ size and colour of the incisor teeth
+ detailed patterning and hairiness of the tail
+ colour and overall shape of the manus and pes (fore- and hind-feet, respectively)
+ size and shape of the pads and claws on the manus and pes
+ size and shape of the scrotal sac in males
+ number and distribution of teats in females.

The following notes are provided as a guide to the kinds of features to look for when examining a rodent specimen.

Body proportions

Murid rodents do not vary much in basic body proportions. The most striking difference between species relates to the relative length of the tail, which ranges from less than 50% of head+body length to more than 200%. Some murid rodents have a distinctly chunky body form with strongly muscled shoulders and neck, while others have proportionally longer or shorter heads; however, such variations are difficult to quantify and are thus of little diagnostic

value. Keep in mind that species with long, thick fur will tend to look more heavily built than those with short, sleek fur.

As in most other groups of organisms, body proportions in murid rodents change during the course of individual growth. As illustrated in Figure 4.10 for *Rattus losea*, the ears and feet undergo a period of early, rapid growth, while the tail grows more steadily through life. Young rodents thus appear to have proportionally larger ears and feet than adults of the same species, and this can sometimes lead to them being identified as different species. Tail length shows a more constant proportional relationship to head+body length.

Fur texture

Mammal fur consists of a number of different hair types and it is variation in the length, thickness, form and frequency of each type that give the fur of each species a particular look and 'feel'. The main hair types found in murid rodents are:

+ **contour hairs**—these make up the bulk of the externally visible fur. They are usually morphologically unspecialised but are often 'banded' in colour
+ **spines**—specialised contour hairs with a flattened (often grooved) midsection. Fur with large and abundant spines often feels quite 'stiff' and will stay in position when brushed forward (Figure 4.11). Usually confined to the back and

flanks, but also present on the belly in some species
+ **underfur hairs**—short, fine hairs that can only be seen by parting the contour hairs. Dense underfur will give the fur a 'woolly' texture
+ **guard hairs**—long and often quite thick hairs that project some distance (sometimes several centimetres) beyond the contour hairs. These are generally longest down the centre of the back, especially on the lower back.
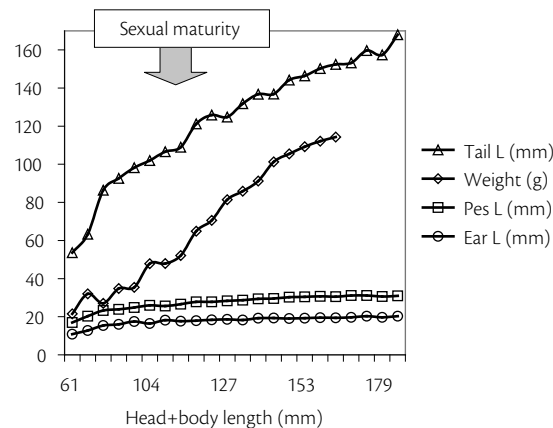
Figure 4.10   Changes in body proportion during growth in male *Rattus losea*, illustrated by plotting each of tail length, body weight, pes length and ear length against head+body length. The pes and ear attain their final size at an early stage of growth, while the tail continues to grow well past the time of sexual maturation (at around 45–50 g).

In juvenile rodents, the outer fur consists of contour hairs and short guard hairs, and it is always soft. Spines only emerge following completion of one or more **moults**, and the guard hairs also only become conspicuous with increasing maturity. Moults occur as 'waves' of hair replacement running backwards and upwards from the shoulders and lower flanks, and they take place throughout life. Early moults are quite orderly and may be visible on the flanks and back of juveniles and subadults as bands of different fur colour or texture. The moulting process in adults is more erratic and is generally difficult to detect. However, it is worth noting that the brightness or 'freshness' of the fur colour will vary according to the moult stage of the individual.
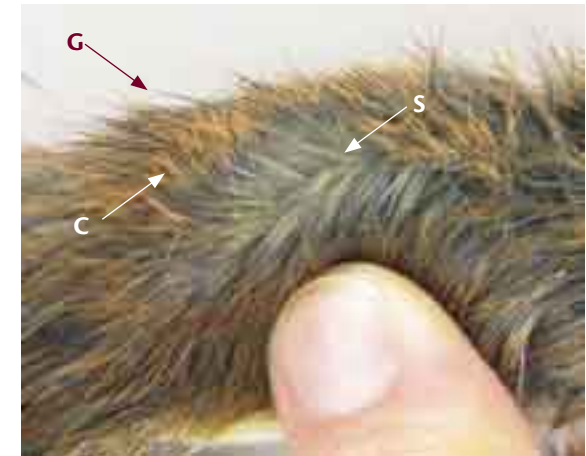
Figure 4.11   Dorsal fur of a species of *Niviventer*, illustrating three of the four hair types: guard hair (G); contour hair (C); and spine (S). The underfur is visible as very fine fibres among the spines and contour hairs.

Fur texture may vary greatly within a species, especially where one species spans a broad altitudinal or latitudinal range. Not surprisingly, populations living in regions with a cool climate tend to have denser, woollier fur than those living in hotter regions.

Colouration

Fur colour in rodents is a complex feature that requires some specific terms if it is going to be useful for identification. This is because the different kinds of hairs (described above) often differ in colour, while some kinds of hairs are commonly 'banded', with different zones of colour along individual hairs. In addition, the fur of most murid rodents is coloured differently on their back (**dorsum**) and belly (**venter**). These colours may blend gradually on the flanks, or they may be sharply demarcated (see Figure 4.12). In a few species, a third, distinct band of colour is present on the flanks.

In describing the fur colour of a rodent, it is often useful to note its 'overall colouration', as might be observed by holding it at arm's length. Hence, a species might be described as 'overall, dark grey above, with a sharply demarcated cream venter' or ' overall, dorsum reddish brown, merging into a buff venter'. However, it is often necessary to be far more specific. When describing banded hairs on the dorsum or venter, it is usual to distinguish the outermost colour (called 'tipping' or a 'wash') from the deeper, basal colour, e.g. 'fur on belly grey-based, with cream tipping'. The

colour of individual hair types on the back is often noted, e.g. 'contour hairs reddish-brown with dark-grey bases, guard hairs clear or with short, black tips'. Where the different hair types are strongly contrasting, the overall colour may be described as 'peppered', e.g. 'dorsum orange–brown, peppered with black'.

In taxonomic descriptions, fur colour is sometimes specified more carefully, using various colour standards taken from soil science or other sources. This level of detail is generally not helpful for field identifications.

Two regions of the body deserves special mention in regard to fur colour, namely the head and the pectoral region or 'chest'.

In most rodents, fur colour on the head is a continuation of that on the body, with the belly

Figure 4.12   An example of sharp demarcation in fur colour between the dorsum and venter, in *Leopoldamys sabanus*.

colour typically extending onto the throat and chin. However, a significant number of species show more complex fur colouring on the face (see Figure 4.13). The most common elements are:

- an **eye ring**—usually a narrow band of dark hairs encircling the eye
- a **facial mask**—a more extensive strip of dark fur running through the eye and onto the side of the snout
- a **cheek patch**—usually made up of pale hairs and situated below the eye, sometimes extending onto the lower part of the muzzle
- a **preauricular patch**—consisting of a narrow fringe of distinctly coloured hairs along the anterior margin of the ear.

Figure 4.13   Examples of facial patterning among murid rodents. An orange cheek patch and dark eye ring in *Chiromyscus chiropus* (top left); a white cheek patch and lower muzzle in *Mus terricolor* (top right); and an orange preauricular patch in *Rattus argentiventer* (bottom).

The chest in rodents sometimes bears a distinctly coloured patch of fur. This may be cream or white set against a darker background colour, or it can be a darker patch (or mid-ventral line) of fur set against an otherwise pale venter. In some species, this patch is reddish-brown and has the appearance of a 'stain'. This may be due to the presence of skin glands in this region (found in some murids and in many other groups of mammals), but little is known about this phenomenon in Asian rodents.

In *Nesokia indica*, the head and shoulders are more brightly coloured than the rest of the dorsum (see Chapter 11). This degree of patterning on the body is common in other groups of rodents (e.g. squirrels) but it is quite rare among Asian murids. One very unusual group of murids (*Chrotomys* spp.), that is sometimes trapped in rice fields and gardens in the Philippines, is distinguished by the presence of dark longitudinal stripes along the dorsum.

A final word on colouration concerns intraspecific variation and 'aberrant' patterns. Coat colour variation in *Rattus rattus* has been noted earlier. The venter is particularly variable in this species, with many populations showing a mixture of pure-white and grey-based ventral fur colours. This variation is under simple genetic control and strong natural selection can lead to some segregation of these colour forms by habitat. As a general rule, dark-bellied forms seem to be more common around villages where white-bellied individuals might be easier to observe and kill.

Variation in dorsal fur colour (various shades of browns to black) is also found in *R. rattus*, and the best known example is the melanistic form (the true 'black rat') that is common in Europe and some other parts of the world. Melanism is rare among Asian populations of *R. rattus*, but it has been observed in various other species, including *Rattus norvegicus* and *R. losea*. Other species of rodents generally show less variation in dorsal and ventral fur colour within any one population, but there are many examples where fur colour differs between populations, especially where one species occupies a range of habitats.

Aberrant colour patterns include individuals with one or more, randomly positioned spots of contrasting colour, or in some cases, with a 'saddle' of pale fur that runs up from the belly on both sides and may even encircle the whole body. These aberrant patterns may occur in low frequency in all species and in some cases they reflect previous injuries (e.g. burns or torn skin). Albino individuals presumably occur in all species, but these would be unlikely to survive for long in the wild.

Vibrissae

Vibrissae (often called whiskers) are specialised hairs that are connected to special sensory nerves. In murid rodents, they are found only on the head and lower forelimbs. The head vibrissae are arranged in seven or more groups, the placement of which is fairly constant within and between species

(Figure 4.14). The most obvious and functionally most important group are the **mystacial vibrissae** that occupy either side of the snout. These are highly mobile and are used in orientation and movement. The other groups are used mainly for orientation or, in the case of those clustered around the mouth, in the positioning and protection of the lips during gnawing, digging and food ingestion.
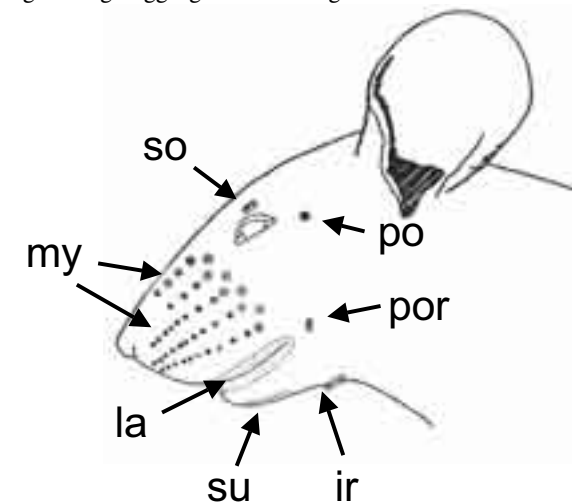
Figure 4.14 Terminology of vibrissal groups on the head of a murid rodent: interramal (ir); labial (la); mystacial (my); postorbital (po); postoral (por); supraorbital (so); submental (su).

All vibrissae grow out of specialised follicles; hence their position is constant through life. Worn vibrissae are replaced by a new shaft that grows from the same follicle. Because the old and new vibrissae can coexist for some time, the exact number of vibrissae is quite variable.

Although all murid rodents probably share the same basic set of vibrissal groups, the vibrissae themselves can vary greatly in thickness, colour and length in different species. As a rule, the vibrissae appear to be especially thick and long in the more highly arboreal species (e.g. *Leopoldamys* spp.), and noticeably short and fine in some of the more terrestrial or fossorial forms (e.g. *Bandicota* spp.). Notably, however, there is no obvious difference in vibrissal thickness or length between the arboreal *Rattus rattus* and its terrestrial, burrowing relatives *R. argentiventer* and *R. losea*.

External ears

Murid rodents have very simple external ears (Figure 4.15). However, the ear flap or **pinna** varies in both length and relative breadth between species. It also varies in the degree of pigmentation, from relatively pale to quite heavily pigmented and dark.

Figure 4.15 The external ear or pinna of two murid rodents showing a difference in the degree of hairiness of the ear between *Leopoldamys sabanus* (left) and *Bandicota indica* (right).

In all species, the inner and outer surfaces of the pinna are covered with fine hairs. These are more conspicuous in some species than others, and in a few species they form a delicate fringe around the margin of the pinna (Figure 4.15).

The ears of unweaned pups are very small and fleshy. Rapid growth of the ears usually starts towards the end of the second week of life.

Incisors

All rodents have only one incisor in each side of the upper and lower jaws. These teeth grow continuously and the animals must gnaw on hard material regularly to stop them from overgrowing. Enamel is restricted to the front and outer surface of each tooth.

The upper incisors of Southeast Asian murid rodents vary in relative width and orientation, and in the colour of the enamel. The widest incisors, with a combined upper incisor width greater than 3 mm, are found in the species of *Bandicota* and *Nesokia*. *Nesokia indica* is unique in having the paired lower incisors wider than the paired upper incisors.

Incisor enamel in murids is usually a dark orange on the upper pair and slightly paler on the lower pair. Some species have much paler enamel—perhaps best described as pale yellow or cream coloured (Figure 4.16).

The upper incisor tips point vertically downwards or even slightly backwards in most murids (Figure 4.17). However, species that excavate extensive burrow systems often use their incisors to dig and transport fragments of soil and rock. In these species, the upper incisors usually point slightly forward. This is best seen in *Bandicota bengalensis* and *Berylmys berdmorei*, both of which are strong diggers. In contrast, other

Figure 4.16 Incisor teeth of two murid rodent species, showing differences in relative width and in the colour of the enamel. *Berylmys berdmorei* (left) has relatively narrow incisors with pale enamel; *Bandicota indica* (right) has broad incisors with dark orange enamel.

Figure 4.17 Differences in upper incisor orientation among murid rodents; curved backward in *Rattus rattus* (left) compared with forward pointing in *Berylmys berdmorei* (right).

burrowing species like *Bandicota indica* and *Rattus argentiventer* have unspecialised incisors.

In some *Mus* species (e.g. many *M. musculus*), the cutting edge of the upper incisors bears a distinct notch. This is produced through wear against the lower incisors and may not be present in all members of a population.

Tail

The degree of hairiness and scaliness of the tail clearly distinguishes each of the four major groups of rodents found in Southeast Asia (differences noted on page 38). Among murid rodents, the tail also provides a suite of useful diagnostic characters, including:

- its length relative to the body
- the form, size and colour of its scales
- the number, length and colour of its hairs
- the presence of a terminal hair tuft or, less commonly, of a lateral hair fringe.

Tail length is variable in all species and should be used with caution in identification. More highly arboreal species generally have longer tails than terrestrial forms, presumably reflecting the use of the tail as a balance organ. Tail length may be under strong selective pressure in populations of some species that occupy a range of different habitats (e.g. *Rattus rattus*). Relative tail length is most usefully expressed as a proportion of head+body length. As noted above, the tail grows at approximately the same

rate as the head+body in rodents, hence relative tail length is not greatly affected by individual age.

Although the tail is scaled in all murids, the size and shape of the **scales** varies between species (Figure 4.18). The size of scales is usually expressed as the number of rows that occupy a 1 cm section, as measured one-third of the way down from the tail base. While this value is highly correlated with overall body size (larger species tend to have lower counts), there are significant differences in mean counts between species of similar body size (e.g. between *Rattus rattus* and *R. argentiventer*; the latter having larger tail scales and lower scale counts).
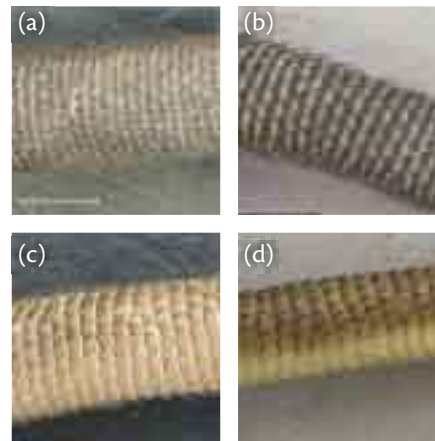
Figure 4.18 Variation in tail morphology among four murid rodents: (a) *Bandicota indica*; (b) *Bandicota bengalensis*; (c) *Chiromyscus chiropus*; (d) *Niviventer* sp. Examples (a) and (b) are 'hairier' than the other two. Examples (c) and (d) are 'bicoloured'. The latter has a sharp boundary between the upper and lower surfaces.

Individual tail scales are essentially rectangular in shape in all Asian pest murids. However, they vary in the extent to which the posterior margin of each scale is prolonged to overlap the scale behind. The extent of overlap is also indicated by the amount of pale skin that is visible between the scale rows (contrast Figure 4.18a with b). 'Strongly overlapping' scale rows are typical of *Rattus* and *Berylmys* species, and some *Bandicota* species. 'Non-overlapping' or 'weakly overlapping' scale rows are found in *Mus* species and in various genera of forest rats (e.g. *Niviventer*; Figure 4.18d).

All arboreal murids use their tail to grasp onto branches or foliage while climbing, but only a few show any obvious morphological specialisation. In a few highly arboreal groups (e.g. the New Guinean *Pogonomelomys* spp.), the upper surface of the very tip of the tail bears a patch of smooth skin—a specialised grasping organ.

Tail colouration in murid rodents is often characterised as being either 'unicoloured' or 'bicoloured'. In a typical **unicoloured** tail—such as occurs in all *Bandicota* and *Nesokia* species, in most *Rattus* species, and in some *Mus* species (e.g. *M. musculus*)—the tail scales are heavily and evenly pigmented at all points on the tail (Figure 4.18a–b). In *Berylmys berdmorei*, the tail is also unicoloured but the scales are weakly pigmented in juveniles and seem to be largely free of pigment (thus 'flesh-coloured') in adults. In a typical **bicoloured** tail

(Figure 4.18d), the scales on the upper half of the tail contain dark pigment while those on the lower half are unpigmented or contain white pigment. The boundary between the upper and lower portions of a bicoloured tail is usually sharp; however, it is diffuse in a small number of taxa, including some *Rattus* species (e.g. *R. norvegicus* and *R. nitidus*) and *Chiromyscus chiropus* (Figure 4.18c).

A different type of tail patterning, sometimes also referred to as 'bicoloured', features a contrasting white or cream-coloured terminal portion (Figure 4.19). This condition is occasionally found as a variant in *Rattus* and *Bandicota* species—but with the white tip usually not more than 5% of total tail length. However, it is common, or even represents the typical condition, in some forest murids, such as some species of *Maxomys*. In some cases, the two forms of tail patterning are found in combination: dorso-ventral distinction combined with an all-white tail tip (e.g. Figure 4.19).

Figure 4.19   A pale tail tip in *Leopoldamys sabanus.*

In all species, tail colouration tends to be more intense and the boundaries more sharply defined in juveniles and subadults than in adults.

In all murids, small **hairs** emerge from under the posterior margin of each scale (Figure 4.18). There are usually three hairs per scale (occasional scales may have five), but in some species this is reduced to a single, very short hair per scale. The hairs also vary in length between species, ranging from less than a scale length to more than two scale lengths. In most species, the hairs become longer towards the end of the tail, and it is not uncommon for the tail to end in a distinct 'tuft' of hairs (Figures 4.19–4.20). However, in some species (e.g. *Bandicota* spp.) the reverse is true and the terminal portion of the tail is almost naked.

Figure 4.20   A strong terminal tail tuft in *Chiromyscus chiropus.*

The tail hairs also vary in colour between species, ranging from clear to white or black (contrast Figure 4.18b with 4.18c). Species with bicoloured tails usually have dark hairs along the upper surface and white hairs below; however, there are exceptions in which the hairs are dark against both pale and dark surfaces.

In a few Asian murid species (e.g. *Chiropodomys* spp.), the lateral tail hairs are elongated and project outwards to form a distinct **lateral tail fringe**.

Fore-limb

The fore-limbs of rodents are used in many tasks including locomotion, climbing, digging, grooming, sexual grasping, and the manipulation of food items. Perhaps because of this multifunctionality, they are very conservative in morphology and show only slight variations in proportions and detailed form, even in species with quite specialised patterns of locomotion (e.g. hopping rodents).

Small vibrissae (carpal group) are found near the wrist in all groups of rodents including murids. Murids lack a second group of vibrissae (anconeal group) that are located near the elbow in some other rodents.

The **manus** or fore-foot of murid rodents, also sometimes referred to as the 'fore-paw' or 'hand', has four well-developed digits, each with a sharp claw. A fifth digit (the innermost one) is reduced to a small nubbin with a flattened nail. The claws tend to be larger and more elongated in species that spend much of their time digging, but smaller and sharply recurved in arboreal species. More generalised terrestrial species tend to resemble the arboreal group in the size and shape of their claws.

The palmar surface of the manus has five fleshy pads in all species (Figure 4.21). These tend to be smaller and more discrete in terrestrial species, but larger and grouped closer together in the more arboreal forms.

In murids, the underneath of each digit bears a series of well-defined transverse ridges called **subdigital lamellae**. These are absent in members of the family Rhizomyidae, and replaced by smooth or randomly creased skin (Figure 4.21).

Figure 4.21 The manus (fore-foot) of a murid rodent (*Rattus rattus*; left) and a rhizomyid rodent (*Rhizomys pruinosis*; right). Note the subdigital lamellae under the toes of the murid rodent only.

The pattern of fur colouring on the fore-limb and manus varies somewhat among the murid rodents. In the most common condition, the general fore-limb colour extends onto the upper surface of the wrist, giving way on the lower wrist and digits to a contrasting zone of white or transparent hairs (Figure 4.22a). In a few species (e.g. *Bandicota* spp.), the fore-limb fur colour extends over the manus to part-way along the digits (Figure 4.22b). Even less often, as seen in *Rattus nitidus*, the white fur of the manus extends partway up the fore-limb, forming a more elongated 'glove' (Figure 4.22c). A final variant,

found in species of *Leopoldamys*, has a well-defined strip of dark fur extending down the centre of the wrist (Figure 4.22d).

Figure 4.22 Variation in the patterning of the manus (fore-foot) among murid rodents: (a) *Rattus rattus*; (b) *Bandicota indica*; (c) *Rattus nitidus*; and (d) *Leopoldamys sabanus*.

Hind-limb

The hind-limb of rodents is used more exclusively for locomotion and it shows more obvious patterns of

specialisation. Among the Southeast Asian murids, this is most clearly expressed in the morphology of the **pes** or hind-foot. As a general principle, terrestrial rodents have long, narrow feet that enhance running speed, while arboreal rodents have short, broad feet that provide better purchase and are also better suited for grasping (Figure 4.23)

Figure 4.23 Pes (hind-foot) shape among murid rodents, contrasting a highly terrestrial species (*Bandicota savilei*; left) with a highly arboreal one (*Chiromyscus chiropus*; right). Note differences in relative digit versus heel lengths, claw size and shape, and plantar pad size.

The pes of murid rodents has five distinct digits, the innermost digit being the shortest (Figure 4.24). All digits have subdigital lamellae and apical pads as described for the manus. The number of subdigital lamellae (as counted on the central digit) is relatively constant (±1–2) within each species but differs

between them. This partly reflects the length of the digits (e.g. low counts of 4–6 in the short-toed diggers such as *Bandicota* spp.; counts of 7–8 in most other species). However, some species also seem to have unusually small and numerous lamellae, with very high counts obtained in some species of *Maxomys* and *Chiromyscus*.

In most species, well-formed claws are present on all five digits (see Figure 4.24). Some of the more highly arboreal forms have a small, flattened nail on the innermost digit only (see Figure 4.25)—or, in one species, *Vandeleuria olearea*, on both the innermost and outermost digits. The form of the apical pads and claws mirrors that seen in the

manus—specialised diggers tend to have small apical pads with large, forward-projecting claws; while arboreal and more generalised terrestrial forms have prominent apical pads and sharp, recurved claws.

The plantar surface of the pes usually has six large fleshy pads—four **interdigital pads** arranged in an arc at the base of the digits, and two **metatarsal pads** ('inner' and 'outer') situated further back on the sole (Figure 4.24). In many species, the two outermost interdigital pads have a small accessory pad fused to their outer margin, and this sometimes gives them an upside-down U-shaped appearance (e.g. *Chiromyscus chiropus*; see Figure 4.23). The **inner metatarsal tubercle** in many murids is elongated and curved posteriorly, giving it a comma-like shape (Figures 4.24 and 4.26). In a few groups of murids (e.g. *Mus* spp.), the skin between the

primary plantar pads is covered in fine tubercles; more normally, it is smooth (Figure 4.26).

The plantar pads in terrestrial species (especially those that habitually dig) tend to be relatively small and low, and their surfaces generally appear smooth or weakly striated. In contrast, the pads of arboreal forms are usually larger, more prominent and more obviously striated (see Figure 4.23). These adaptations present obvious advantages for climbing.

Figure 4.24 Left pes (hind-foot) of *Leopoldamys sabanus*, illustrating the major morphological features of the plantar surface. Note that all digits bear sharp claws.

Figure 4.25 Left pes (hind-foot) of *Chiromyscus chiropus*, showing the flattened nail on the first digit of this species.

Figure 4.26 Left pes (hind-foot) of *Rattus norvegicus* (left) and *Mus cookii* (right) illustrating contrastings of (i) inner metatarsal tubercle—elongated in *R. novergicus* versus rounded in *M. cookii*; and (ii) the skin between the interdigital pads—granular in *M. cooki* versus smooth in *R. norvegicus*.

The skin on the upper surface of the pes is covered in very fine, scale-like structures and it is also sparsely covered in a layer of fur, with hairs extending onto the toes. The skin varies in colour from essentially transparent (flesh-coloured) to white and dark brown or grey. It often appears to be speckled with colour due to the presence of scattered, pigmented scales.

The fur on the pes is sometimes completely dark (e.g. *Bandicota indica*) or pure white (e.g. *Rattus nitidus*), but more often it consists of both pale and dark hairs. These may be randomly mixed, giving the upper surface of the foot a grizzled appearance (e.g. *Bandicota bengalensis*), or they may be segregated into a distinct pattern. The most common pattern is a narrow band or wedge of dark hairs extending forward from the ankle, along the outer side of the pes (Figure 4.27). In some species, the dark hairs are concentrated on the front of the pes, around the bases of the digits. As in the manus, the toes are usually clothed in white or clear hairs; however, even

Figure 4.27   Upper surface of the left pes (hind-foot) of *Rattus sikkimensis*, illustrating the common patterning of a wedge of dark hairs extending forward from the ankle.

these hairs are dark in some examples of *Bandicota indica*. A few species have pale, gingery fur on the upper surface of the pes (e.g. *Chiromyscus chiropus*, some *Rattus rattus*), sometimes in combination with dark brown or black hairs.

Scrotal sac

In adult males of most murid rodent species, the **testes** are held in a prominent scrotal sac that overhangs the base of the tail and hides the anus from view (Figure 4.28; see also Figure 4.2). The scrotal sac is most prominent in the smaller species, such as *Mus* spp., in which the testes are largest relative to body size. However, some much larger-bodied species (e.g. *Rattus* spp.) also have quite large testes (length in adult 20–30 mm) that occupy a prominent scrotal sac. In contrast, the species of *Bandicota* and *Nesokia* have relatively small testes (rarely more than 25 mm in length) and these occupy a poorly developed scrotal sac that barely projects past the anus. The more protected location of the testes in these species may be related to their burrowing habits.

The **epididymal pouch** (see Figure 4.28) is a small posterior extension of the scrotal sac that houses the paired **cauda epididymes**, the organs in which mature sperm are stored. The epididymal pouch is prominent and darkly pigmented in most murids. In contrast, it is poorly developed and weakly pigmented in the species of *Bandicota* and *Nesokia*.
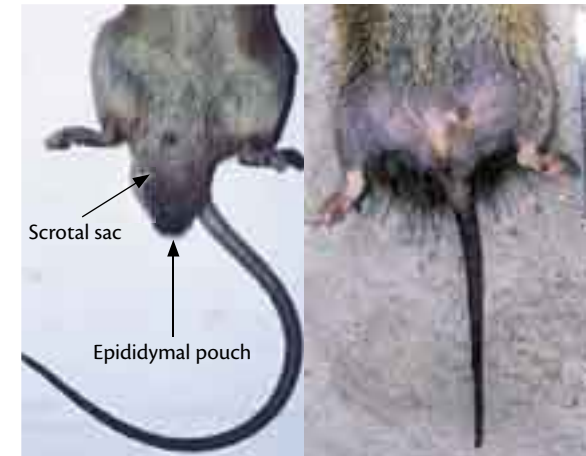
Figure 4.28   Scrotal region of two adult male murid rodents with proportionally very different sized testes: *Rattus exulans* (left) with relatively large testes, has a large scrotal sac and prominent epididymal pouch; *Nesokia indica* (right) with relatively small testes, has a small scrotal sac and indistinct epididymal pouch.

Mammae

The number of teats differs between some genera and species of murid rodents (Table 4.1). This makes the mammae useful for taxonomic diagnosis, but generally only for adult females. As mentioned earlier, the mammary formula is usually expressed as the sum of three parts: pectoral + postaxillary + inguinal (e.g. 1+2+2 for *Mus domesticus*; see Figure 4.4). Although this system is adequate for most species, some individuals of *Bandicota bengalensis* have numerous teats in more or less continuous series along either side, sometimes as many as 18 on one side alone (Figure 4.29). This is best expressed as a total count.

Some other species also show individual variation in teat number. However, in most cases this variation affects only the postaxillary teats. For example, *Rattus rattus* may have one or two teats in this position, sometimes with different numbers on opposite sides of the same individual. A variable mammary formula can be written as 1+1/2+3.

Cranial features

Rodents can also be identified from features of the skull and teeth. However, this is really a specialist task and it is beyond the scope of this book to review all of the diagnostic characters.

Anyone who is seriously interested in conducting taxonomic research on rodents should prepare some representative skulls, using the methods described on page 37. You will also need to learn the complex terminology used by rodent taxonomists to identify all of the individual features of the molar teeth and the cranium. Some useful introductory references are given under Further reading.

Figure 4.29 Adult female of *Bandicota bengalensis* illustrating the unusually large number of teats that often occurs within this species.

Table 4.1 Distribution of the major pest rodent species according to mammary formula.

Mammary formula	Species included
0+1+2	some *Rattus steini*
0+2+2	some *Rattus steini, R. mordax, R. praetor*
1+1+2	*Berylmys bowersi, Cannomys badius, Nesokia indica, Rattus exulans*
1+2+2	*Berylmys berdmorei*, all Southeast Asian *Mus* spp., *Mus musculus* Group
1+0+3	some *Rhizomys pruinosis*
1+1+3	*Rattus losea*, some *R. rattus, R. tiomanicus*, some *Rhizomys pruinosus, Rhizomys sinensis, Rhizomys sumatrensis*
1+2+3	some *Bandicota bengalensis, B. indica, B. savilei, Rattus argentiventer, R. nitidus, R. norvegicus*, some *R. rattus, R. sikkimensis, R. turkestanicus*
numerous	some *Bandicota bengalensis*

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CHAPTER 5

Population studies

Introduction

Rodent population studies attempt to document and explain variation or changes in the abundance of one or more species. These studies form the basis of any ecologically based rodent management system, as they help us to understand the major factors that control or regulate the population growth of pest rodent species and to identify the vulnerable points in the system that might allow effective intervention.

The population abundance of any individual species is determined by the numbers of births and deaths in a given area, and by the number of animals moving into (immigration) and out of (emigration) that area (see Figure 5.1). Each of these factors may be influenced by seasonal or longer-term climatic cycles, fluctuations in the abundance of food or predators, or changes in land-use patterns. The population

abundance of other species that compete for food or space may also be important.

The most basic type of population study is one that simply documents changes in animal abundance through time and space. This information can be obtained by taking a census of population size at various localities or at various times. Methods for
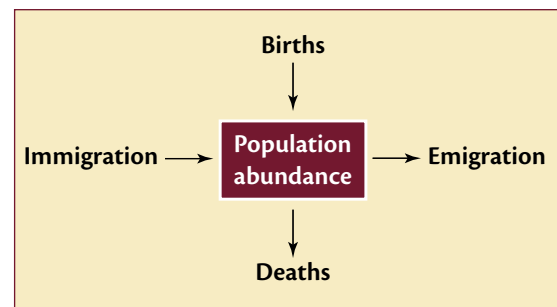
Figure 5.1 Simple conceptual model of the four factors that determine population size in a given area.

carrying out a population census are described in this chapter.

Census data can provide useful insights into the relationship between population abundance and some potential causal factor such as variations in rainfall or temperature, or between population abundance and crop damage. However, census data alone generally will not provide any real insight into the underlying ecological dynamics of the system. To understand why population abundance varies in time and space, it is necessary to not only study the changes in population abundance, but also study each of the main factors—breeding activity, mortality (including predation) rates, and movements. In Chapters 6 and 7, we describe methods for studying reproduction and movement of rodents, respectively. We do not cover methods used to estimate mortality rates, or to study the impact of predators. However,

these topics are covered in some of the general references listed under Further reading at the end of this chapter.

There are two main approaches to studying the abundance of animals in the environment. The first approach is to estimate the actual population size or **population density** (number of animals per unit area). If these estimates are taken simultaneously at different locations or repeatedly at one location, you will be able to study variations in population size through time or space. However, methods for estimating actual population density are laborious (see below) and before embarking on such a study, it is wise to ask first whether such data are really needed. For comparative studies, the alternative approach of taking **relative estimates of abundance** may be both adequate and far more cost effective. These simpler methods will be described first.

Relative estimates of abundance

Relative estimates of abundance do not give any absolute value for population size but they do allow you to make comparisons between localities or between time periods. One of the simplest measures of relative abundance is **trap success**, already introduced in Chapter 3. However, other relatively simple and inexpensive methods such as the use of tracking tiles, census cards, visual surveys and active burrow counts are also worth considering, especially if these methods are used in combination. Each of these methods is described briefly in the following pages.

Trap success

Trap success is usually calculated for single-capture traps (either live- or kill-traps) as the number of rodents captured divided by the total number of traps set. This value is usually multiplied by 100 to give percentage trap success. For example, if trapping occurred for 3 consecutive nights with 50 traps set each night, and the number of rats caught on each night was 5, 7 and 3, respectively, then the total trap success is (15 rats/150 traps) × 100 = 10% trap success.

Various adjustments are sometimes made to the raw trap-success figure. As mentioned in Chapter 3, one adjustment that is commonly made is to subtract the number of 'null' traps, i.e. traps that were sprung without making a capture, from the total number of traps set. A similar, but more sophisticated, adjustment takes account of the impact of occupied traps on overall trap success. Every time an animal is caught, there is one trap fewer available to make more captures. The number of active traps thus reduces progressively throughout the night. Caughley (1977) recognised that this situation reflects a simple frequency–density relationship approximated by the equation:

$$\text{Adjusted trap success (ATS)} = \ln\left(1 - \frac{\text{animals caught}}{\text{number of traps}}\right) \times (-100)$$

A simple, step-by-step method for use with a calculator is as follows:

- divide number of animals caught by number of traps, e.g. 22/52 = 0.423 (unadjusted or raw trap success)
- store the answer in the calculator's memory
- subtract memory from 1 (i.e. 1 minus recall memory), 1 − 0.423 = 0.577
- take the natural log (ln) of that = −0.55
- convert that to a percentage, −0.55 × −100 = 55% (adjusted trap success or ATS).

Tracking tiles

Tracking tiles are flat squares of metal (Figure 5.2), ceramic, vinyl or wood (usually around 250 × 250 mm) that are covered with a layer of grease or mud and placed in positions where rodents are likely to be moving during the night. The following morning, the tiles are inspected for signs of rodent activity. This may take the form of complete footprints, a tail swipe, or just the marks of the rodent's claws. It is generally not possible to identify individual footprints to species. It is usually also

difficult to tell how many rodents have visited the tile, so most people just record the activity as 'yes' or 'no'.

In rice fields, an alternative to tracking tiles is to smooth a set length (e.g. 1 or 2 m) of mud along a bund between rice fields. The number or length of rodent track-ways can be taken as a measure of activity. To be effective, the mud needs to be moistened and smoothed late in the afternoon or early in the evening, after the sun has fallen. This method is not suitable for use during periods of heavy rain. At such times, the use of grease is preferable as it is unaffected by rain.

Tracking tiles are sometimes used in combination with single-capture traps, with tiles and traps interspersed along a trap-line or within a trapping grid. Although neither method gives an absolute estimate of abundance, the combination does provide a useful independent measure of the effectiveness of the single-capture traps.

Figure 5.2   Squares of metal (250 × 250 mm) with grease (tracking tiles). Footprints can be seen on one tile, on the right.

Census cards

Census cards are used to estimate relative abundance of the house mouse in grain-growing areas of Australia (Figure 5.3). Squares of paper (100 × 100 mm) are marked with a grid and then soaked in vegetable oil (or canola oil), which is attractive to mice. The paper squares are pegged to the ground with metal wire.

Census cards are set out in lines of 10 or 15, with 10 m between each card. Lines should be set along a range of habitats such as channel banks, small banks, large banks and along edges of paths or roads. The following morning, the number of squares consumed by the mice is recorded. The average percentage of each card consumed is calculated as an index of relative abundance. Census cards tend to be consumed more when there is little alternative, high-quality food available. The method is thus subject

Figure 5.3   Census cards, before and after one night. Approximately 40% of the card on the right was consumed by mice.

to some of the same limitations as the use of baited single-capture traps. Census cards cannot be used during periods when heavy rainfall is expected.

Burrow counts

The number of rodent burrows in a given area is a useful index of the relative abundance for many ground-dwelling species. It is obviously of no use for tree-dwelling species or those that build grass or leaf nests on the ground. In some cases, the burrows of different species can be identified from their size or morphology, but this will depend on the number and variety of species found in any area.

In taking burrow counts, it is important to distinguish active from abandoned burrow systems, and to distinguish rodent burrows from those excavated by crabs or other creatures. A technique used in Indonesia involves locating all burrows along a transect of a given length. Each burrow entrance is plugged with a thin layer of mud (Figure 5.4). It is important to mark the location of all burrows so that they can be found the next day, or to draw an accurate map. The following day, the number of freshly reopened entrances is recorded. Footprints made by the rodents are often seen in the mud. In the Mekong Delta in Vietnam, dry grass is used instead of mud to seal burrow entrances.

The number of reopened burrows does not tell you exactly how many rodents are present along

the transect. In many species, the burrow systems may have multiple entrances and only some may be reopened. Some rodents may have chosen to remain within the burrow, while some burrows may house more than one rodent, especially during the breeding season. Many studies have shown strong seasonal changes in the average number of animals per burrow system. Excavating a set number of burrows per sampling period to estimate the occupancy rate can reduce this uncertainty.
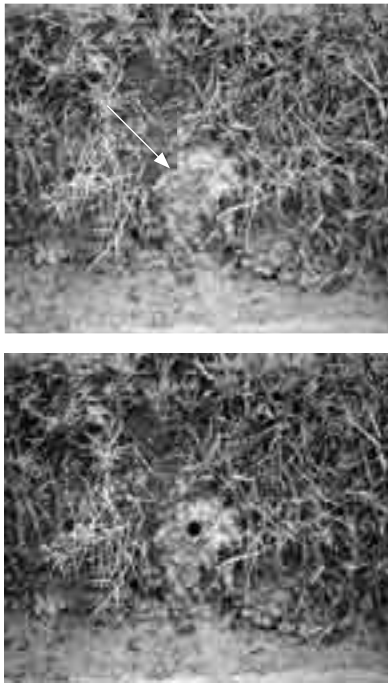
Figure 5.4   (Top) Rat burrow covered with mud (arrow). (Bottom) Rat burrow that has been sealed with mud and reopened.

Visual surveys

Under some conditions, it is possible to count the number of rodents that are active at night in a particular location. This is usually done with a torch—an activity that is known as 'spotlighting'. The basic method involves walking at a constant pace along a transect and counting the number of active rodents that are detected either from their movement or by their eye shine (usually glowing red). With experience, it may be possible to identify different rodent species from sightings of this kind.

For spotlighting to be an effective and useful tool, the method must be standardised. The observer, the observer's pace, the route taken, the time of night and the strength of the torch must all be kept as constant as possible. Factors that may interfere with the ability to see or hear the animals, such as rain or dense plant cover, should also be recorded for each survey period.

Calibrating relative estimates of abundance

All relative estimates of abundance can be made more useful if they are 'calibrated' against estimates of actual population densities, as suggested above in the case of burrow counts. However, it is also important to realise that the appropriate calibration factor may vary between seasons or stages in a cropping cycle. For example, methods that rely on baits (single-

capture traps and census cards) will almost certainly have a lower relative success rate during periods when the local environment contains abundant alternative food.

One approach worth considering is to use a variety of these methods for estimating relative abundance in combination. During the course of a full year's cycle, each method can be expected to provide different kinds of information that, when added together, might give a better overall picture of the relative intensity of rodent activity through time and across a range of different habitats.

Estimates of population size

With rodents, it is generally not possible to count all of the animals in a population. The next best thing is to estimate the number of animals in a given area, using one or more of the following methods.

One way of estimating population size is to convert relative abundance data obtained from trapping or from visual surveys into population density values. To do so, you will need to make some fairly large assumptions. For trapping data, you will need to estimate the trappability of each species—that is, the proportion of a population that you would expect to enter the traps each night. As mentioned earlier, this value may vary seasonally, depending on both the availability of other foods around the traps and

on the activity pattern of the animals (which in turn might reflect breeding, dispersal activity etc.). For visual survey data, you will need to estimate the proportion of a population that you would expect to be active at any one time, the likelihood of observing an individual animal, even if it is active, and the width of the transect (i.e. the distance of reliable detection). Visual surveys are often used to estimate population densities of large, easily spotted animals. For rodents, the error factor is probably too large to make the method useful, except perhaps in very open habitat.

Capture–mark–release methods are the most commonly used technique for estimating population size. As the name suggests, these methods all require that captured animals are marked in some way and then released at the point of capture. After one or more nights, the locality is trapped again. Population size is then estimated by comparing the number of recaptures with new captures in the sample. If the trapping is continued over several nights, the proportion of new captures can be compared with the numbers of animals caught once before, twice before etc. Various methods are available for estimating population size from the recapture data. However, before moving on to these, we will review some of the basic equipment and methods used in a capture–mark–release study.

Equipment

When collecting data for a capture–mark–release study, the following equipment is required (Figure 5.5):

+ a large bag to hold the rodent; this can be plastic or cloth—a cloth pillow case is ideal
+ rulers to take length measurements; transparent plastic rulers are good because it is easier to see what you are measuring; steel rulers are easy to disinfect and will last longer
+ a balance for weighing rodents; a spring balance (e.g. Pesola) is best, but any balance which is portable and hardy will be sufficient (ensure the weight range is suitable for the animals you are trapping; in many regions you may need two or more Pesola balances, one for mice (to 60 g), one for rats (to 300 g) and one for *Bandicota* (to 1 kg)
+ individually numbered ear-tags or an ear-punch for marking animals; an applicator is also useful
+ a simple taxonomic key for species identification in the field (see Chapter 11)
+ data sheets, data codes, pencils, pens; data must be recorded in a systematic, logical and consistent way. Standard data sheets and codes **must** be used—this way, no information will be forgotten and comparisons can be made with other sites and countries. A sample data sheet is provided in Appendix 1.

Figure 5.5   Equipment needed for a live-trapping study, clock-wise from top: field manual with codes and taxonomic key, field data sheets, pencil, plastic ruler, ear-tags, ear-tag applicator, Pesola spring balance and cloth bag for holding rodents.

It is a good idea to carry duplicates of essential equipment. We find that it is best to carry everything in a small bag that attaches around the waist.

If possible, two people should work together to collect population data. One person handles the rodents and takes measurements, while the other person records the data.

Marking techniques

Most capture–mark–release studies require that every captured animal is assigned a unique number. This number must either be attached to the animal in some way, or else encoded into a marking system that can be applied to the animal. The numbers or coded marks must remain visible on the animal for

the duration of the study and also must have little or no impact on the animals' behaviour, fitness or survival. Three alternative methods for marking are described here.

Ear-tagging

Some capture–mark–release studies use metal ear-tags that are imprinted with a four-digit number (0001 to 9999) (Figure 5.6). The tags have one short side with a point and two longer sides, one with the imprinted numbers and one with a slot (for attachment). The tags are easy to apply, once the correct technique has been demonstrated, and they are easy to read on subsequent captures.

Figure 5.6   Ear tag in a mouse.

The pointed side of the tag is pushed through the base of the ear, just under the fold of cartilage. It is best to have someone show you the correct location

as the animal can easily rip the tag out if it has not been applied correctly. The point of the tag is then fed through the slot and flattened to reduce the risk of anything catching under the tag and causing the ear to rip. If ear-tags are applied in the correct position and with care, they will generally stay in place for many months and have no effect on the animal's behaviour.

Ear-punching

This method is of limited use for capture–mark–release studies because of the limited number of individual marks that can be applied (Figure 5.7). However, the method is mentioned here because it is sometimes useful to mark groups of animals with a single type of mark. Examples would be a study involving trapping every second month, where animals are marked according to the census period in which they were first captured (which will provide information on survival rate between trapping periods) and a study in which animals are

Figure 5.7   Right ear-punch in a mouse.

marked according to the habitat in which they are first and subsequently captured (to analyse patterns of movement between habitats).

Ear-punches should be made with a good-quality ear-puncher of the kind used to mark laboratory animals. Ear-punches are less obvious than some other marking techniques and they probably have little impact on fitness. However, natural wounds to the ears can sometimes lead to incorrect identification.

The codes given in Table 5.1 and illustrated in Figure 5.8 show how an ear-punch numbering system works. For example, for census or habitat number 4, you would ear-punch all animals in the lower position of the left ear.

Table 5.1   Combinations of ear markings.

Position on ear	Number
Lower right	1
Upper right	2
Upper left	3
Lower left	4
Lower right + upper left	5
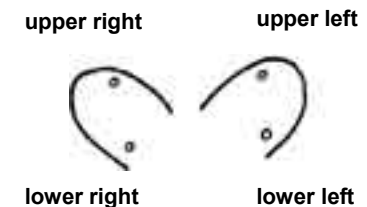
Figure 5.8   Positioning of ear-punches.

More complex combinations of punches can be used to increase the number of codes. However, care must be taken to ensure that the codes can be read without introducing errors.

Other methods

A range of alternative methods are available that could be used on rodents in field studies. These include ear slits, colour markings, tattoos, and shavings. Some references are provided at the end of this chapter (Further reading).

Calculating population size from capture–mark–release data

A wide range of methods is available to estimate population size from capture–mark–release data. Many of these methods are very sophisticated and they all rely on critical assumptions, including:

- the population is closed to additions (births or immigration) and deletions (deaths or emigrants)
- all animals are equally likely to be captured in each sample
- marks are not lost and are not overlooked by the observer.

The second assumption is often called the assumption of 'equal catchability'. This assumption is unlikely to hold true for many wild populations of animals, where the probability of capture is likely to be influenced by age, sex, social status, trap placement in relation to individual territories, and prior history of capture (e.g. 'trap-shy' versus 'trap-happy' individuals). A trap-happy animal becomes easier to catch after being caught once; a trap-shy animal becomes more elusive.

Many of the available methods also depend on high recapture rates (>20%). In our experience, recapture rates for Southeast Asian rodent populations are typically very low (often less than 1%), hence these methods will not produce useful population estimates. One of the simpler methods, called the Petersen Estimate, is explained in Box 5.1. This method is only appropriate where recapture rates exceed 5%.

To convert estimates of population size into a population density, we need to include some estimate of the area that is effectively sampled by the trapping grid. For a very sedentary species, this area my not be very much larger than the trapping grid itself. However, for more mobile species, the effective trapping area may be considerably larger. To convert a population estimate into an estimate of population density, we therefore need some information on the movement patterns of the particular species. Methods for studying movement patterns of rodents are described in Chapter 7.

Box 5.1   The Petersen Estimate for calculating population size

This is one of the simpler methods for estimating population size (number of animals per unit area). The calculations can be done on a calculator or using a spreadsheet program such as Excel on a computer. The Petersen Estimate can be calculated easily following the steps below:

- 1st trapping—mark animals caught and released (M)
- 2nd trapping—capture marked (m) and unmarked animals (total = n)
- Calculate proportion of population marked (Y):

$$Y = \frac{\text{Number marked } (m)}{\text{Number caught } (n)}$$

- Estimate population size:

$$N = \frac{\text{Size of marked population } (M)}{\text{Proportion of population marked } (Y)}$$

There are some important assumptions for this method:

- marked and unmarked animals are captured randomly
- marked animals are subject to the same mortality rate as unmarked animals
- marks are not lost or overlooked.

Here is a simple example to illustrate this method. Trapping with 50 traps set in a grid produced 15 rats caught, marked and released on the first night. On the second night, 13 rats were caught, including 5 marked animals. For this example, $M = 15$, $m = 5$ and $n = 13$, hence:

$$Y = \frac{m}{n} = \frac{5}{13} = 0.385$$

$$N = \frac{M}{Y} = \frac{15}{0.385} = 39$$

The estimated population size is 39 rats.

A range of more sophisticated methods is available free from the Internet (see Further reading).

Further reading

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CHAPTER 6

Reproduction and growth in rodents

Introduction

Breeding is the main reason why populations increase in size. This is especially true of many rodent species that are capable of rapid population growth, especially when conditions are favourable. Rapid population growth is generally due to a combination of two factors—namely, a **high reproductive potential** and a **short period of maturation** to sexual maturity.

The reproductive potential of a species can be thought of as the possible number of offspring that a typical female can produce during her life. This is affected by four main factors:

- length of the gestation period (i.e. the period between conception and delivery)
- litter size (i.e. number of offspring per delivery)
- length of time between delivery and the next conception

- the reproductive life of females (i.e. the period of time from the first litter to the last litter or until death).

Rodents typically have short gestation periods, with high litter sizes and an ability to fall pregnant again within a few days of delivery. These factors alone would ensure a high reproductive potential. However, many rodents also attain sexual maturity at very early ages, due mainly to rapid growth during the first few weeks of life. This latter factor is particularly important in allowing a population to respond to relatively short-term increases in the availability of food. The particularly short period to sexual maturity of many murid rodents is without doubt one of the main reasons why so many of the major agricultural pests belong to this one family of mammals.

In this chapter, we provide information on the reproductive anatomy of rodents, the changes that occur both during and after pregnancy, and the process of growth and maturation of the young. We also discuss some key reproductive parameters that will assist you to make sense of your observations of reproductive activity within a population of rodents.

Basic reproductive anatomy

The external features of the reproductive system were described in Chapter 4. Here we will concentrate on features of internal anatomy. These are usually examined by making a careful incision along the midline of the belly, starting from just below the ribcage and running down to just above the genital papilla. Care must be taken not to cut into the intestine or any embryos that may be present in the

abdominal cavity. To this end, it is best to make the incision with a pair of sharp scissors rather than with a scalpel blade.

Male reproductive tract

The male reproductive tract (Figure 6.1) consists of the paired testes, epididymes and ducti deferens, accessory sex glands and the centrally located penis. Elements of the **urinary tract** are also labelled on Figure 6.1—notably the paired kidneys and ureters, and the centrally located bladder.
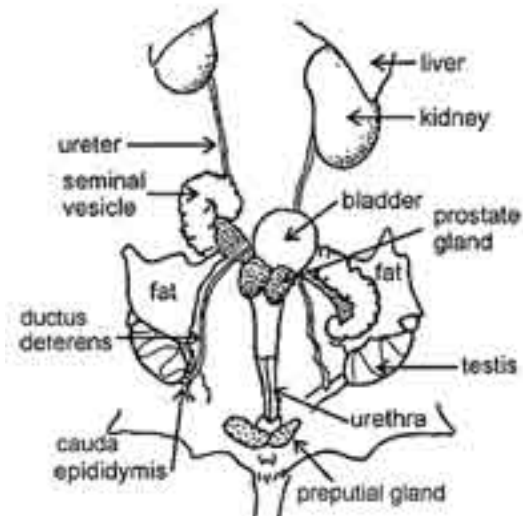
The **testes** produce sperm and also synthesise and release male sex hormones. In a juvenile rat, they are located high in the abdomen, just behind the kidneys. With maturation, the testes enlarge in size and move backwards—first into a position at the base of the tail, and finally into the scrotum. Sperm are produced in the testes, then move into and through the **epididymes**, where they mature and are stored. Contractions associated with sexual stimulation move the sperm out of the epididymes and through a tubular transport tract beginning with the **ducti deferens**. Secretions are added by various **accessory sex glands** (e.g. prostate glands and seminal vesicle) to produce an ejaculate that exits the body along the urethra, a canal supported by the penis.

In many rodents, large **preputial glands** are present on either side of the penis. These release strong-smelling fluids into the urine that are important in various kinds of behaviour, including scent marking.

Female reproductive tract

The female reproductive tract (Figure 6.2) consists of the paired **ovaries**, the **Y-shaped uterus** within its elongate **uterine horns** and basal stem, and the centrally located **vagina**. The urinary tract consists of the same elements as in a male. In many rodents, females also possess large **clitoral glands** on either side of the genital papilla, with ducts opening into the genital papilla.

Juvenile female rodents have an **imperforate** vagina, which is to say that it is sealed over by a thin layer of skin called the **hymen**. The hymen appears as a small, shiny patch of skin just behind the genital papilla. Internally, the uterine horns are narrow and thin-walled, and have an inconspicuous blood supply. The ovaries and fallopian tubes are also small.

With the onset of sexual maturity, the ovaries enlarge and start to secrete female sex hormones. The effects of these hormones are felt throughout the body:
- the ovaries begin to produce mature eggs
- the uterine horns elongate and become thicker, and develop a more conspicuous blood supply

Figure 6.1   Male reproductive and urinary tracts of a typical murid rodent (redrawn by M. van Wensveen, after Cook 1965).
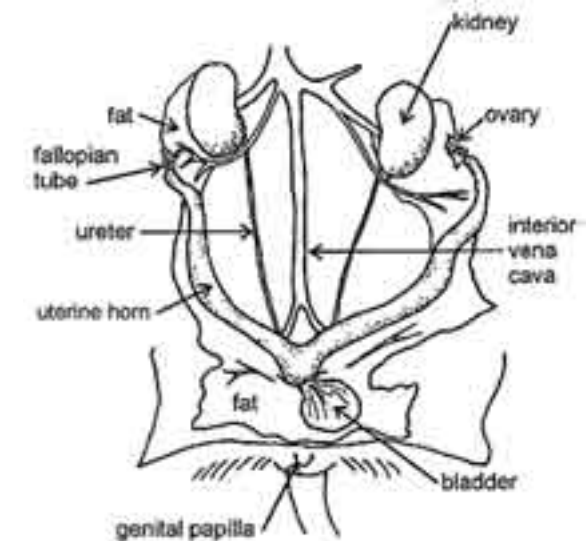
Figure 6.2   Female reproductive and urinary tracts of a typical murid rodent (redrawn by M. van Wensveen, after Cook 1965).

- the hymen is resorbed, resulting in an open or **perforate vagina**
- the teats and associated mammary tissue enlarge.

Pregnancy and embryonic development

Mature eggs are released from the ovaries into the **fallopian tubes** (see Figure 6.2) every four days on average in the pest species of *Rattus*, every five days in *Mus domesticus* and every 3–8 days in species of *Bandicota* (see Chapter 11 for information on particular species). If conception occurs, the fertilised eggs remain in the fallopian tubes for up to three days before they move into the adjacent uterine horn. Here, the fertilised eggs, now at the blastula stage of development, move into small pockets in the wall of the uterus where they will undergo implantation and embryonic development. A critical part of this process is the formation of a **placenta** between each embryo and the wall of the uterus, starting around day 7. The placenta establishes a blood supply (via the **umbilical cord**) between the female and each developing embryo, and provides the embryo with nutrition for rapid growth.

The **gestation period** (the period from conception to delivery) varies greatly between different groups of rodents. However, among the major groups of pest rodents, the gestation period is usually very short, falling between 19–25 days.

If conception occurs while a female is still lactating from a previous litter, the blastulae may not implant immediately after entering the uterus. Instead, they may remain in a period of suspended development, until triggered by some hormonal cue to resume the process of implantation. In laboratory rats (*Rattus norvegicus*), implantation is commonly delayed by a period of 6–9 days (i.e. occurs on day 8–11 after mating), but in some 10% of cases this is extended to a delay of 12–19 days. The length of this delay is related to the strength of the sucking stimulus and thus to the previous litter size. The average gestation period for lactating laboratory rats is 26 days, compared with 22 days for non-lactating females. In laboratory strains of the house mouse (*Mus musculus domesticus*), the delay in implantation is typically shorter, with only 5% extending implantation beyond day 7. The average gestation period for lactating mice is 24 days, compared with 20 days for non-lactating females.

The gestation period is divided into three time intervals called **trimesters**—each trimester making up approximately one-third of the total period. Here we illustrate the major stages of embryonic development for a rat with a total gestation period of 20–22 days, such as occurs in *Rattus rattus*.

Trimester 1

In the early stages of trimester 1 (up to days 5–6), the only evidence of pregnancy will be an obvious increase in blood supply to the uterine horns. At this stage, there is no obvious swelling of the uterine walls, hence it will not be possible to count the number of embryos. After 5–6 days, the embryos begin to interact directly with the tissues of the uterine wall, which responds by forming a series of distinct bulges, one for each embryo (Figure 6.3). These are easy to count, even though they measure less than 5 mm in diameter.
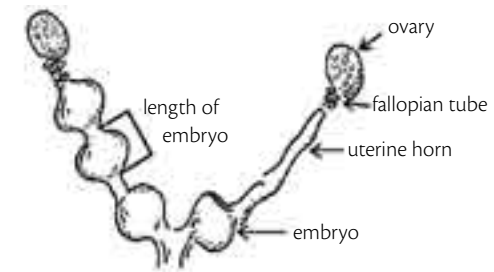
Figure 6.3 Condition of the uterus at late trimester 1 of embryonic development in a rat (after Theiler 1972). The recommended method of measuring the uterine swelling is also shown.

Trimester 2

This is a period of rapid internal development of the embryo, marked by the beginnings of the nervous, circulatory and alimentary systems, and of the skeletal column. Limbs initially appear as 'buds'

without distinct toes (Figure 6.4). The placenta also develops rapidly during this period to provide the nutrition needed for embryonic growth.

Trimester 3

In this trimester, the body grows rapidly in preparation for birth. Fingers and toes form and then separate, details of the ears, eyes and skin emerge, and elements of the circulatory system become visible through the pale skin (Figure 6.5).

Within a single species, a good relative measure of the stage of development is a simple linear measurement of a uterine swelling. We prefer to measure this parallel to the long axis of the uterine horn, as shown in Figure 6.3. Although this information is not yet available for many species, it can be gathered during the early part of a study for each of the captured species.

Embryos can fail in their development at any stage. If this occurs during trimesters 1–2 or early in trimester 3, the embryo will be resorbed by the uterus. A resorbing embryo will gradually decrease in size, while the remaining live embryos get larger. For this reason, it is sometimes possible to tell them apart. However, embryos that fail during the first trimester may be impossible to detect other than by microscopic examination of the ovary. Embryos that die during the last few days of pregnancy will be delivered as stillbirths.

Delivery of the young is triggered by further hormonal activity on the part of the ovaries. This causes the uterus to contract, leading to expulsion of the young, and to detachment of the placentae from the wall of the uterus. As each placenta pulls away, it leaves behind an open wound in the uterus, one for each embryo. The scar tissue that forms over each wound is called a **placental scar**. These scars are visible through the uterine wall (Figure 6.6).

Immediately after delivery, the placental scars are large and reddish brown, still with an obvious blood
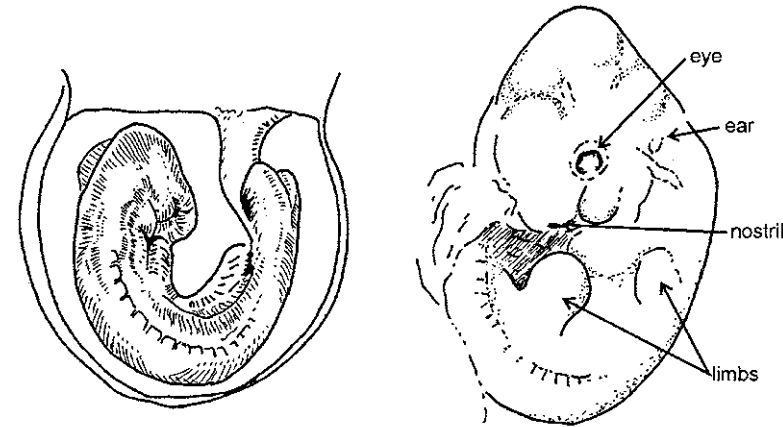
Figure 6.4   Early (left) and late (right) trimester 2 of embryonic development in rats (after Theiler 1972).
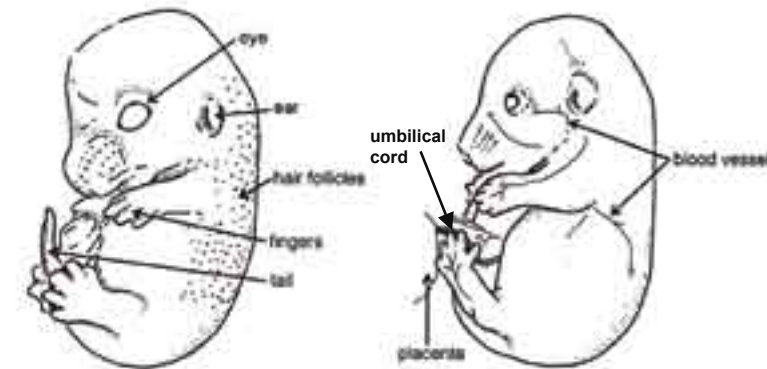
Figure 6.5   Early (left) and mid (right) trimester 3 of embryonic development in rats (after Theiler 1972).

supply. As the uterine horns thicken and narrow, the scars become smaller and take on a yellowish colour, and they lose their blood supply. Over time, they first become darker, and then smaller and less distinct. In the laboratory rat, placental scars generally remain visible throughout the adult life of a female.

By carefully examining the wall of the uterus, it is often possible to distinguish various sets of scars, based on differences in the size and intensity of the scars. However, where a female has experienced three or more pregnancies, it may not be possible to distinguish among scars of the earliest sets.
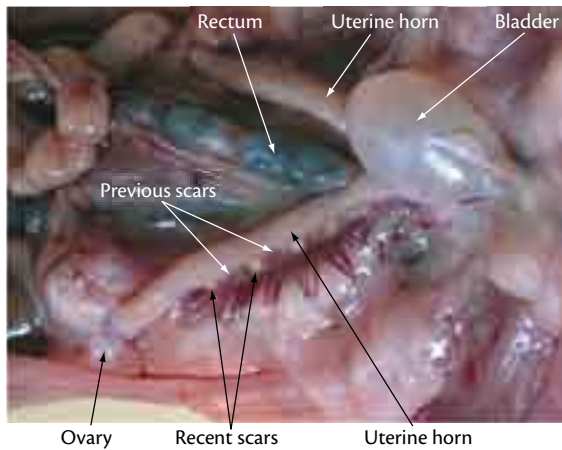
Figure 6.6 Dissection of a female rat showing a uterine horn with two sets of placental scars. The rectum (lower gut) has been pulled aside to expose the reproductive tract. The rich blood supply to the uterine horn is also clearly visible.

Growth and maturation after birth

The sequence and timing of maturation is very similar in all of the major pest species of rodents (Table 6.1). Newborn pups weigh just over 1 g in *Mus* species, and from 3–6 g in the pest species of *Rattus* and *Bandicota*. Newborns of all species are hairless except for small vibrissae on the snout. The eyes and ear canals are closed, and the external ear (pinna) is flattened against the head (Figure 6.7).

By the end of day 3, the pinna is usually erect. Fine dorsal hairs are visible to the naked eye by day 3–5 and teats are first seen on females as small, pigmented spots during the same period. At around day 7–9, the incisors start to erupt, the belly becomes covered with fur, and the pups begin to stand and walk. By day 13–14, the external ear canal is open and the pinna has become thinner and starts to enlarge. The eyes may open from day 11–12 in *Rattus* and *Mus*, but not until after day 14 in *Bandicota* species (Figure 6.8).

Pups of all species begin to take solid food brought into the nest by adults from the end of the second week. However, weaning is generally not completed until the end of week 3 or 4. After weaning, young rats and mice are effectively independent from the mother, although in some species they may continue to inhabit the same burrow complex for some time.

Table 6.1 Summary of key events and parameters in the development and growth of the young of some of the major rodent pest species. All values are in days, except for 'Weight at birth' which is in grams. A question mark (?) indicates that data are not available. Value prefixed with a 'c.' are approximate mean values.

Key event or parameter	*Rattus rattus*	*Rattus norvegicus*	*Rattus exulans*	*Mus musculus*	*Bandicota bengalensis*	*Bandicota indica*
Gestation period	20–22	20–25	19–21	19–20	21–25	21–25
Weight at birth (g)	3.0–6.4	4.3–6.2	2.8–3.1	1.2	3.5–5.0	?
Pinna of ear unfolds	2–5	2–3	2–5	2–3	?	?
Dorsal hairs are visible	3–5	3–5	3–5	2–3	?	?
Incisors erupt	7–12	9–13	7–11	5–7	?	?
Ear canal opens/pinna thins and elongates	10–14	c.12	11–14	c.13	?	?
Eyes open	11–15	13–17	12–16	12–14	14–18	18–22
Weaning occurs	23–28	21–28	21–28	c.24	c.25	c.28

Figure 6.7   Two- or three-day-old pups of *Rattus rattus*. No fur is present and the pinna has not yet unfolded. Each pup is about 50 mm in length.

Figure 6.8   A pup of *Bandicota bengalensis*, probably around day 13–14. The ears have started to thin and enlarge, but the eyes remain closed.

Attainment of sexual maturity

The length of time to sexual maturity varies between species. Although the onset of breeding is presumably limited by maturation of the reproductive organs, it is also influenced by many environmental factors, including the nutritional state of the young animals, aspects of social behaviour linked to population density and habitat structure, and possibly also crop maturation.

In wild populations of *Rattus norvegicus*, ovulation (oestrous) cycles may start at any time from 40 days of age but with a mean age of around 100 days. The vagina opens at any time over the same period, with a mean age of 72 days in a laboratory-reared population. In laboratory-reared males, testes descend into the scrotal sac during the period 15–51 days of age. Occasional mature sperm are present in the epididymes from around 50 days, but at much higher densities from 70 days onwards.

In *Rattus argentiventer*, females show an open or perforate vagina at a mean age of 33 days, and are pregnant at a mean age of 49 days. Males show descent of the testes from 26 days, with scrotal testes in more than 90% of individuals by day 40. Mature sperm are first observed in the epididymes from 70–90 days.

In laboratory mice (*Mus musculus domesticus*), vaginal perforation occurs between 28–49 days, with a mean of 35 days. Ovulation commences soon thereafter, but the first mating is often delayed by one or two weeks. Male mice become sexually mature slightly later than females.

Life span and menopause

Even when reared under optimum conditions, most rodents have a maximum life span of only 2–3 years. Females of *Rattus norvegicus* and *Mus musculus domesticus* stop ovulating (i.e. enter **menopause**) altogether between 12–18 months of age, although average litter size declines well before this age. Under natural conditions, very few individuals are likely to survive to such advanced ages.

Assessing reproductive activity from external characteristics

In capture–mark–release studies, we have to rely on external signs to assess reproductive activity.

In males, the only indicator of sexual maturity is the condition of the testes and scotal sac. Although the process of testicular enlargement and descent is a gradual one, we find it useful to distinguish three conditions of the testes:

- non-descended (scrotal sac undeveloped)
- partially descended (scrotal sac visible but not to the full extent, generally lacking a distinct epididymal pouch)
- fully descended (scrotal sac developed to the full extent, generally with a distinct epididymal pouch).

As noted in Chapter 4, the testes in species of *Bandicota* and *Nesokia* do not attain such a large size relative to body size as they do in the species of *Mus* and *Rattus*, and they would probably be scored as partially descended, even in fully adult individuals.

Males are generally not the primary focus for breeding studies. However, in a reproductive study you may wish to check for the presence of sperm in a urine sample. This is usually done by placing a urine droplet on a microscope slide and examining it at approximately 20–40 times magnification for evidence of motile sperm.

In females, the external signs of sexual maturity include an open vagina and enlargement of the teats. Signs of sexual activity are less obvious. The best indication is the presence of a yellowish vaginal plug that forms from vaginal secretions and ejaculate, and persists for 1–2 days after mating.

Pregnancy is usually evident by day 13, at which time the abdomen should be visibly enlarged. The teats also become more prominent during the final week or so of pregnancy. With some experience, it is possible to confirm whether or not a live animal is pregnant by using a technique termed **palpation**. To do this, run your thumb and first finger down each side of the lower abdomen, applying gentle pressure over the area covering the uterus and intestines. Embryos will feel smooth and round. In contrast, faeces will feel harder and more discrete.

It is generally impossible to detect a first trimester pregnancy by palpation, and it requires experience to accurately detect the second trimester embryos. By the time embryos have entered the third trimester they are much more obvious. Because an animal should not be recorded as pregnant unless the researcher is positive that this is the case, the palpation method will usually result in a serious underestimate of the pregnancy rate. Also be aware that palpation can lead to prenatal losses if it is not done gently.

A female that is currently nursing a litter of pups will have at least some enlarged teats and active mammary glands. Teats that are producing milk will be swollen at the base and often lack fur around their base; active status can be confirmed by gently squeezing the base of the teat until a droplet of milk is released. If the number of young is less than the number of available teats, some of the teats and associated glands may be inactive. For this reason, you may need to check more than one teat to confirm active lactation.

At the end of a period of breeding activity, the teats of an adult female rodent become smaller and the fur will grow back around the bases. However, the teats remain larger and more raised than those of a sexually immature individual.

We recommend that teats be scored as one of three categories (Figure 6.9):

- low and indistinct (fur at base)
- raised but not lactating (fur at base)
- raised and lactating (no fur at base).

Figure 6.9 Classes of teat on female rats. Top, teats are low and indistinct in a juvenile rat; middle, teats are raised but have fur around the base in a non-lactating adult; bottom, teats are raised and lack fur around the base in a lactating adult rat.

Assessing reproductive activity from internal characteristics

It is possible to obtain much more information on breeding activity where rodents are being sacrificed as a routine part of a population study.

In male rats, the condition of the testes should be examined to confirm whether or not the animal has reached sexual maturity. This is particularly important for those species (e.g. *Bandicota* and *Nesokia* spp.) where the scrotal sacs never become very prominent. A fully mature male in which sperm is being actively produced will have large testes, each with a prominent blood supply, and enlarged epididymes with highly convoluted, sperm-filled tubules. During periods when sperm is not being produced, the testes and epididymes reduce somewhat in size, the epididymal tubules become harder to see, and the blood supply to the testis becomes less obvious.

If greater certainty is needed, you can check for the presence of mature sperm in the **cauda epididymis**, the bulb-shaped part of the epididymis that projects posterior to the testis (Figure 6.1). This is done by carefully removing this part of the epididymis with sharp scissors and smearing the cut surface across a microscope slide. The sperm can be made more visible by application of a general stain such as gentian violet.

In female rats, the full picture of reproductive activity can only be obtained by examining both the ovaries and the uterus. However, accurate interpretation of the ovary generally requires histological examination, hence we will concentrate here on features of the uterine horns that can be observed by eye or with a dissecting microscope.

The uterine horns will generally fall into one of the following categories:
* very thin and short, with a poorly developed blood supply (Figure 6.10a). This condition is typical of juveniles; the vagina is either imperforate or very recently opened
* slightly thicker and more elongated, with a more obvious blood supply but without embryos or placental scars (Figure 6.10b). This condition is typical of an individual that is entering its first breeding season. The vagina should be checked for presence of a vaginal plug—this will indicate that mating has occurred
* thicker and with embryos present in one or both uterine horns (Figure 6.10c). Early-stage embryos take the form of small swellings. As the embryo and placenta develop, the uterine horns become wider and thinner-walled. If no placental scars are visible, the animal is probably in its first pregnancy. However, scars can be difficult to see when the pregnancy is in the third trimester and the wall of the uterus is very stretched (Figure 6.10d)
* elongate and wide, with very thin walls, but without embryos (Figure 6.10e). This condition is typical of the period immediately after delivery of the young. The placental scars appear as large discolourations of the uterine wall
* elongate and thick walled, with obvious placental scars but no visible embryos (Figure 6.10f). The uterine horns compact and thicken within a few days of delivery, ready to receive a new batch of fertilised eggs.

As mentioned earlier, recent placental scars are large and either reddish-brown or yellowish in colour. These become smaller and darker with time, but they probably remain visible through life. A female may have numerous sets of scars, and it should be possible to distinguish two or more sets based on their size and colour. However, once three or more sets are present, it may become difficult to distinguish between the earlier sets.

A count of the total number of scars is a useful measure of the reproductive output of a female. However, for two reasons, the total number of scars cannot be used as an exact count of the number of young produced during the life of the animal. Firstly, resorbed embryos also leave scars in the uterine horns. Inclusion of resorbed embryos would lead to an *overestimate* of the number of live young. Secondly, where there have been two or more separate pregnancies, it is possible that some of the later placentae have formed over the top of previous scars. In this case, a count of scars will *underestimate* the total number of young.
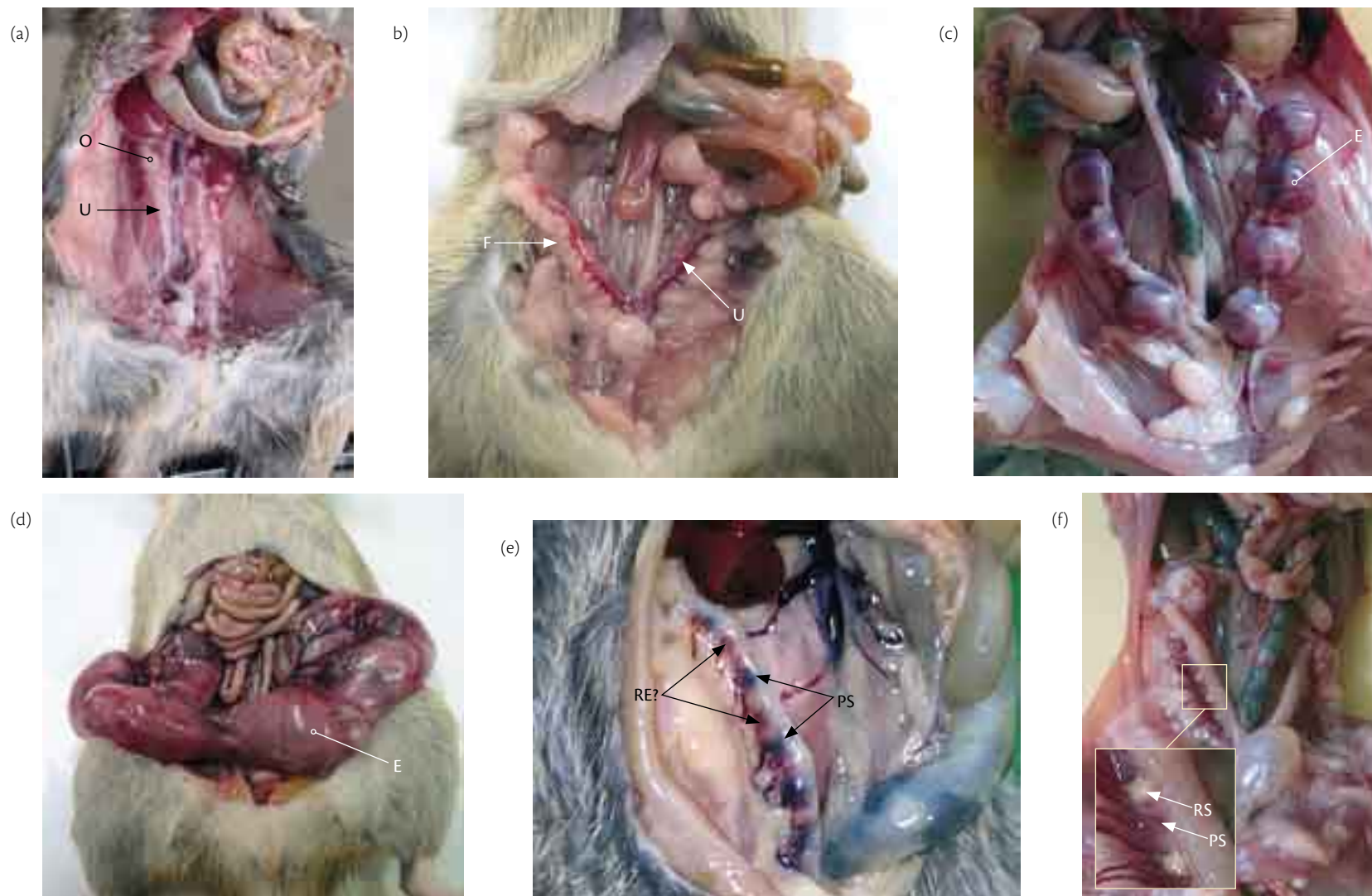
Figure 6.10   Comparison of the female reproductive tract: (a) juvenile—showing the ovary (O), and the uterine horn (U) which is very thin with an inconspicuous blood supply; (b) subadult entering its first breeding cycle—uterine horn is thicker, with conspicuous blood supply. Note extensive fat deposits (F) along uterine horns; (c) adult in second trimester of pregnancy with seven healthy embryos (E); (d) adult in advanced third trimester of pregnancy—the embryos (E) are clearly visible through the highly stretched uterine wall; (e) adult that has recently given birth. Within a few days, the large, dark placental scars (PS) will reduce in size and become paler. The smaller discolorations, each of which has a separate blood supply, probably represent resorbing embryos (RE?); (f) adult showing two sets of placental scars—the larger, yellowish scars (RS) are from a recent litter, while the smaller, darker scars (PS) are from a previous litter.

Key reproductive parameters

For population studies, it is important to determine the following key reproductive parameters.

Commencement and cessation of the breeding season

There is some debate over whether it is female or male reproductive activity or behaviour that really controls the timing of breeding activity. However, in either case, it is the occurrence of pregnancy in females that defines the **effective breeding season** and for this reason, we will maintain our focus on female reproductive condition.

Not all rodent species show a discrete breeding season. However, most of the pest rodents seem to stop breeding during periods of extended fallow, when food is scarce or of low quality.

The breeding season of a population can be said to start with the first successful mating after a period of non-breeding. Where the gestation period of a species is known, the date of conception of a pregnant female can be estimated by observing the trimester of development and counting back the likely number of days since conception. The breeding season is finished when the last litter of pups is weaned. This can be estimated directly by digging up numerous burrows and observing the growth stage

of the litters. Alternatively, it can be counted forward from the last captures of pregnant females (provided the average time to weaning is known). The last captures of newly weaned young could also provide the same information.

Percentage of adult females in breeding condition

Not all adult females will necessarily breed continuously through any particular season. The intensity of effective breeding activity can be estimated in two ways:

- calculating the proportion of adult females that are pregnant during a given trapping period (by palpation or from presence of embryos in the uterus by necropsy)
- calculating the proportion of adult females that are lactating during a given trapping period.

During the breeding period, the proportions of females that are pregnant versus lactating will probably shift, but the changes should be complementary. A useful index of overall breeding activity is the proportion of adult females that are pregnant and/or lactating.

Because the earliest stages of pregnancy are difficult to detect, either by palpation or by necropsy, the pregnancy rate is always underestimated by a significant factor. With necropsy data, pregnancy

may go undetected for the first 5–6 days, representing 25% or so of the total gestation period. For estimates based on palpation, the proportion of undetected pregnancies may be much higher. However, in both cases, the exact proportion will depend on the age distribution of the pregnancies, which can be expected to shift through the breeding season. Thus, during the early part of a breeding season, a very high proportion of pregnancies may go undetected. Later, as the number of new pregnancies falls away, a much higher proportion of pregnancies would be detected by either method.

As noted before, any females that live beyond 12–18 months of age probably stop ovulating. Under natural conditions, very few individuals are likely to survive this long, hence the inclusion of post-oestrus females is unlikely to cause any significant bias in estimates of pregnancy rate.

Percentage of adult females that produce multiple litters within one season

Females that have produced multiple litters within one season will be simultaneously pregnant and lactating. You would also expect to see recent scars on the uterus, especially if conception has occurred immediately after birth. However, these scars may be difficult to see if the second pregnancy is in the second or third trimester and the uterine wall is very

stretched. Removing the embryos from the uterine horns will generally make it easier to count the previous scars.

Average litter size

The average number of young per delivery is an important determinant of the potential rate of population increase. This is usually estimated from the number of embryos present in pregnant females or from counts of recent scars in recently post-natal females. Because some mortality occurs at all stages of pregnancy including birth, these counts are likely to slightly overestimate actual litter sizes. Captive breeding of rodents allows a greater degree of control over litter sizes. However, captive-born litters may be either be smaller or larger than those produced under wild conditions, depending on how well the particular species responds to the artificial diet and living conditions.

Within any one species of rodent, litter size is usually positively correlated to body weight (i.e. larger individuals have more young). In addition, there is often a difference between the number of pups in the first and subsequent litters—this may be partly due to continued growth of the female. Litter size usually peaks around the third or fourth litter, and then falls after that.

Pre-weaning mortality rate

Although litter size is very high in many species of rodents, there is sometimes also a high rate of pre-weaning mortality. This occurs as a result of starvation, predation by animals such as snakes and carnivorous invertebrates, and infanticide both by the mother and by other members of the same species. Because these events generally take place below ground, they are very difficult to observe or even estimate. Perhaps the best way of estimating these parameters is to excavate a sample of burrows at various times through the breeding season. The number and size of surviving pups can then be compared with the number of recent scars as a measure of original litter size.

Recording reproductive data

In Appendix 2, we have provided an example data sheet for recording breeding information. We recommend that you record the capture or sampling day as a **Julian date**, by which is meant the number of the day from day 1 through to day 365 (366 in a leap year). Dates entered in this way are easier to manipulate in computer applications and mathematical models than dates entered in the traditional calendar format (e.g. 2–11–2001). Tables for calculating the Julian date for both normal and leap years are given in Appendix 3.

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CHAPTER 7

Studies of movement

Introduction

Movements of rodents and other animals are studied for many different reasons. One common reason is to understand the way in which individual animals use their local environment. Where are nests or burrows situated in relation to feeding areas? How far does an animal move in one night or over longer periods such as a week or month? Do males and females have different patterns of movement? What pattern of movement do juveniles follow when they become independent? How do movements of one animal affect the movements of others in the same area? Information of this kind is essential to building a complete picture of any species' biology, and is also valuable when looking for ways in which a pest species might be controlled through habitat manipulation or specific management actions.

Another reason to study movements is to understand the contribution of immigration and emigration to changes in local population density or community composition. Indeed, without knowing something about the seasonal and longer-term pattern of movements, it is often difficult to know whether local changes in population density are due to increased breeding or survival or to changes in the pattern of habitat use by members of a more stable population.

These examples emphasise the fact that studies of movement can be used both to frame and to test hypotheses. Gathering information on how members of a particular species move around in their environment is one part of putting together a basic biological picture for the species. When combined with information on population densities and breeding activity, this knowledge can be used to develop specific hypotheses about how the species

functions in time and space. Testing these hypotheses often requires additional studies of movement, but this time the observations must be made within the context of a carefully designed and replicated study that will provide data of appropriate quality and quantity.

Some basic concepts

Animals move around in the environment for many different reasons and at differing levels of regularity. Daily patterns of movement are generally motivated by the need to locate food and water, to avoid predators and to maintain social interactions. Less regular movements might be undertaken to protect resources or to find a mate to reproduce. In some species, occasional, larger-scale movements interrupt the regular pattern. These occasional

movements often result in the construction of a new nest or burrow and the establishment of new feeding areas. Such events might be triggered by the depletion of local food supplies, by some disturbance in the previous location, by social conflict following the arrival of competitors in the area, or by an environmental change (e.g. rising watertable) that makes a previous locality unsuitable for continued use.

The area used by an individual animal in the course of its regular pattern of activities is sometimes referred to as its **territory**. However, this term has connotations relating to the defence of resources and we prefer instead to use the neutral term **home range** to refer to the area used on a day-to-day basis. A home range might be a territory if it is defended. A territory is always, at the same time, a home range. Other useful concepts are **range span**—the largest distance across a home range; and **range overlap**—the proportion of the home range that is used by more than one animal of the same species.

Movements between habitats are sometimes stimulated by changes in the availability of food resources or shelter. Such changes are particularly dramatic in agricultural landscapes, where the harvest of mature crops or tillage of fallow fields can represent a crisis for the rodent community. However, patterns of movement may also reflect differences in the rate of reproduction and population growth between habitats. Ecologists sometimes distinguish between source habitats and sink habitats. A **source** habitat is one where breeding takes place at sufficiently high rate to sustain the population, whilst also supporting a net emigration of animals away from the habitat. A **sink** habitat is one where little or no breeding takes place, and where the population is replenished primarily through immigration. Source habitats thus supply sink habitats with animals.

Techniques for studying movement

A variety of field methods are available to study patterns of movement. Most of these methods are time-consuming and some require the use of expensive equipment. As with any other component of an ecological study, movement studies should be guided by one or more specific questions or hypotheses. These will help you to identify the most appropriate methods and to design a study with adequate sample sizes and, if necessary, with appropriate replication (see Chapter 2).

Capture–mark–release trapping

Capture–mark–release studies, as described in Chapter 5, often provide some information about the local movements of rodents. However, unless large numbers of traps are set across sufficiently large areas, the likelihood of obtaining any significant information about movements within and between habitats is slight.

The technique of marking groups of individuals with a common ear-punch is worth considering if you suspect that there are periods of mass movement of animals between habitats. However, this will only be practical if you are able to capture a sufficiently high proportion of the total population in each of the habitats.

Spool-and-line methods

Spool-and-line methods have been used since the 1920s to study movements of mammals. The method involves attaching a spool of fine thread to a captured animal. The loose end of the thread is attached to a fixed object at the point of release such that the thread spools out or unwinds as the animal moves away. Commercially available spools are enclosed in shrink-wrap, leaving an open end where the thread comes out. For rodents, the spool is fixed to the back of the animal with a non-toxic, fast-drying glue (Figure 7.1).

Where the animal is trapped and released close to its burrow or nest, it will often not emerge again until the following night. Provided that the animal does not dislodge the spool in the meantime, the thread will then track its movements through one or more subsequent activity periods. The number of periods

Figure 7.1   Gluing a tracking spool to the back of a rat. In this case, the spool is glued to the fur. For longer-term tracking, over several days, it would be advisable to glue the spool to the skin after first shaving a patch of fur.

represented will depend on the distance travelled by the animal relative to the length of thread in the spool. In general, the weight of the spool should not exceed 5–10% of the body weight of the animal. For a rat-sized rodent weighing 100–150 g, this usually means a thread length around 100–300 m long. An exhausted spool will usually fall off after a few days. If the spool is attached during the animal's active period, the first 20–30 m of line should be disregarded since the released animal may not show natural behaviour during the 'escape'.

The spool-and-line method can be used to answer many basic ecological questions. It can be used to locate the nests or burrow sites of a cryptic species, to confirm that a particular species is responsible for damage observed within an area of crops, or to determine the general mode of habitat use (e.g. use of trees). It is also sometimes used to quantify the pattern of habitat use, based on the proportion of the line that passes through different habitats. However, this type of information is not always easy to interpret because the distance travelled through each habitat may not simply equate to time spent in the habitats or reflect their relative importance to the animal.

Spool-and-line tracking of a large sample of animals within a population will allow you to calculate values that we refer to as **average nightly range** and **average nightly range span**. Unless you are working with a highly sedentary species, these values will almost certainly be smaller than average home range and range span values estimated for the same population.

Spool-and-line methods are simple to use and relatively cheap. The main limitation of the technique is that each animal is usually tracked for only one or a few nights. Repeated capture and spooling of the same individual is not recommended, as this is likely to impact on its behaviour. The method is most appropriate in areas with moderately dense ground cover, providing numerous points for attachment of the thread and minimal chance of disturbance by large animals. Under open conditions, there is much greater potential for disturbance of the thread by wind and livestock.

Radio-tracking

The development of small radio-transmitters caused a revolution in the study of animal movements. Other methods are either effective only for very short periods, as in the spool-and-line method, or they are effective only if a marked animal returns to a certain location, as in capture–mark–release trapping and the use of passive integrated transponder (PIT) tags (described below). In contrast, a radio-collared animal can be followed to its exact location, provided that it stays within the range of a receiver. This is an invaluable advantage, especially for the study of highly mobile species. However, radio-tracking equipment is expensive (each collar costs >US$50) and the radio-tracking process is labour-intensive and sometimes very difficult in rugged or densely vegetated habitat.

Radio-tracking is the most versatile of the methods described here. It can be used at a very simple, descriptive level to locate nest of burrows of highly secretive species or to follow and observe highly mobile species that might otherwise be very difficult to locate. More intensive tracking of individuals can provide information on home-range size, on patterns of habitat use (including the timing of activity) and on social behaviour (contact with other members of the same species). Finally, if tracking is continued for sufficiently long periods, you might also obtain useful information about patterns of dispersal and survival.

Radio-tracking methods also can be used in the context of more structured experiments. This will often involve selecting contrasting pairs of sites that differ according to some key attribute. For example, to test the hypothesis that rodents will travel further from a refuge habitat to attack crops at the ripening stage than at maximum tillering, you would need to investigate rodent movements at sites that differ only in the crop stages. The contrasting pair would need to be replicated, giving a minimum of four sites in total. Another kind of study might involve tracking different sub-populations within a single locality. This approach could be used to test the hypothesis that male rodents have larger home ranges than female rodents within a common habitat. Again, for a real test of either hypothesis, replication is needed, with tracking of both males and females in at least two different sites.

Practical considerations sometimes limit the number of sites and/or animals that can be tracked within a single time period. For this reason, the design of many radio-tracking activities is a compromise between methodological and practical issues.

Equipment

Transmitters

Transmitters emit a radio signal which is detected using an antenna and receiver (see below). The usual signal band is 150–151 MHz but this may vary from country to country. For use on rodents, radio-transmitters have an external antenna and are fitted to plastic collars (Figure 7.2).

Transmitters differ in size and weight, mainly determined by the size and durability of the battery and whether or not an amplification system (second stage) is incorporated. Larger transmitters, suitable for use on a rat-sized animal, should last for 2–3 months and emit a strong signal that can be located many hundreds of metres away. Small transmitters, suitable for mouse-sized animals, will last for only 2–4 weeks and emit a weaker signal that may not carry much beyond 150 m. Some commercially available transmitters can be turned off using a small magnetic switch.

Figure 7.2   A radio-transmitter fitted to a plastic collar. This transmitter and collar set is suitable for attachment to a rat-sized rodent such as *Rattus argentiventer* or *Bandicota bengalensis*.

It is a good idea to attach a small piece of highly reflective tape to the base of the antenna on each transmitter. This is more easily detected by torch light than the animal's eye shine, and will help to minimise any disturbance of the animal. Even a brief glimpse of the reflective tape also will remove any doubt that an animal seen scampering away is the one wearing the radio-collar.

Antenna and receiver

The most commonly used antenna is a three-element, folding 'Yagi' (Figure 7.3). However, you can also make a simple but effective antenna from about 3–5 m of coaxial cable fixed to a wooden or plastic pole. The antenna is connected to a receiver unit that can be tuned to the individual signals emitted

Figure 7.3   Radio-tracking with a three-element, folding 'Yagi' antenna.

by each of the transmitters in use. Receivers are expensive pieces of equipment and great care should be taken to keep them clean and dry. Ideally, you should have at least one backup receiver with you in case of equipment failure.

Field procedure

A radio-tracking study involves the following key steps:

- selection of sites
- capture, collaring and release of animals
- tracking and marking of radio-locations
- mapping of habitat and radio-locations
- knowing when to stop
- recovery of radio-collars
- data analysis.

Ideally, all animals should be collared and tracked simultaneously across all sites, so that weather conditions etc. are standardised within the samples. However, in many cases, it may not be possible logistically to radio-track at all sites simultaneously, as this would require multiple sets of tracking equipment and a large number of people. Additionally, in many cases, the animals are captured, collared and released over a number of successive days and this means that the radio-tracking effort, even at one site, is often staggered in time.

Where simultaneous radio-tracking is not practical, you should make sure that your sites or sub-populations are interspersed in time. For example, if you need to radio-track at two treatment and two control sites, these should be alternated (i.e. treatment 1 ➔ control 1 ➔ treatment 2 ➔ control 2). This will allow you to analyse the data in two ways: by site type (treatment versus control) and by tracking period (either early versus late, or using sampling order in a rank correlation analysis).

Selecting study sites

Apart from the general issues relating to experimental design, there are some important practical considerations when selecting a site for radio-tracking:

- avoid sites with overhead power lines, which can interfere with the signal
- think about general site access (including wet-weather access) and site security (possible theft of traps, posts and harassment of field workers)
- if possible, avoid working close to houses or other buildings—radio-tracking will be done late at night as well as during the day and disturbance of nearby residents should be minimised
- be aware of the location of large channels, creeks or rivers that may need to be crossed during the night
- if possible, select sites with elevated channel banks, dunes or other high points, which will improve detection of signal (if these are not present and the site is completely flat, you may need to consider using ladders to help to locate any animals that have moved away)
- if possible, select sites where there is some prior information about the rodent population.

Catching animals and fixing radio-collars

In all experimental studies, we make the assumption that the procedure does not significantly alter the natural behaviour of the animal. In radio-tracking studies, it is important that the initial capture and handling of the animal does not cause excessive stress or disruption to its usual activity pattern. For this reason, we strongly discourage the use of any capture method that involves major disturbance of nest sites, such as excavation of burrows. We also recommend that all animals are collared and released as soon as possible after the time of capture.

With these limitations, most radio-tracking studies will probably need to begin with a period of intensive trapping, either using single-capture traps or linear trap–barrier systems (see Chapter 3). However, in some cases, it may be possible to capture animals by driving them into nets or by flushing them from daytime retreats in wood- or straw-piles or the thatched roofs of houses. When using the flushing method, it is important to erect a plastic fence or net around the habitat to minimise the chance of escape or injury to the animals.

As mentioned above, the transmitter should not exceed 5–10% of the animal's body weight. Where the study involves two or more species of different adult size, or adults and juveniles of the one species, it may be necessary to have at least two different-size transmitters at hand. Before fitting the collar, record basic information about the captured animal—the species, sex, age—and take some basic measurements (at least the animal's weight). Each collar will have a unique frequency and associated channel number. This number can be used to identify all of the data associated with that particular rat (e.g. rat no. 24: *Rattus rattus*, male, weight 48 g; capture location, time and date).

The job of fitting a radio-collar is best done by two people—one to hold the animal steady while the other fits and adjusts the collar (Figure 7.4). For a rat-sized animal, this is best done with the animal partially enclosed within a cloth bag. First, adjust the collar's tie until it will slide easily over the animal's head. Gradually tighten the collar until it will no longer slide back over the ears, but not so much that it will restrict breathing. It should be possible to rotate a correctly fitted collar around the animal's neck, but without leaving any space for the animal to insert a fore-limb between its neck and the collar.

Once the collar is fitted, place the animal back inside a bag, bucket or trap and observe its behaviour over a period of a few minutes. If the animal is moving freely and the collar appears to be firm, restrain the animal again and cut away the excess cable tie. If it is too tight and the animal is having difficulty breathing, cut the collar off and try again after the animal has had a rest. It is important to collar an animal quickly and efficiently so that the animal does not become too stressed, as this may affect their movements once released. The animal should then be released close to the point of capture. The release point should be marked with a piece of flagging tape labelled with the rat number and the date.

Radio-tracking and marking radio-locations

Although radio-tracking can be done by one person alone, for safety reasons we recommend that each team consists of two people. This is particularly important for night work or tracking in rugged terrain.

Begin radio-tracking a day or two after initial capture and release. This should give the animal time to get over any capture stress and to become used to carrying the radio-collar. Initially, tracking will be slow as you become familiar with the local terrain and the usual location of each animal. Most rodent species are nocturnal and will be spending the daytime inside one or more burrows or nests. It is probably best to begin a radio-tracking session with what is called a **daytime fix**. Using the original capture location as a guide or starting point, tune the receiver to the specific frequency or channel of the particular radio-collar. Holding the antenna vertically, and with the gain (volume) up full, perform a slow sweep of the surrounding area. Use the fine-tuning on the receiver to obtain the best sound—a clear pulse, sounding like 'choc'. You will probably
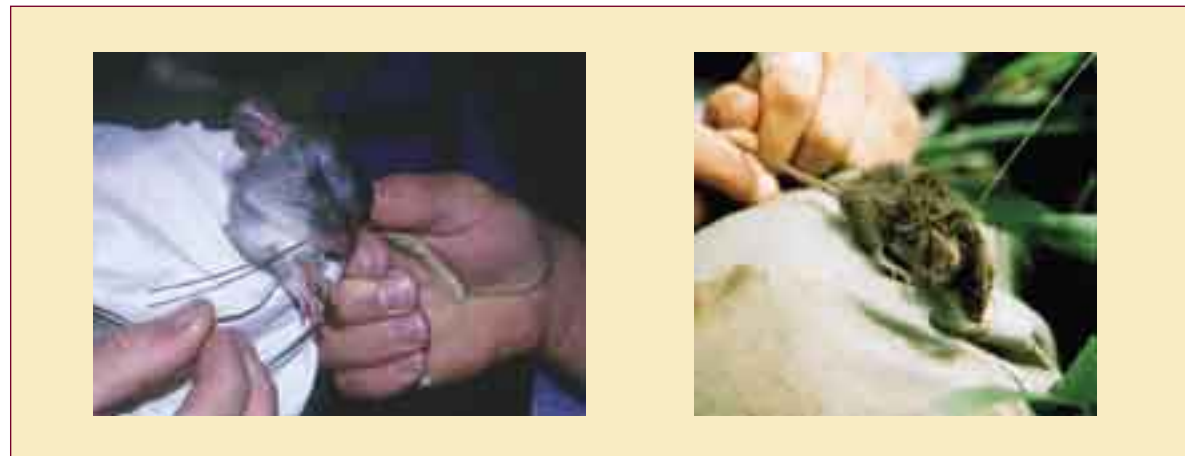
Figure 7.4   Fitting a radio-collar to a *Berylmys berdmorei* (left); and a *Mus musculus* with radio-collar attached (right).

hear a range of high to low pitched pulses, but the middle frequency is usually the clearest. Point the antenna to where the pulse is strongest, then turn the volume down until the signal loses the low and high pitch pulses. Repeat the sweep and the adjustments of tuning and volume until you are confident about the direction of the signal. If there is no signal, you may have to search around or move on to the next animal and try again later.

Once you have identified a general direction, make a mental note of the bearing and then move off at an angle of approximately 30° from that bearing. Listen for the signal at regular intervals, always performing a general sweep to make sure that you have the direction correctly fixed. If you get contradictory signals, begin the whole process again (signals are sometimes bounced around and your original fix may have been an echo).

In areas with dense ground cover of crops or weeds, it is unlikely that you will actually see the collared animal, at least not without causing an unacceptable level of disturbance. In such situations, most radio-locations will be obtained through the general method known as **triangulation**. This is illustrated in Figure 7.5, using the example of a rat that is sheltering in the centre of a rice paddy.

Unless a collared animal has been seen, the only way to be absolutely certain of its location is to perform a complete circle around the source of the signal.

During the daytime, there is little risk of disturbing the animal, so you can afford to make increasingly smaller circles until you have found the exact position. When tracking in areas with tree cover or buildings, be aware of the possibility that the signal may be emanating from a nest located above your head. This may result in confusing signals unless the antenna is pointed directly at the nest site.
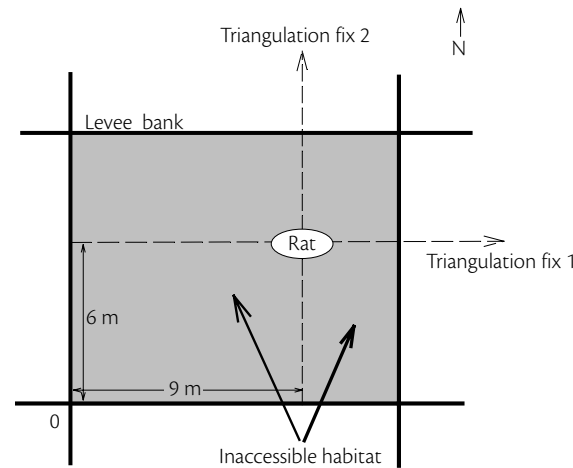
Figure 7.5 Fixing the approximate location of an animal through the process of triangulation. From a starting point (0; bottom left hand corner), pace or measure the distance in the north–south direction to where the signal is strongest and perpendicular to the levee bank (6 m). Do the same in the east–west direction (9 m). The point of intersection of the two fixes is the approximate location of the animal. Where levee banks do not run north–south or east–west, you may need to use a compass to determine the correct angle of each triangulation measurement.

The radio-location should be examined closely, but in a way that will not flush out a resting animal. In many cases, you will probably find an active burrow entrance or a nest. Occasionally, this first fix will lead you to a loose radio-collar that an animal has managed to dislodge. Other possibilities, such as tracking the collar to a large snake (with rat and collar inside), should also be considered.

Each radio-location can either be recorded directly according to a coordinate system (see below) or it can be marked with flagging tape for later recording. If the latter option is chosen, the tape must be clearly labelled with the animal's number and the date and time of the fix. We strongly recommend that you also make some general notes about the location. This will help you to relocate the tape and will also be valuable if the tape is lost or disturbed. Make sure that the tape is clearly visible from all angles.

For **night fixes**, it is important that your own movements do not disturb or influence the animal's behaviour or movement pattern. Hence, it is even more important that you use the triangulation method for all tracking. Do not be tempted simply to move in the direction of the initial fix, as you may find that you are actually driving the animal ahead of you.

Because most rodents are nocturnal in their feeding and general movement patterns, we usually try to obtain a number of fixes spread through the night.

Keep in mind that tracking at night is usually much slower than during the day. This is partly due to the greater difficulties of moving around in the dark, but also because the animals may have moved tens or even hundreds of metres away from the location of the initial daytime fix.

Many species are more active during the early part of the night and then again in the hours before dawn, but this may not be true of all species or even all individuals within one species. One approach is to randomise the time at which fixes are taken through the night for each animal. However, there are also practical limitations to consider (such as the need to sleep!). Another important consideration is to leave sufficient time between fixes for the same animal. Generally, in small rodents there should be about two hours between taking successive fixes.

The process for recording night fixes is the same as that described above for the initial daytime fix. However, we strongly recommend the use of labelled flagging tape rather than on-the-spot calculations of positions, mainly because it is much more difficult at night to maintain correct orientation within the landscape. When marking a fix, it is usually best not to risk disturbing the animal by approaching the exact radio-location, but instead to attach the flagging tape where you completed the fix and mark the tape with a direction (use a compass, if possible) and approximate distance (e.g. rat no. 58; 25 m at 210°N of here; 2130 h; 31/7/2003). Additional notes should be taken on each fix, including whether or not the animal was seen or heard, and if so, what it was doing (e.g. climbing in low shrub, running along low bund).

Mapping habitat and radio-locations

Drawing a good map of the study site is an important part of any radio-tracking study. The map should be drawn at a scale that is appropriate to the questions being asked and to the expected scale of movements of the study animals (usually 1 mm = 1 m). The map should also include a link to the coordinate system that you are using to record radio-locations.

A typical map of an agricultural landscape will contain:
- major channels, secondary channels, main levees and fence lines
- boundaries of the major habitat types (e.g. rice paddy, barley crop, vegetable crop, sugarcane, forest remnants, fallow)
- buildings, houses or edge of village
- location of marker posts or other reference points used for recording radio-locations
- other significant features (e.g. haystacks, trees used by rodents).

For each major crop type, you should make detailed notes of the growth stage (e.g. for rice: transplanting, milky stage, ripening, harvesting, stubble).

In a relatively flat, open landscape, you should begin by staking out a grid with regularly spaced wooden or bamboo posts (20 m spacing for rats, 10 m spacing for mice) using a compass to orient the lines. Initially, the grid should be centred on the area where the majority of mice or rats have been trapped. However, it can be progressively extended to include all of the areas used by the radio-collared animals. The grid will serve a dual purpose. It can be used to draw an accurate map of the site and it can also provide a set of reference points for recording radio-locations. For simplicity of future analysis, it is a good idea to orientate the grid to run north–south and east–west. To map a site using this method, you will need tape measures, marker posts (e.g. garden stakes, bamboo posts), flagging tape and marking pens, a compass, ruler and large sheets of graph paper.

In more complex habitat or situations where the radio-collared animals are dispersed over much larger areas, it is often not practical to use the grid method for mapping. In such cases, you should start by producing a larger-scale schematic map that shows the distribution of major habitats and landscape features. This can be measured out with a long tape and compass, or by pacing along compass bearings. In areas where rodent activity is concentrated, you can then produce more detailed maps, either by establishing a local grid or by a tape and compass survey. You should link these detailed maps back to specific features on the large-scale schematic map so that a composite diagram can be produced. If

possible, fix the coordinates of the large-scale map using a global positioning system (GPS).

You should always try to complete drafts of all maps in the field to make sure that all necessary detail has been recorded before you leave the field site.

Knowing when to stop radio-tracking

How do you know when you have enough data on each animal? The answer to this question depends to a large extent on your initial questions. If your goal is simply to learn as much as possible about the natural history of the animals you are following, then the answer is that you should keep tracking until you fail to make any new observations or until the equipment or field resources give out. However, most radio-tracking studies have more specific goals. For example, you may be interested in how rodents respond to an environmental change, such as a cropping cycle or flooding event. In such cases, the duration of a study may be determined by the environmental schedule. Alternatively, you may be interested in estimating certain parameters of spatial behaviour within a static environment, such as home range, range span and range overlap, as introduced earlier.

The statistical methods used to estimate parameters of spatial use allow you to calculate an appropriate number of fixes. As with most statistical methods, progressively larger samples result in smaller proportional error values and tighter confidence intervals. In general, home-range estimates based on fewer than 15 fixes often have proportionally large errors. Increasing the sample size to 30 fixes will substantially reduce the error; however, going from 30 to 50 or even 100 fixes does not really improve the degree of certainty much for all the extra effort. As a general rule of thumb, 20–30 fixes per individual will give a good estimation of home range (and of range overlap when multiple individuals are tracked at one site). Some people prefer to include only night fixes (i.e. those taken during periods of activity) in this total. One good reason for this is that the daytime fixes are often repeats of the same location, i.e. a nest or burrow site, and this violates the statistical assumption that the fixes are independent representations of the home range. Night fixes that are taken too close together in time are also of suspect value for the same reason. Range-span values are less closely related to sample size due to the fact that the value is sensitive to a single, large excursion by the animal in any direction.

Where a particular radio-collared animal has not moved over a period of two or more days, you should consider the possibility that it has either died at that location or that the collar has been dislodged. In either case, it is probably best to investigate the radio-location carefully and retrieve the carcass and/or the collar. If this occurs early during a study period, it may be necessary to fix the collar to a new animal and recommence tracking.

Recovering radio-collars

Radio-collars should be recovered at the end of the radio-tracking study. Hence, you should not allow the battery to run down completely, otherwise you may not be able to find it. Radio-transmitters are expensive and it is also considered unethical to leave animals collared for longer periods than necessary. For most transmitters, the battery can be replaced or recharged to restore them to full function.

Collared animals can be recaptured in traps set close to their nest or burrow, or they can be flushed directly from their daytime retreat, using a plastic fence or netting to encircle the animal. Where an animal is tracked back to a burrow system, this can be fumigated or excavated to retrieve the animal and the collar. Close examination of nests and burrows will also tell you whether the animal was living singly or communally and whether a radio-tracked female was rearing pups. This information may allow you to interpret otherwise unexplained variation in the pattern of movements between individuals or groups of animals (e.g. between pregnant or nursing versus non-breeding females). If recaptured animals are sacrificed, even more information can be obtained by examining their reproductive condition and history and even their disease status (e.g. parasite load may influence behaviour).

Data analysis

Several computer packages are available for analyses of radio-tracking data. One of the more widely used programs is Ranges V, which offers a large range of methods to analyse spatial and temporal patterns of habitat use. The more user-friendly Ranges 6 has just become available on the Internet. In both versions, radio-location data can be imported from spreadsheet or database computer files (e.g. Excel, Access) and the results and graphs can be exported to other applications. Less comprehensive packages for the analyses of radio-telemetry data can be obtained for free from various websites (see Further reading).

Bait markers

Bait markers (or 'biomarkers') work on the general principle that a food item containing an identifiable marker is provided at a known location and point in time. Some time later, animals are caught and analysed individually for the presence of the marker. Depending on the type of bait marker used, evidence of food uptake can be found in faeces or 'scats' (e.g. wool threads, plastic beads), in external tissues such as claws and hair (e.g. rhodamine B, DuPont oil blue A) or in internal tissues such as blood, bones and teeth, and the intestinal tract (e.g. radioactive markers, rhodamine B, tetracycline).

Bait markers are often used to study feeding behaviour. For example, a bait marker can be used to find out which species consume a particular food item, or which species eat from a particular location, such as a grain store. Bait markers can also be used to study movement patterns, typically by posing the question: Where do the animals come from that eat the bait? Finally, bait markers can be used to study aspects of social behaviour, such as intraspecific competition for food or access to particular habitats.

The preparation and application of bait containing a biomarker is usually inexpensive and does not require much labour, even when it is used on a large scale. However, the analysis of samples may require special and expensive equipment and it is usually time-consuming. We will illustrate this class of methods with information on one particular biomarker, rhodamine B.

Rhodamine B

The non-toxic xanthene dye rhodamine B (RB) has been used as a bait marker in several studies involving small mammals. The substance is palatable to rodents and it can be detected under ultraviolet (UV) light in many tissues, including whiskers and blood. In house mice, uptake in bait of 3 mg RB results in the detectable presence after 12 hours of RB in both internal and external tissues (intestines, blood, whiskers) and in excretions (urine, faeces). It remains visible under normal light for up to four days in urine and the digestive tract, and in faeces for up to two days. RB is detectable in blood serum for up to 84 hours using a fluorometer and in whiskers for up to 7 weeks after ingestion. Sampling whiskers or blood has the added advantage that the same individual can be sampled repeatedly. RB is detectable for similar periods in rats and other small mammals.

Bait preparation and delivery

RB can be mixed with grain kernels to produce a 'home-made' bait, or it can be offered as commercially produced, dry extruded pellets (Figure 7.6). Whatever bait is used, care must be taken to ensure that the mixing process gives an even concentration of RB throughout the bait. A concentration of 0.5% RB is palatable to rodents.

Figure 7.6 Rodent bait pellets containing 0.5% rhodamine B under ambient light (left) and under ultraviolet light (right).

RB particles will stick to skin, laboratory benches and equipment, staining everything that comes in contact with it. It is important to have designated RB mixing areas and equipment to avoid contamination of other equipment and materials. Rubber gloves, a lab coat and a face mask should be worn when mixing bait.

Bait can be broadcast or distributed in bait stations. The advantage of using bait stations is that the bait can be provided for a known time period only and at specific locations.

Sampling and detection

Depending on the research questions, animals may be sampled in the general vicinity of a bait station (e.g. for a comparison of bait uptake between sexes or between young and old individuals) or at various distances away from the bait station (e.g. to estimate the foraging range of animals).

For tissues other than vibrissae (whiskers), sampling for RB will need to be carried out within a few days of bait provision. If vibrissae are used, several weeks can elapse between bait distribution and sampling.

Vibrissae —for a particular sampling episode, pluck with tweezers at least two vibrissae (one from either side of the nose) from a restrained live animal. Examine them for the presence of RB-coloured bands under a UV spotlight. Alternatively, examine using a fluorescence microscope at 10× magnification. An animal is scored as RB-positive if at least one vibrissa shows orange fluorescence in the hair bulb or a band of orange fluorescence partway along the shaft (Figure 7.7).

Blood—draw from the suborbital sinus or caudal vein if the animal will be released, or from cardiac puncture if it will be euthanased. Centrifuge a 100 μL sample at 10,000 rpm for 3 min. Remove the serum and freeze at −20°C until it can be analysed with a fluorometer. After thawing, dilute two 20 μL subsamples of blood serum each with 80 μL double-distilled water. Scan the subsamples with a fluorometer and record the photons generated by RB fluorescence as counts per 0.2 s. An animal is considered RB-positive if the fluorometer reading is higher than the average value +3 standard errors of the reading obtained from a series of control samples from mice that have not eaten any RB-bait.

Other tissues—to screen intestine or other tissues for the presence of RB, first necropsy the animal. Freeze the tissues at −18°C until analysis. Inspect the samples for pink colouration under normal light or under a UV spotlight. Comparison with RB-free control animals is necessary to guarantee accurate results.

Figure 7.7   Rat vibrissa (whisker) examined under a fluorescence microscope (ultraviolet light) showing a fluorescent rhodamine B band.

PIT tags

Passive integrated transponder (PIT) tags can be used to monitor small-scale spatial and temporal activity of rodents. A PIT tag is a microchip encapsulated in a glass tube (5 mm long) (Figure 7.8). The tube is implanted under the skin of an animal. These tags are routinely used by veterinarians to individually identify domestic animals.

Studies using PIT tags are usually aimed at small-scale movement patterns (e.g. time of movement in and out of burrows), foraging behaviour (Which animals visit particular feeding places?) or social behaviour (e.g. Which animals share the same burrow?).

PIT tags have no internal power supply but they become energised when they come in close proximity to an electromagnetic field generated by the antenna or a reading device. The reading device retrieves the identification number stored in the chip and records this information along with the date and time that the reading occurred. This information can be downloaded from the reading device and provides a detailed record of which animals have passed by the antenna and at exactly what times.

The advantage of PIT tags is that the activity of free-ranging animals can be observed without external attachments to the animal (spool, radio-transmitter). Disadvantages include the short detection range of

the readers (approximately 50 mm, depending on tag orientation to the antenna) and the high cost of the PIT reader system (US$3 per PIT tag, US$500 for a hand-held reader, >US$3000 for automated reading systems).

The basic equipment for PIT tag studies is the PIT tags, a device to inject a PIT tag under the skin of an animal, a reading device and a computer. Reading devices may be hand-held, where each animal is scanned manually, or automated, with the antenna connected to a data logging system.

Figure 7.8 A passive integrated transponder (PIT) injector (A) with needle (B) and PIT tag (C), and a hand-held reading device (D).

Further reading

Spool-and-line methods

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CHAPTER 8

Techniques for disease studies

Introduction

Diseases probably play an important role in regulating natural populations of many vertebrate species. Human biology provides some of the best examples of how diseases can limit the ability of a species to occupy what, in all other respects, is a suitable environment. For example, before effective medical treatment was developed against trypanosomiasis (sleeping sickness), large areas of West Africa were largely unpopulated due to the high prevalence of this debilitating and fatal disease.

Knowledge about wildlife diseases is often most detailed for what are called **zoonotic diseases** or **zoonoses**. These are diseases that can be transmitted between animal hosts and humans. Rodents carry many zoonotic diseases, such as the plague, arenaviruses and hantaviruses, rat typhus, lungworm infection and leptospirosis. Several of these diseases have played a major role in shaping the course of human history, and some of them continue to cause suffering and hardship in many parts of the world. In addition, new rodent-borne zoonoses are identified on a regular basis. For example, between 1995 and 1999, more than 25 new hantaviruses and arenaviruses were identified in rodents.

Despite the obvious clinical and economic importance of rodent-borne zoonoses, their basic biology is, in general, poorly understood. With few exceptions, little is known about which species of rodents are the major reservoir of each disease, how long the infective life stages of each pathogen (e.g. bacteria, viruses, spirochaetes or helminths) persist in domestic and rural environments, how these diseases are transmitted in wild rodent populations and then to humans, how prevalent these diseases are in both the rodent and human populations, and the basic human epidemiology of these diseases (i.e. incidence of infection, morbidity rates, transmission rates, age and sex-related effects, and effects of socioeconomic status).

The impact of rodent diseases on human livelihoods, in both urban and agricultural communities, also is poorly documented. However, the available evidence suggests that the impact on human health is increasing in developing countries. This trend is probably linked to increased:

* movements of people between rural and urban areas
* movement of people between countries
* human population density, which amplifies the ability of a disease to spread through populations
* clearance of natural habitats, which leads to a higher incidence of rodent–human contact.

Despite these trends, little research is being done on the epidemiology of rodent diseases in either Asia or the Pacific region. The situation is similar in Europe and Africa.

In the Asian context, our most detailed knowledge about a rodent-borne disease relates to leptospirosis. This disease is reported from Indonesia, Vietnam, Australia and the Pacific Islands. Although generally not fatal, leptospirosis is nonetheless having a major impact on rural communities in many developing countries. Surveys conducted in north-eastern Thailand showed a marked increase in the number of diagnosed cases of leptospirosis from 1995 to 2000, with a maximum of 14,608 cases and 365 deaths reported in hospitals in the year 2000. Since 2000, the number of reported cases in north-eastern Thailand has declined, despite increased public awareness and improved hospital testing. This hints at some natural cycle, either in the general environmental prevalence of leptospirosis, or in rodent populations specifically.

Most deaths from leptospirosis involve rice farmers who are regularly exposed to infection as they work their fields. The early symptoms are influenza-like and can easily be mistaken for malaria and dengue fever. Often the disease is neglected in the rural areas until serious clinical damage has occurred. This is unfortunate because the disease, if diagnosed early, can be treated effectively using antibiotics. By improving farmers' knowledge and practices for rat management, the prevalence and impact of various zoonoses, particularly leptospirosis, could be greatly reduced.

Many rodent-borne diseases can infect a variety of other hosts, including livestock and companion animals (cats and dogs). In some cases, these diseases also affect the health of livestock, leading to weight loss, reduced fertility or even death. Examples of diseases that can affect both rodents and livestock include leptospirosis (in pigs and cattle), erysipelas and trichinella (in pigs), tapeworm and other helminths (probably in all livestock). For communities who live in close proximity to their livestock, such as many of the Hill Tribe peoples of Southeast Asia, the cycle of transmission between rodents, livestock and people may be even more complex, and the levels of risk perhaps higher again.

The potential use of diseases or parasites as biological control agents for rodent management has been explored in several countries, including Australia and Malaysia. Biological control can act either on the animal's reproductive system (i.e. by reducing fertility) or on the fitness or mortality rate of infected adults. To be acceptable, biological control must be specific to the pest species. Before options for biological control can be explored for any target rodent species, we need to know, at a minimum, which disease agents are present in the natural rodent populations, the prevalence of infection (proportion of animals infected) for each disease, the processes of transmission, and the impact of each disease at the individual and population levels.

This chapter describes the sampling techniques used for population surveys of helminths, viruses and bacteria. It is not a comprehensive guide to disease sampling but should provide a useful introduction to the subject and associated techniques. Anyone wishing to work in this area is encouraged to contact local health agencies to discuss the most pressing health issues and appropriate sampling procedures.

Helminths

The major groups of helminths

The three most common groups of helminths are nematodes, cestodes and trematodes. Those recorded in Southeast Asia and the Pacific region are listed in Box 8.1.

Nematodes

Nematodes are also called roundworms. They are non-segmented, with an elongated, round body. The body wall is cuticular and there are no cilia (hairs). Sexes are usually separate and the larvae resemble the adults. They have a simple internal structure, with a distinct mouth, straight intestine terminating in an anus, and a simple nervous system (Figure 8.1).

Nematodes commonly occur in the stomach, small intestine, caecum, large intestine, liver, lungs and body cavity of rodents. They occur less frequently in the heart, kidney, eye, mouth, tongue, oesophagus and muscle tissue.
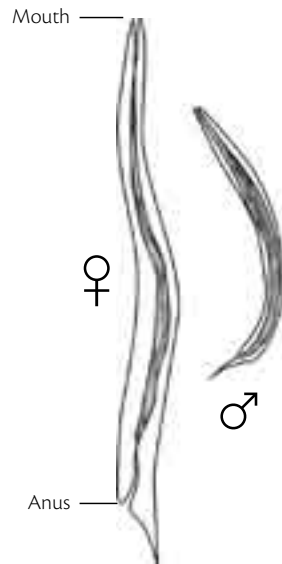
Figure 8.1   A typical nematode body form.

Trematodes

Trematodes are also called flatworms (or flukes) because they have a flattened body. The body wall is thicker than that of a nematode. There are generally two suckers—one located on the ventral surface which is used for attachment, and one surrounding the mouth (Figure 8.2). They also have a simple internal structure, with a distinct pharynx and a blind, forked intestine, but no anus.

Trematodes are most often found in the gut, liver, bile duct, gall bladder, lungs, pancreatic duct, ureter and bladder of the host.
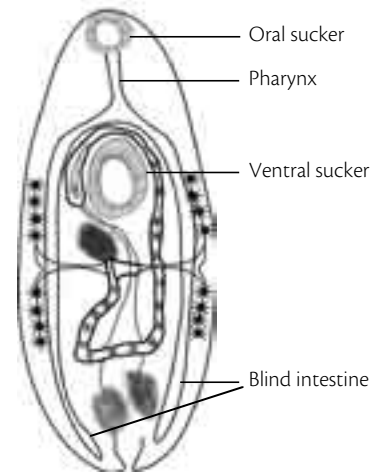
Figure 8.2   A typical trematode body form.

Cestodes

Cestodes are also known as tapeworms. They have segmented bodies and a tough outer surface (Figure 8.3). There are two main external body parts: the scolex, which has hooks and suckers used for attachment (this is the equivalent of a head and is not segmented); and the proglottids or segments, each of which carries one or two reproductive systems. Cestodes lack an alimentary canal.

Adult cestodes are found in the gut and bile ducts that enter the gut. Larval cestodes occur in organs such as the lungs and liver.
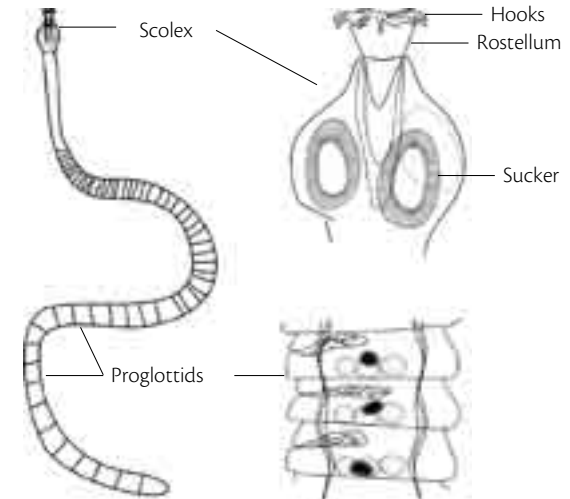
Figure 8.3   A typical cestode body form with details of the scolex and representative proglottids.

Where and how to look for helminths

Laboratory procedures

Rats are easiest to necropsy for parasites when they are freshly dead. If this is not possible, rats can be frozen and the necropsy conducted at a later date, after thawing.

Essential equipment includes good-quality forceps and scissors, glass Petri dishes, a stereomicroscope and light source, gloves and rubbish bags. Glass containers, suitable labels (jewellers' tags are good), pencils and formalin (see Chapter 4) will be needed for labelling and preserving specimens.

Where possible, work on a clean laboratory surface and use clean glassware. Wear disposable gloves and place the used gloves, other used disposable items, and the necropsied rat body in a strong bag at the end of the session. Where possible, incinerate the bag. All used equipment and benches should be thoroughly cleaned with detergent and water after the session and sterilised with 70% ethanol, if available.

Organ examination

For helminth examinations, we recommend the following procedures:

Skin (with fur)**:** after skinning the animal, place the skin in saline solution (0.8% NaCl) and stretch it out. Parasites will be drawn out into the saline.

Tongue: remove from mouth and flatten between two Petri dishes. Examine under a stereomicroscope with not less than 12× magnification.

Oesophagus: as for tongue.

Lung: remove a sample of lung and flatten between two Petri dishes. Examine under a stereomicroscope with not less than 25× magnification.

Liver: examine the surface visually first—some nematodes (e.g. *Calodium*; formerly *Capillaria*) create distinctive white tracks along the surface of the liver. If necessary, cut the sample into smaller pieces and flatten each piece between two Petri dishes and examine under 12× magnification.

Stomach: open the stomach and tease out the stomach contents onto a Petri dish. Examine both the stomach lining and the contents under not less than 12× magnification.

Duodenum and small intestine: extract the tissue from the body and 'unwind'. Spread out in a Petri dish so there is no overlap. Flatten with another Petri dish and examine under not less than 12× magnification.

Caecum and large intestine: as for duodenum and small intestine.

Box 8.1 Previously recorded helminths

Helminth diseases that are a potential risk to humans or livestock in Southeast Asia and the Pacific region are listed below.

***Paragonimus* spp.:** these trematodes are widespread throughout East and Southeast Asia and have a large number of mammalian hosts, including rodents which can act as reservoirs.

***Hymenolepis* spp.:** these cestodes infect humans throughout southern Asia. The exact role of rodents in transmission is unclear.

***Railletina* spp.:** rodents are the primary host for these cestodes. Infection occurs by ingestion of food contaminated with the intermediate host (arthropods including beetles and house flies).

***Schistosoma japonicum*:** infection by this trematode is one of the most serious health problems in the developing world. Humans are the primary host but many wild and domestic animals also act as reservoirs.

***Angiostrongylus cantonensis*:** the adult form of this nematode lives in rodent lung tissue. Infection of humans occurs by ingestion of the intermediate or paratenic hosts—generally a gastropod (snail or slug) or freshwater prawns or terrestrial crabs. The parasite is carried by many rodent species throughout Southeast Asia. Infection in humans is of concern because the larval nematodes migrate to the spinal cord and brain; this condition can be fatal in young children.

Calodium hepaticum (formerly *Capillaria hepatica*)**:** an extremely common nematode infection of rodents. Humans may be infected, but infections are rarely fatal.

***Trichinella spiralis*:** nematodes that infect rodents through the ingestion of infected pig meat. Similarly, the infection is passed on to humans by ingestion of infected meat.

Measures of helminth infection

There are two main measures of helminth infection:

Prevalence of infection: this is simply a measure of the percentage of animals infected with a particular parasite. For example, if 25 rice-field rats (*Rattus argentiventer*) from a sample of 75 rats had their livers infected with the nematode *Molinacuaria indonesiensis*, then the prevalence would be 33%.

Intensity of infection: this requires the number of individual helminths of a particular species to be counted per animal. The mean intensity is a population measure that refers to the mean level of infection per rodent. This mean is calculated from only those animals that are infected. So, working from the previous example, we would conduct counts of the number of parasites in each of the 25 animals infected with *M. indonesiensis* and then estimate the mean level of infection in these 25 animals only, omitting the animals that had no parasites.

Preserving specimens of helminths

Unless you are very familiar with their taxonomy, parasites can be difficult to identify. If you are unsure about a specimen, it may be best to preserve the parasite and seek assistance from a specialist.

Carefully extract the specimen from the organ or body part. Try to keep the body in one piece. If this is not possible, then preserve all the pieces, as they may be needed to determine species and sex. Different parasites must be preserved in different ways:

Nematodes: preserve the specimen in hot 2–5% formalin (approximately 80°C).

Trematodes: preserve the specimen in hot 2–5% formalin. If there are two specimens, preserve one in cold 2–5% formalin and one in hot 2–5% formalin.

Cestodes: remove the tissue sample containing the parasite and place in a Petri dish of water. If the parasite is in the gut, open the gut to let the water bring the parasite out. Do not scrape the parasite out as this might break the parasite body or head and damage or lose hooks that may be present. Once the parasite is free of the tissue and relaxed in the water for 10–15 minutes, preserve in hot 2–5% formalin.

If possible, use small glass containers with screw-on lids that fit securely. For each specimen, record on a small piece of card (with pencil) the species of the host animal, the location and habitat of the host, the date, the collector's name, the tissue from which the sample was collected, and what you know about the parasite. Put this card into the solution with the parasite.

If you send the specimen to an expert in another country, make sure to follow all regulations for the import/export of biological material.

> **WARNING:** do not inhale fumes from hot formalin. This fixative is a strong irritant and the fumes can damage your eyes or respiratory tract, and may cause cancer with prolonged exposure. Formalin should be heated in a well-ventilated area.

Viruses and microbial diseases

Viruses and microbial organisms of various kinds can infect many different types of tissues within the body. Those recorded in Southeast Asia and the Pacific region are listed in Box 8.2. Methods used in the isolation of particular pathogens are often quite specific and require particular culture media and environmental conditions. For this reason, most epidemiological studies begin with a serological survey based on blood samples.

When an animal is invaded by a potential pathogen such as a virus or bacterium, the white blood cells react to proteins on the surface of the pathogen and form antibodies that are specific to its molecular structure. These antibodies are found in the blood serum and specific tests can be performed to identify particular antibodies. Note, however, that most serological tests do not tell us whether the animal is currently infected with a replicating virus or bacterium, only that the animal has been exposed to the pathogenic agent sometime during its life.

We will concentrate here on methods used to collect, preserve and analyse blood samples.

Box 8.2   Previously recorded viral and microbial diseases

This section concentrates on rodent-borne zoonotic diseases — they present a potential risk to humans or livestock in Southeast Asia and the Pacific region.

Hantaan virus (haemorrhagic fever): there is a group of hantaviruses that has been detected in urban populations of rodents in many parts of the world. The virus is passed from host to host via infected saliva, urine and faeces. Some strains have little effect on humans; others cause major illnesses with a wide variety of symptoms.

Tick typhus (*Rickettsia conori*): the principal reservoir for this disease is the dog, but rodents are also important reservoirs. The disease in humans results from a bite from an infected tick. The tick particularly involved in transmission is found throughout Asia.

Scrub typhus (*Orientia tsutsugamushi*): a variety of rodents throughout Asia are the principal reservoir for this disease, which is transmitted by larval trombiculid mites called 'chiggers'. Mortality rates in humans are low if treatment is sought early.

Murine typhus (*Rickettsia typhi*): reported throughout Southeast Asia, this disease is spread by flea bites or contact with infected faeces or crushed fleas. The disease causes a wide range of symptoms in humans, but the mortality rate is low.

Queensland tick typhus or spotted fever (*Rickettsia australis*): occurs down the eastern coast of Australia and is carried by ixodid ticks. Natural reservoirs of the pathogenic organism appear to be marsupial mice, bandicoots, possums, rats and mice. The disease causes a wide range of symptoms in humans, but the mortality rate is low.

Leptospirosis: caused by a variety of spirochaetes, leptospirosis is one of the most prevalent zoonotic diseases carried by rodents in rice fields. Almost all rodent species in Southeast Asia can act as hosts. Human infection occurs when an open wound comes into contact with water, moist soil or vegetation contaminated by rat urine. The mortality rate is low for most strains. The symptoms are similar to influenza and last from several days to three weeks. Symptoms of leptospirosis can be confused with those of malaria and dengue fever, and many cases are probably misdiagnosed. People working in rodent-infested plantations or fields are most at risk.

Rat bite fever (*Spirillum minor*): caused by a spirochaete, this disease is transmitted by rodent bites and is found throughout the world. Incubation takes several weeks and symptoms usually appear after the wound has healed.

Plague (*Yersinia pestis*): a bacterial disease that can be treated with antibiotics if diagnosed early. The cycle of this disease is mammal to flea to mammal, with rodents as the primary host. Whilst advances in medical science make it unlikely that plague will erupt again in global pandemic, as it did on various occasions through history, it still presents a serious health problem in many parts of the world. The last major epidemic of plague in Asia and Australia occurred in the first decade of the 20th century.

Salmonellosis: *Salmonella* bacteria infect humans worldwide, usually through ingestion of water or food contaminated by faeces of an infected animal but also through eating incorrectly prepared foods. There are many strains with variable severity of impact.

Toxoplasmosis: caused by a coccidian *Toxoplasma gondii*, for which the domestic cat is the primary host. Many other mammals, including rats and mice, may act as intermediate hosts.

Collecting and processing blood samples

It is best to take blood samples from freshly caught rodents. Never take blood samples for viral testing from animals that have been housed together for more than three days. Transfer of infection by close contact may lead to a virus being present in all animals to be sampled, giving false prevalence results.

The following procedure can be followed for the collection of sera for viral testing.

+ Anaesthetise animals one at a time using carbon dioxide (as described in Chapter 3), until they are unconscious but not dead. If bottled carbon dioxide is not available, then carbon monoxide can be used via exhaust fumes from a petrol-fuelled car (diesel fumes are not effective). However, we strongly recommend the use of carbon dioxide.
+ Open the chest cavity and draw up to 3 mL of blood directly from the heart using a needle and syringe. A 5 mL syringe and 21 gauge needle is an efficient combination. If possible, angle the needle up into the ventricle of the heart, along the line of the body. Try to avoid air bubbles as these may lead to lysis of the sample (broken blood cells). New equipment must be used for each animal. After collecting the blood, use cervical dislocation to ensure that the animal is dead.

+ Remove the needle from the syringe and transfer the blood to small plastic tubes with lids (2.5 mL Eppendorf tubes are ideal). These tubes should be clearly labelled with a number or code that identifies the individual rodent. Note that rapidly forcing blood through a needle will result in lysis of the sample, hence the importance of removing the needle.
+ Put the sheath back on the used needle, and store the needle and syringe in a solid container. At the end of the sampling session, the container should be incinerated, if possible.
+ Leave samples for approximately 1 hour at room temperature (<30°C) or until a blood clot has formed in the tube.
+ Score the sample by separating the clot from the walls of the tube using a clean needle or pipette for each sample. Alternatively, you can use a probing instrument if it is dipped in alcohol and sterilised with a flame between samples.
+ If you have a centrifuge, leave the samples for 1 hour and then spin them to increase the yield of sera. Make sure that the lids are secured. Ideally, spin the samples for 5 minutes at 2500–3000 rpm. If you do not have a centrifuge, store samples overnight in a refrigerator.
+ Remove the sera (clear liquid) into tubes with a pipette. Use a new pipette for each sample. When possible, separate the serum into at least two tubes—this provides a backup in case something happens to the first sample and will also allow you to do other tests at a later date.

+ Label the tubes clearly with the rodent's identification number, the date, and number of samples. A black permanent marker pen is recommended. Clear labelling of samples is essential. Samples that have illegible or smeared labels are usually worthless.
+ Once all of the sera have been collected, store the tubes immediately in an upright position in a freezer. Ideally, storage should be at or below −50°C. However, sera can be stored for up to a month at −18°C, the temperature of a basic household freezer.

Lyophilising (freeze-drying) samples

Samples that have been prepared by lyopholisation or freeze-drying can be transported to a testing laboratory without having to remain frozen. This is a major advantage if samples have to be transported a long distance, or if the local transport systems are unreliable.

If you have access to a lyophiliser or freeze-dryer, carefully follow the directions of the manufacturer of the equipment. If samples to be lyophilised are frozen, thaw at approximately 4°C, either on ice or in a refrigerator. If the samples are to be lyophilised the next day, thaw them in a refrigerator overnight. It is vital that someone is in attendance while the samples are being lyophilised. If there is a blackout or if the samples are not fully lyophilised by the end of the day, remove them from the lyophiliser, recap

them and refreeze immediately. When you are ready to begin again, thaw the samples and then begin the process again.

When the samples are fully lyophilised, recap them well. If possible, cover the lids with parafilm or thick tape. The lyophilised samples can be stored in a clearly labelled plastic bag and placed within an airtight container until they are ready to be sent for testing.

Sampling design for rodent disease studies

Sampling for rodent diseases is unfortunately often dictated by logistics and money. Samples are taken when and where opportunity permits, and statistical analyses are designed around the available data.

Two strategies for sampling are described in this section: one determines a sample size in advance and the other does not. There are limitations to each, but they can be used as a guide when embarking on sampling for disease.

A general rule, applicable to both sampling strategies, is that you should try to obtain a cross-section of the population, as there may be an age or sex bias in any disease prevalence. It may also be possible to maximise the use of animals by taking blood or

tissue samples from animals that have been killed for some other purpose (e.g. for taxonomic or breeding studies).

Optimal sample size for detecting a disease (predetermined sample size)

To determine the sample size required to investigate whether a population is infected or not with a particular pathogen, the following equation can be used:

$$ n = \frac{\left[1-(1-CL)^{1/d}\right]\left[N-(d-1)\right]}{2} $$

where n = the required sample size
   N = the total population size
   d = the number of diseased animals in the population
   CL = the confidence level as a fraction.

Table 8.1 is given as an example.

As can be seen from the table, for detection of disease at very low prevalence, the sample size needed is very high. At high disease prevalence, the necessary sample size for detection is low, even when the population is large.

Sequential sampling (no fixed sample size)

Even where the optimal sample size can be estimated, it is often not possible to meet the required numbers, either because of inadequate field time (especially with detailed parasitology) or because of budget limitations (especially when testing for more than one virus).

A method that can minimise the sample size (and therefore save time and money) is sequential

Table 8.1   Calculations of the sample sizes (numbers in cells) required to accurately determine (at 90% confidence limits) the prevalence of a disease (%d) within populations of variable size (N).

%d	N								
	10	20	50	100	150	200	500	1000	5000
1%	10	20	50	90	117	136	184	205	224
5%	10	18	30	36	39	40	43	44	45
10%	9	13	18	20	20	21	21	22	22
20%	6	8	9	10	10	10	10	10	10
50%	3	3	3	3	3	3	3	3	3
75%	2	2	2	2	2	2	2	2	2

sampling. This type of sampling does not rely on choosing a sample size in advance. Instead, observations are made one at a time, and after each observation a decision is made either to stop sampling or to continue sampling. An example is given in Figure 8.4 (with 90% confidence limits).

This method is useful for determining the prevalence of a parasite, but could not be used to determine the prevalence of a bacterium or virus unless facilities were available for immediate testing.

Prevalence is defined as the percentage of animals infected by a particular disease agent. If the prevalence is low or high, there is little variance in estimates, therefore sample sizes of around 30 animals are typically adequate. If the prevalence is between 30% and 70%, then sample sizes will generally need to be around 60 to 70 animals.

The sample size required to provide a 90% or 95% confidence interval of the prevalence can be calculated from published tables. These tables and other useful tools for quantitative epidemiological studies are also available on the Internet (see Further reading).

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Figure 8.4 An example of sequential sampling to determine the prevalence of a particular disease (for example, a particular parasite). Animals are examined one by one for the presence of the parasite. Animals continue to be examined until the result is either over the top line or below the bottom line of the graph. If the result falls in the top section, we are 90% confident that the prevalence is greater than 20%. If the result is in the bottom section, we are 90% confident that the prevalence is less than 10%. If the result falls between the two lines, the prevalence is uncertain and another sample is taken. A detailed description of sequential sampling can be found in Krebs (1999) *Ecological Methodology, Chapter 9.*

CHAPTER 9

Assessing crop damage and yield losses

Introduction

The ultimate goal of most rodent management activities is to reduce the impact of rodents on crop production. To measure the impact of our actions, we need simple and effective methods for assessing the level and consequences of damage inflicted by rodents.

Rodents can attack crops at any stage during production and storage. It is convenient to break the resulting impact into two components, namely:

* **preharvest** impact, caused by rodents to growing crops, through to harvest
* **postharvest** impact, caused by rodents during any period of storage.

When talking about rodent impacts on crops, it is essential to distinguish between crop damage and

crop loss. Rodent **crop damage** is the actual physical harm inflicted by rodents on crops or produce. It can occur at any stage during the production and storage of crops, and includes the excavation and consumption of newly sown seed, the cutting and removal of tillers and attached panicles in cereal crops, and the gnawing of tubers or fruits. In stored crops, damage includes both direct consumption and contamination with urine or faeces.

Crop loss caused by rodents is measured at the point of harvest for preharvest impacts, or at the point of consumption or sale for postharvest. These losses are the cumulative result of damage that occurs during crop growth and storage, respectively. The relationship between damage and loss is very complex, especially in the case of preharvest impacts, and it often not possible to directly equate the two figures.

In most rodent management projects, our ultimate goal is to reduce crop losses caused by rodents. The most direct way of measuring the success of any rodent management system is therefore to measure the yield at harvest and at the point of sale or consumption, and to compare these values either with the situation before rodent control measures were adopted or at similar sites where no measures were taken. Good estimates of yield are also needed to calculate the potential economic benefit of any rodent control method. This involves calculating the value of any extra crop produced, either in energetic or cash terms, and then weighing this benefit against the cost of the rodent management actions, including both materials and labour. In a final benefit to cost analysis, we might also consider other factors or side benefits, such as potential improvements to human or livestock health.

Although our primary focus is generally on crop losses caused by rodents, there are various situations in which it is necessary or advisable to measure rodent damage to crops. Firstly, measurements of damage are often necessary during the problem definition phase of a new project (see Chapter 1), where we need to get a quick quantitative estimate of rodent impacts across a range of crop types. By combining damage estimates with farmer knowledge of past and present crop losses, we can quickly build a good general picture of rodent impacts in a new area. A second reason why we might want to study rodent damage in addition to yield loss is to understand the relationship between the timing and intensity of crop damage, and any changes in rodent abundance, breeding activity and movement patterns. For example, we might wish to know whether rodent damage is more or less intense at particular stages of crop growth, and whether these periods are connected to the onset of breeding activity or to periods of dispersal. A third reason is that crop losses are caused not only by rodents but by other pests and diseases as well, hence some measurement of damage together with yield loss is necessary to attribute the losses to each of these factors. Finally, as we will explain in the final section of this chapter, the relationship between rodent abundance and crop damage in any given cropping system is of great theoretical and practical interest, as it is this relationship that will allow us to set targets for rodent control.

In this chapter, we describe some techniques that can be used to estimate both the level of rodent damage and crop loss in field crops, and the level of damage and loss to stored foods. You may need to modify or adapt these techniques to work in particular crop types or field conditions.

Methods for estimating damage

With experience, it is usually easy enough to distinguish damage caused by rodents from that caused by insects or other pests. However, quantification of rodent damage is complicated by two issues. The first is the complex relationship between the timing of the rodent damage and its impact on final crop yields. The second is the fact that rodent damage is often unevenly distributed within the agricultural landscape.

Timing of damage

Damage can occur at any time during the growth of crops through to the time of harvest. The impact of this damage on final crop yields will depend on both the severity and timing of the damage, and on the ability of the particular type of crop to compensate for any damage by putting on extra growth following damage.

In cereal crops, **growth compensation** has two components—tiller regrowth and panicle filling. Any tiller that is cut through by a rodent is likely to regrow. If this occurs before the maximum-tillering stage, the tiller may go through normal panicle initiation. These tillers may be shorter than undamaged ones but they often produce a normal-sized panicle. A tiller that is cut after the plant has entered the panicle-initiation stage generally will not be able to produce a new panicle. However, the plant may compensate for this loss by diverting its resources into the remaining panicles. This can lead to panicles with larger or more numerous grains. Once a cereal plant enters the panicle-ripening stage, it is unable to compensate for any subsequent rodent damage. Crop damage that occurs during the ripening phase will have the most immediate impact on crop yield. However, we should not underestimate the potential impact of damage at earlier stages. The point at which growth compensation will cease to be effective against rodent damage needs to be investigated for each crop type.

It is important to emphasise from the outset that assessment of crop damage at one point of time may not provide a good estimate of yield loss. For example, in rice crops, estimates of damage are usually taken in the week before harvest. This will only detect fresh damage and will not reflect the cumulative damage from the maximum-tillering stage through until harvest. The few estimates available for rice crops indicate that estimates of

damage taken in the week or so before harvest would need to be multiplied by four or five times to estimate yield loss. However, it should be noted that these estimates pertain to lowland irrigated rice crops and to damage mainly caused by *Rattus argentiventer*. Many more studies are needed of this important relationship.

Spatial distribution of damage

The distribution of rodent damage is often uneven within a single field or among a group of fields (Figure 9.1). In many cases, areas of particularly heavy damage are adjacent to local features that provide refuge or breeding habitats for rodents, such as large bunds or channel banks. However, in several parts of Southeast Asia, the highest rat damage is often found in the middle of rice fields rather than around the edges, producing the so-called 'stadium effect' (Figure 9.2). This unusual pattern presumably reflects some aspects of the feeding behaviour of the major pest species.

Damage assessment is simplest where the damage is randomly distributed in a field and more complex when it is very uneven or patchy in distribution (Figure 9.3). In the following sections, we will describe methods that are suitable for estimating damage that is randomly distributed within a field (Figure 9.3a) or distributed in a structured manner (Figure 9.3b–e).

Researchers have compared different sampling designs for plant disease and insects and found that sample size was more important than sampling pattern when the disease distribution was random, while the sampling pattern was more important when disease distribution was aggregated or patchy.
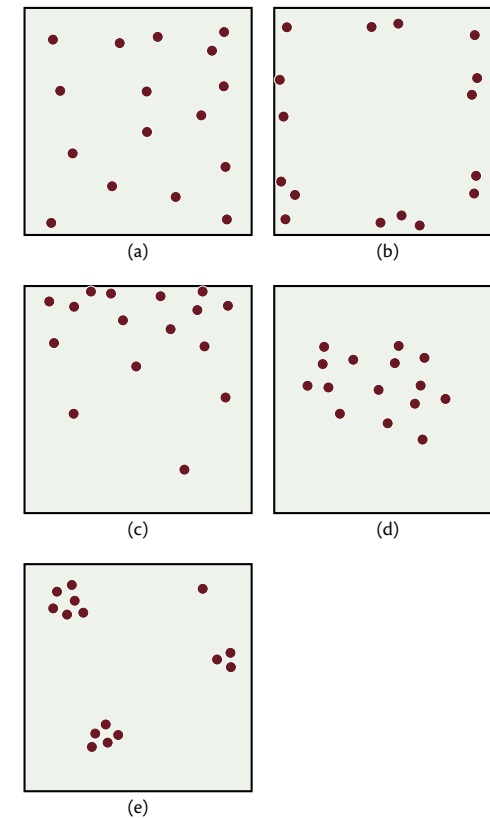
Figure 9.3 Five different patterns of damage within a field: (a) random damage; (b) highly structured damage (close to margins of field); (c) highly structured damage (decreasing away from upper margin of field); (d) highly structured damage (in centre of field — 'stadium effect'); (e) highly structured (clumped) damage.

Figure 9.1 Localised patches of heavy rat damage to rice in Myanmar. This damage was most likely caused by *Bandicota bengalensis*.

Figure 9.2 'Stadium effect' of rodent damage to rice in Indonesia (indicated by arrow).

Estimating damage at sowing/ transplanting

Rodents often damage crop plants immediately after sowing or transplanting, or during the first week or two of plant growth. The seeds or germinating plants provide a high-quality food resource for rodents. Typically, this damage results in the complete removal of individual plants. Provided that the level of damage is not too severe or where damage is evenly spaced, the remaining plants may be able to compensate for this damage by putting on extra growth. However, in some cases, the early crop damage is sufficiently heavy or in large enough patches to cause significant losses in crop production.

Early crop damage is generally easy to detect but difficult to quantify. We will describe separate methods that can be used for crops that are sown and those that are transplanted.

Sown crops

Many crops are direct seeded, either by broadcasting by hand, mechanised sowing, or dibble-stick methods. Rodents often enter the fields to dig up and consume the newly sown seed, or to feed on the early shoots after germination (Figure 9.4).

In order to estimate the level of damage to seeds or new shoots, we need information on (1) the quantity of seed that was spread; and (2) the number of seeds or seedlings that were removed or damaged. Obtaining this information can be difficult.

One way to estimate the proportion of seed lost to rodents is to compare the number of plants that germinate per unit area with the quantity of seed that was sown across the same area. Farmers

Figure 9.4   Examples of rodent damage to newly planted crops. Top: *Rattus argentiventer* footprints in a recently sown rice crop where seeds are germinating. Bottom: a newly sown wheat field in Australia, showing evidence of digging for seed by *Mus musculus*.

generally know the seeding density and it is easy to count emergent plants using a quadrat sampling method. However, this technique will overestimate the level of damage if some of the sown seed failed to germinate. (Our experience with wheat seed in Australia is that only 60–80% of sown seeds will germinate.)

An alternative way of measuring the extent of rodent damage to early crop stages is to compare the number of emerging plants in areas that have been damaged by rodents with areas that have been protected from rodent damage. The usual method is to set up exclusion plots (see below). Three or more exclusion plots are required to achieve adequate replication, and the unprotected crop should be sampled with quadrats of the same size as the plots. The distribution of these quadrats should adequately reflect the pattern of damage—either randomly placed if the damage appears to be randomly distributed, or arranged as a stratified random sample if the damage appears to be patterned in some way (see below).

Data from exclusion plots are used to determine damage according to the following formula:

$$\text{Damage rate } (\%) = \left(1 - \frac{\text{Number of plants in unprotected area}}{\text{Number of plants in protected area}} \right) \times 100$$

Transplanted crops

Rodent damage in a seedbed is probably best estimated by using the exclusion plot method.

For transplanted crops, the number of seedlings per hill and the density of hills are generally known to a fair degree of precision. Where such information is available, reasonable estimates of rodent damage can be obtained by using a quadrat sampling method. An exclusion plot method would probably yield more reliable estimates, but would involve considerably greater labour input and cost.

Exclusion plots

Exclusion plots are representative areas of crop that are protected against rodent damage by a rodent-proof fence or barrier. The reduction in crop yield caused by rodent damage is calculated by comparing the yield of the protected crop with unprotected areas in the surrounding field.

The main consideration when designing an exclusion plot is that the barrier will effectively stop all rodent pests from climbing, burrowing or gnawing their way into the enclosure. The choice of fencing material and dimensions of the barrier will need to take into account both the size of the particular pest species and its climbing and digging capabilities (see Box 9.1).

Box 9.1 Design of exclusion plots

Two examples will serve to illustrate some of these design principles. In Australian wheat crops, small plastic fences were built to protect small areas against damage by house mice, *Mus domesticus.* Each exclusion plot measured 2 × 2 m, from which the central area of 1 m^2 was harvested to determine yield. The plastic fence was 200 μm thick and 0.6 m high, with the bottom 10 cm dug into the ground. The fence was supported by metal fencing pickets and held taut against wind by wire strung through a fold along the top of the plastic and fixed to the top of the pickets. The plots were erected as soon as possible after sowing (i.e. the afternoon after the farmer had sown the crop). Exclusion plots were set at varying distances from the edge of the crop (e.g. 10, 20, 50 m), with two plots set at each distance to achieve replication. Counts of plants at emergence of the crop were conducted from the central 1 × 1 m area of the plot and related to the abundance of mice at different sites.

In deepwater rice crops in Bangladesh, researchers set exclusion plots that were 5 × 5 m (Figure 9.5). These were constructed of wire netting fixed to poles at the corners and sides, and with a 30 cm strip of galvanised metal sheet at the top to prevent rats from getting a foothold. The base of the fence was buried 10 cm in to the ground. Fences were made 195 cm high, which was 35–45 cm higher than the maximum flood depths of previous seasons. During peak floods, the fence was further extended using plastic sheeting. The full exclusion plots were harvested in this particular study to compare yields.
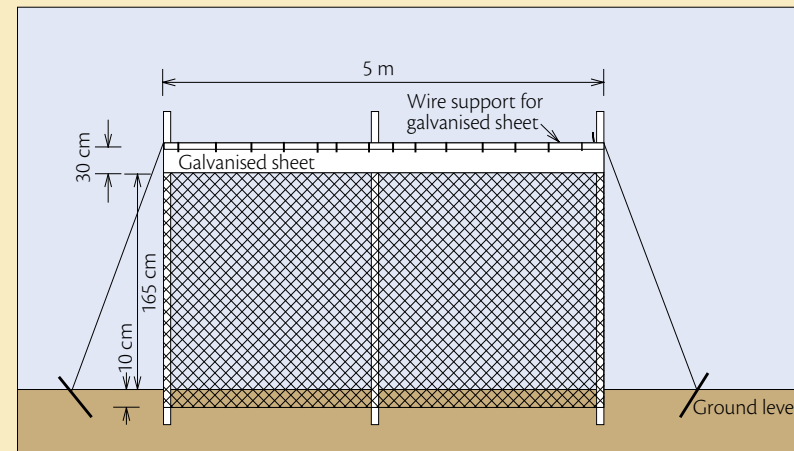
Figure 9.5 Wire exclusion plot used to protect deepwater rice from rats in Bangladesh (adapted from Islam et al. 1993).

Another important consideration is that the fence of an exclusion plot does not influence crop growth within the plot. This is probably of greatest concern where a plastic fencing material is used, because this may lead to changes in local air movements and light and humidity levels. One commonly used method for overcoming this problem is to calculate crop yields from the central portion of the exclusion plot, excluding any areas that grow close to the fence, so the larger the plots the better. Barriers constructed from open mesh wire are probably better in this regard, although they may not be effective against some rodents that have excellent climbing ability. All types of fences present the possibility that they may provide perching sites for birds, thereby increasing bird damage to the exclusion plot.

Estimating damage at later stages of cereal crops

Larger rodents usually gnaw through a panicle-bearing tiller near its base, leaving behind a neatly cut surface with a characteristic 45° angle (Figure 9.6). They then feed on the panicle where it falls or drag the tiller away to a safe place, such as a burrow. Very small rodent species, such as species of *Mus*, will climb the tillers and either snip away the panicle or else feed on individual grains without removing the panicle. Different methods are required to assess damage of each type.

We can estimate the proportion of damage to cereal plants at any particular stage of crop growth by examining a sample of individual plants. For each plant, we can record the number of tillers that are uncut, recently cut, previously cut and regrowing, or previously cut and not regrowing. The sum of these will give the total number of tillers for the individual plant. This is a laborious process when repeated on a large scale, hence you will need to decide how many times you can afford to repeat the process.

If damage assessment can be done only once, then we recommend that you do it as close as possible to harvest time. For the reasons set out above, this will provide a *minimum* estimate of yield loss.

If damage assessment can be carried out more than once, we recommend that it is done at the booting (panicle initiation) stage and again just before harvest. The choice of booting stage reflects widespread reports and field observations that rodent damage is particularly intense at this stage.

Figure 9.6 Rat damage to rice tillers (circled). Note the 45° angle of the cut through the tillers.

Different methods are needed to quantify direct panicle damage caused by mice or other small climbing rodents. For panicles at the ripening stage, the usual method is to estimate the proportion of grains that have been removed or damaged. For earlier stages of growth (e.g. tillers at the booting stage), you should count the number of damaged versus undamaged tillers.

Random and stratified random sampling

Random sampling methods are appropriate where the damage appears to be genuinely random in its distribution, even where the underlying landscape shows discrete structured variation. Selecting sampling points or quadrats in such a situation is simply a matter of deciding on a plot size (e.g. 1 m² quadrats) and sampling density (e.g. 5% of the total area), and then generating random numbers (from tables or by using a hand calculator) to identify the sampling points.

Stratified random sampling methods are appropriate where damage does not appear to be random, regardless of whether the underlying variation is discrete or continuous (Figure 9.7). An example of discrete variation is where rodent distributions are influenced by the presence of two or more clearly defined soil types or habitats (Figure 9.7a). An example of continuous variation is the distance of the

crop from an irrigation canal. Structured variation of this kind is illustrated in Figure 9.7b. Our recommendation is to always use stratified sampling.

The sampling procedure for stratified random sampling begins with the definition of the **strata**—the layers of variation within the landscape. If these are discrete, you will first need to calculate the relative proportion of each stratum within the total study area. The number of sampling points or quadrats placed within each stratum is then scaled to reflect these relative proportions. This scale may be a direct proportional one, or it may be biased towards greater sampling of the more common units or greater sampling of the less common units, depending on the primary purpose of the sampling. Within each stratum, sampling points or quadrats should be chosen randomly, using the process described above.
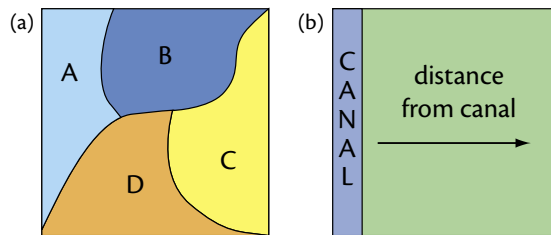
Figure 9.7   Two kinds of underlying structure that may exist within a landscape. Examples of **discrete** or categorical variation (a) would be areas planted with different crop types or with underlying soil types. **Continuous** variation (b) is observed along a gradient away from a key factor—in this case, the distance from refuge habitat along a major canal.

For sampling of continuous variation, you will first need to decide upon the key factor that encapsulates the variation. In the example introduced above, this would be the distance from the irrigation canal. The next step in this example would be to decide whether the gradient of variation is likely to be a linear function of distance, or some more complex function. An appropriate sampling density at any point along the gradient is then decided on the same grounds as those already mentioned for the sampling for discrete strata. Once again, sampling points or quadrats should be chosen randomly at each position along the gradient.

In a situation where it is not obvious whether rodent damage is spread randomly or unevenly within a field, we recommend that stratified random sampling is used. The results of this sampling will always be equal to or better than the precision that you can get from random sampling with no stratification.

A worked example of stratified random sampling of rodent damage in a rice field is given in Box 9.2.

Where the rodent damage in a field is obviously very patchy (e.g. Figure 9.3e), neither of the fully random nor stratified random sampling methods is likely to give a reliable estimate unless the sampling density is extremely high. In such a case, you may need to consider using a different approach based on the principle of adaptive sampling (for details, see Krebs 1999, Ecological Methodology, Chapter 8). However,

all of these methods are more complicated to apply in the field and we do not recommend their use unless you think the stratified random sampling method is giving very inaccurate damage estimates.

Estimating damage to vegetable and upland crops

Methods used to assess damage in vegetables and other upland crops need to embody the same principles of randomisation and adequate sampling intensities as those used for cereal crops. However, two factors combine to make the process of quantifying rodent damage to these crops more straightforward. The first is that the damage generally affects the fruits, pods, cobs or tubers (see Figure 9.10), and rarely has any significant effect on vegetative growth. Because a damaged tuber or fruit is generally not considered edible or saleable, simple counts of the numbers of damaged versus undamaged fruits or tubers are generally adequate to quantify the extent of damage. However, this approach fails to take into account any potential compensation in the size of remaining undamaged fruit or tubers following damage.

Any method for scoring damage may need some adjustment for particular kinds of vegetable and upland crops. For example, damage to maize crops is usually counted as the number of cobs on fallen tillers and gnawed cobs on standing tillers. Damage

Box 9.2 A protocol for stratified random sampling of rodent damage in rice crops

We have used the method described here to quantify rodent damage to transplanted rice crops in Indonesia and Vietnam. In both of these studies, the level of rat damage was usually highest in the middle of the crop and lowest around the edges.

The method is an example of a stratified random sampling approach based on a continuous gradient—distance from the edge of the field. Our example is based on sampling of a rectangular field with dimensions 500 m × 300 m.

To begin, establish a baseline along the long axis of the field. Set four transects perpendicular to the baseline, running in from the edge of the crop (Figure 9.8), and spaced at 20 m intervals. If possible, try to keep transects 1 and 4 at least 50 m away from other roads, major channels or villages, as these may produce

atypical levels of damage. To fully sample the variation within the field, we would define and sample the five strata that represent five equal-width zones from the edge to the centre of the field (Figure 9.8).

At each point, assess 10 plants along a line perpendicular to the transect. Score every fifth plant, as shown in Figure 9.9.

For each plant, count the numbers of:
- tillers with recent damage by rodents
- undamaged tillers bearing mature panicles
- undamaged tillers that either lack or bear immature panicles (perhaps indicating earlier damage by rodents).

Record the information on a standard damage assessment data sheet (an example is provided in Appendix 4).

Sampling of 10 plants at each sampling point provides an estimate of the proportion of tillers damaged within each stratum. The four transects are replicates, so for each of the five strata we have examined 40

plants for damage. The total number of counted plants is 200 for the entire field. Each plant examined will have one or more tillers. Given these data, the estimated proportion of rodent damage for the entire field is given by the equation:

$$\hat{p}_{ST} = \frac{\sum N_h \hat{p}_h}{N} \tag{1}$$

where:

$\hat{p}_{ST}$ = Stratified mean proportion damaged by rodents
N_h = Size of stratum h (in number of sample units)
$\hat{p}_h$ = Estimated proportion damaged for stratum h
N = Total field size (in number of sample units)

The standard error of this stratified mean proportion is:

$$SE(\hat{p}_{ST}) = \frac{1}{N} \sqrt{\sum \left[\frac{N_h^2 (N_h - n_h) \hat{p}_h \hat{q}_h}{(N_h - 1)(n_h - 1)} \right]} \tag{2}$$

where:

$SE(\hat{p}_{ST})$ = Standard error of the stratified mean proportion
$\hat{q}_h = 1 - \hat{p}_h$
n_h = Sample size in stratum h (= 4 in this case)

and all other terms are as defined above.

Figure 9.8 Layout of transects to measure damage by rodents in a rice field.

Figure 9.9 Measure damage on every fifth plant.

Box 9.2 (cont'd)

Confidence limits for the stratified mean proportion are obtained using the *t*-distribution:

$$\hat{p}_{ST} \pm t_{\alpha}(\text{standard error of } \hat{p}_{ST}) \qquad (3)$$

The additional information needed to calculate the stratified mean proportion of damage is the size of the five strata and the total size of the field, and the key point is that these 'sizes' must be in 'sample size units'. If the five strata occupy equal areas within the sampling area, the size of each stratum is 20% of the total sampling area, so only one parameter must be estimated. If you know the size of the sampling area in m², and you know the average area sampled to obtain 10 plants, you can get the total size of the field in sampling units by division. Note that these estimates can be approximations, and the resulting damage estimate is robust.

To estimate damage in a field, the area represented by each stratum must be calculated, taking into account the geometry of the field. Note that the outer strata (i.e. those close to the edge) will cover larger areas than those towards the centre of the field.

assessment for cassava and other root crops may need to use another measure based on counts of underground tubers, while damage to groundnut could be based either on counts of damaged pods or on counts of lost or damaged nuts.

Damage in vegetables and other upland crops is generally assessed shortly before harvest. The simplest method involves the use of *transect counts*, as illustrated in Box 9.3. Other useful methods include *quadrat sampling* and *variable area transects*. References to these alternative methods are included in Further reading.

Figure 9.10   Examples of rodent damage to maize (top) and ripening tomato (bottom).

Estimating preharvest yield loss

There are two established methods for estimating yield loss.

The first method is to convert damage estimates into yield losses. As already mentioned, this relationship is complicated by two factors:

- the possibility that damage has occurred throughout the growing period, with a cumulative effect on yield at harvest
- the phenomenon of growth compensation by plants following damage.

One way of learning about the relationship between damage and loss is by simulating rodent damage to crops. Experiments have been conducted in which rice plants were cut experimentally at different intensities and at different stages over the growth period of the crop. As expected, the results showed that damage inflicted at later growth stages caused a proportionally greater reduction in yield at harvest than damage at earlier stages. Compensatory growth was observed in all treatment plots and the yield was fully compensated if damage occurred early. Studies of this type can be conducted relatively easily, but sufficient people are required to implement the treatments.

Box 9.3   Transect method for damage assessment in vegetables and other upland crops

This method is easiest to use in well-organised fields with crops planted in straight rows. However, the same method can be adapted for less structured fields.

Divide the area of crop into four equal-sized areas and assign one transect to each area (Figure 9.11). Begin each transect away from the edge of the plot. Assess rodent damage on every second plant until

you obtain estimates for 10 plants on each transect. For each plant, count the number of tillers, pods or cobs damaged by rodents and the number of undamaged tillers, pods or cobs. Record all the information on a standard damage assessment data sheet (see Appendix 4), taking care to record the crop type. The data can be analysed in the same manner as shown in equation 1 in Box 9.2.
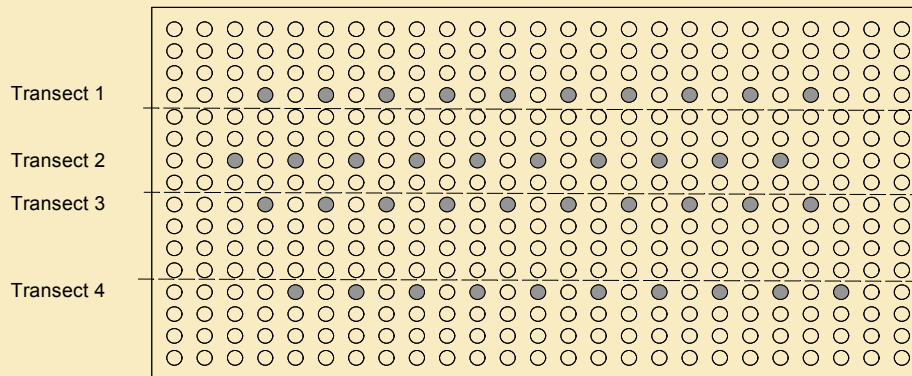
Figure 9.11   An example of the layout of the four transects to sample for damage in small vegetable plots.

Estimating postharvest damage and loss

The second method is constructing exclusion plots (see Box 9.1). This provides a more direct way of calculating yield loss but care is needed to ensure good experimental design, including sufficient replication.

Postharvest damage to stored vegetables or fruits is usually obvious from the signs of gnawing. In contrast, damage to stored cereal grain is not so easily observed and often must be inferred from general signs of rodent activity in and around the storage containers, such as the presence of faeces, hairs or urine smears. Contamination of stored grain reduces its value and the presence of rodent saliva or urine also poses a risk for the transmission of diseases.

Postharvest losses are rarely taken into account in the calculation of rodent impacts. This situation reflects two deeply held beliefs. The first is that total postharvest loss is often difficult to estimate with any degree of reliability. The second is that rodent damage to stored grain is difficult to distinguish from damage caused by other pests. Although there is some element of truth behind both of these beliefs, there are also experimental approaches that may help overcome the difficulties.

The reliability of estimates of postharvest loss to stored grain is really a function of the level of record keeping of ingoing and outgoing produce. In a situation where produce is stored for a period before sale, the calculation of total loss is generally straightforward, assuming that the quantity of harvested crop and the quantity sold are both reliably documented. Contamination of produce during its time in storage may also reduce its final sale value. Although the total financial loss may be easy to calculate in such cases, it may be difficult to attribute this loss to any one particular pest.

Much greater difficulty will be encountered in situations where stored grain or other crop produce is used either exclusively or primarily for household consumption. Under these circumstances, the crop produce is generally held for long periods and used in small amounts each day. Records of usage are rarely, if ever, kept, hence it is often very difficult to calculate exactly how much of the stored crop has been used by the household and how much has been consumed by rodents and other pests.

One method that is currently being trialled in several parts of the world is to monitor grain loss from a container placed within the larger storage area. This method is described in some detail in Box 9.4. Limitations of the method include the fact that it is only sensitive to losses from the open surface of the store, and would not record losses from penetration of the store container from below, and the possibility that feeding from the container either occurs at higher or lower intensity than that from the general surface of the store. We have tried to estimate the extent of any feeding bias by measuring the level of contamination by hairs and faeces of both the container and the surface of the general store area. The results obtained thus far from these studies appear promising.

The relationship between rodent abundance and rodent damage

Although it is probably true in general terms that more rodents will produce more damage, this relationship may not be a simple linear one. Many aspects of rodent ecology and behaviour are **density-dependent**, which is to say that they change in response to changes in population density. A simple example is a shift in diet from one preferred food item to a broader range of foodstuffs as population pressure starts to limit access to the various food resources. Another example might be a decrease in the breeding rate among adult females as population densities rise, perhaps due to competition for nesting sites or to increased social tensions. These complex ecological and behavioural interactions may lead to variable levels of crop damage at different population densities.

There are two reasons why it is important to understand this relationship. The first is that we might be able to predict the likelihood of serious crop damage based on some information on rodent abundance. For example, if we know that critical levels of crop damage are only likely to occur if population density exceeds a certain threshold level (e.g. >20 individuals per hectare), we may wish to monitor rodent abundance during the early part of a season and then use our knowledge of potential population growth rates to forecast the likelihood of serious damage. Typically, this information would be fed into a decision analysis that also included the cost of any rodent control actions and the potential losses associated with not taking those actions.

The second reason for wanting to know about the relationship between rodent abundance and damage is to set appropriate management goals (Figure 9.12). To illustrate this process, let us assume that our management goal is to keep rodent damage below a certain specified level, such as below 5% (measured in a standard way). Our first step would be to consult the relationship between rodent abundance and damage to estimate the corresponding population density. We would then ask what management actions would be required to keep the rodent population density at or below this level. If the cost of these actions was unacceptably high, we might then revise our original goal to find a point where the benefit to cost ratio is acceptable (e.g. keep damage below 10%).

Box 9.4   Estimating postharvest loss from a grain store

The method described here is suitable for monitoring loss from open storage units of the kind used widely across South and Southeast Asia. These units are generally made from woven bamboo, sometimes sealed with mud or animal dung. The storage units are often raised on stilts or placed on a low platform but they are sometimes placed directly on the ground.

A wide close-weave basket of known diameter and weight is partially filled with a standard quantity of unmilled paddy rice (e.g. 5 kg). This is placed on the surface of the rice within the storage container. The store owner is asked not to take rice from or add rice to the basket. The basket with its contained paddy is weighed at regular intervals to chart the rate of loss of grain (Figure 9.12). When the quantity of remaining paddy falls below a certain level (e.g. below 1 kg), the basket is refilled to its original weight. The moisture content of the rice in the basket and near the surface of the grain store is recorded each time, using a standard field gauge. These values allow the weight of the basket rice to be adjusted as necessary to match that of the general stored rice.

The rate of consumption of paddy from the basket is calculated as a loss rate per unit surface area (e.g. if 0.5 kg of rice is removed over an 8 week period from a basket with a surface area of 0.5 m², the loss rate is then 0.125 kg/m²/week). This value can be multiplied by the surface area of the grain store to calculate an overall rate of loss from that store.

This method relies on several critical assumptions. The first is that the rate of consumption from the basket is equivalent to that from the surface of the wider grain storage. This may not be the case if the rodents either feed preferentially from the basket or else avoid the basket. A method to control for any bias in feeding location is discussed below. The second is that no grain is lost from damage to the base or sides of the grain store. This may be difficult to monitor where the store is placed directly on the ground. The third is that no loss occurs as a result of animals scuffing rice out of the basket. This is more difficult to control, other than to make the basket quite deep, at the risk of reducing access by some species of rodents.

If feeding in the basket is a truly random sample of behaviour within the store, we could expect the same level of contamination in each, and the same level of damage to remaining grains. To measure contamination, we take a standard container (e.g. a cup) from each of the basket and the surface of the surrounding grain store and count faeces and hairs in both samples. We then take a subsample of 100 paddy grains and count the number of unfilled grains, the number showing rodent tooth marks and the number showing insect damage (typically visible as bore holes). If we do find a difference in the level of contamination or damage, we would then need to consider whether the level of contamination is proportional to the amount of feeding activity. A good way to start would be to ask whether the level of contamination and damage are correlated across a range of replicated samples.

Figure 9.12   Postharvest loss experiment in Bangladesh.

The same approach might be used in a crisis management situation where high levels of rodent damage are occurring. In such contexts, we would consult the relationship between rodent abundance and damage to find out what proportion of the existing population would need to be culled to bring the population density back to acceptable levels of associated damage. This information would help us determine appropriate methods and to estimate the cost of the necessary actions.

To determine the relationship between rodent abundance and damage for any particular combination of rodent species and cropping system, you will need data from across over a wide range of rodent population densities and levels of damage. As explained in Chapter 5 and in this chapter, each of these measurements is fraught with complexity, related to factors such as growth compensation and availability of alternative food in the case of rodent damage, and trappability and the mobile and highly dynamic nature of the rodent population itself in the case of rodent abundance. To accommodate this variability, it will be necessary to obtain numerous data points, which in turn implies considerable field effort. However, as indicated above, the heuristic value of the relationship between rodent abundance and damage is sufficiently great that the effort will be richly rewarded.

In **Model 1** (Figure 9.13, left), damage is directly proportional to the abundance of rodents, up to a point where 100% of the crop is damaged. Below this threshold, a reduction of rodent abundance by a given percentage will result in a reduction of damage by the same percentage. Above the threshold, the reduction of damage will be less than the reduction of rodent abundance.

In **Model 2** (Figure 9.13, centre), the amount of extra damage decreases as rodent abundance increases (this might occur if it becomes progressively more difficult for rodents to find undamaged plants).

Under this model, the proportional reduction in damage will always be lower than the reduction in rodent abundance, but particularly so in the upper part of the curve.

In **Model 3** (Figure 9.13, right), rodent damage increases more rapidly above a certain threshold in rodent abundance (this might occur if rodents switch to eating and damaging the crop only above a certain population density). In this situation, any decrease in rodent abundance will result in a proportionally higher decrease in damage, especially if rodent abundance moves from above to below the threshold value.

There are likely to be other types of abundance–damage relationships. By knowing the shape of the curve in any particular situation, it should be possible to develop targets for control. A critical value for developing targets is the threshold of damage that farmers are willing to accept.
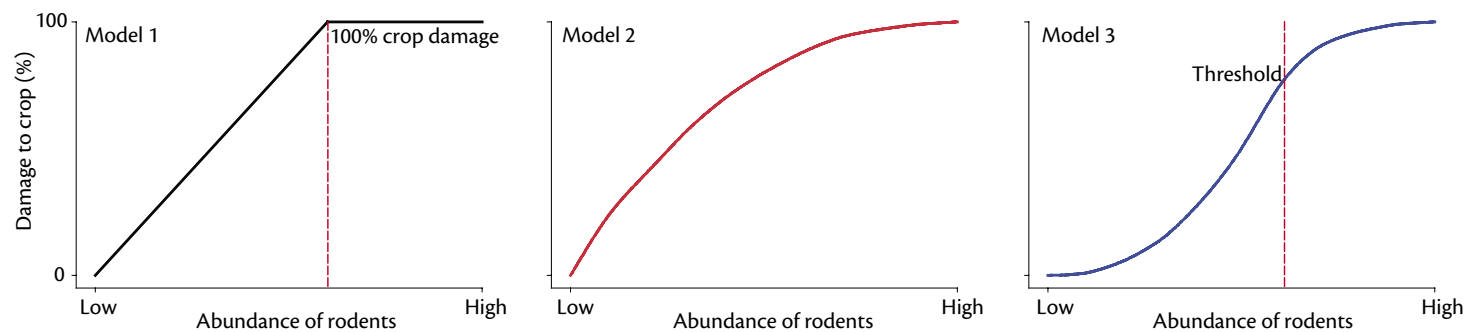
Figure 9.13   Conceptual models of the relationship between rodent abundance and rodent damage to crops.

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CHAPTER 10

The 'human factor' in rodent management studies

Introduction

In Chapter 1, we suggested that the viability of any rodent management option should be judged against each of three criteria:

- ecological sustainability
- cultural acceptability
- socioeconomic sustainability.

So far in this volume, we have focused on methods that will allow you to gather information relevant to the first and last (in part) of these criteria. In this chapter, we introduce some methods that should allow you to explore the cultural and socioeconomic context of rodent management. Our treatment of these methods is much less comprehensive than for the biological and agricultural methods. This is partly because we do not have specific expertise in these fields. However, it is also because the consideration of cultural and socioeconomic factors is a relatively new development in the area of agricultural research in general, and even more so in the field of rodent management. We hope that publications listed under Further reading will provide interested readers with a pathway into relevant literature.

A conceptual framework

We can learn much about cultural and social issues by listening to what people say and by observing what they do. In the context of rodent management projects, where the research phase is often of quite short duration, the challenge is to organise and interpret these observations in ways that help us to understand why people would choose to take certain actions and not others.

A useful conceptual framework is available from previous studies of farmers' beliefs and associated decision-making behaviour in relation to insect pest management. These studies in turn draw upon a much larger body of theoretical literature related to **decision-making** as a process or system.

Two simple flow models help to illustrate how decision-making theory can help make sense of human behaviour. The first is an example of what is termed a 'belief model'. It illustrates the notion that people's behaviour is influenced by their perceptions of risks and benefits associated with particular pests and management actions. Each of the four major components of this model (Figure 10.1) can be quantified to some extent, either by calculating the monetary value of potential benefits or losses, or by ranking the importance of various influences on a subjective scale (i.e. as more or less important).

The second model illustrates a broader 'theory of reasoned action' (Figure 10.2). This model emphasises the social context of human behaviour by indicating that a person's behaviour is often a compromise between what they would like to do, based on their personal preferences, and what they feel they 'should' do, based on the beliefs, attitudes and values of other family members, neighbours and the wider society. This compromise is mediated by the strength of each individual's motivation to comply with the societal pressures or 'norms'.
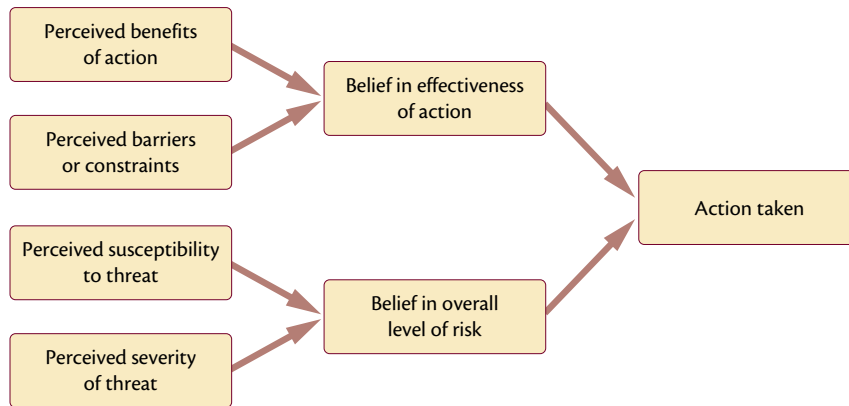
Figure 10.1   Belief model designed for studying behaviour in regard to pest management (modified from Heong and Escalada 1999, Fig. 1).
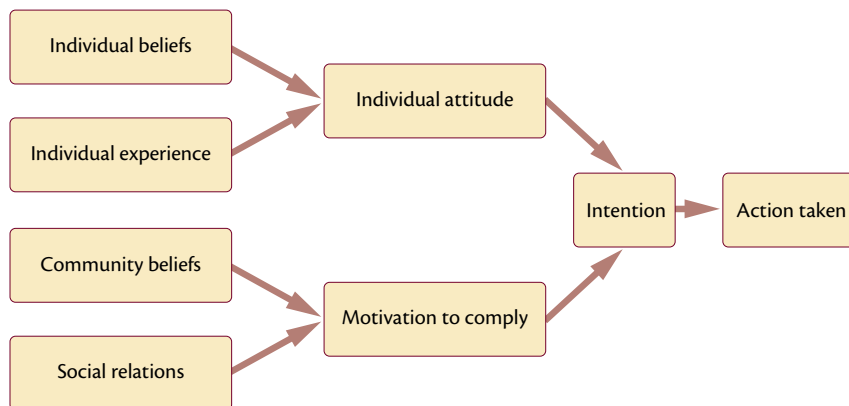
Figure 10.2   Behavioural model based on the theory of reasoned action (modified from Heong and Escalada 1999, Fig. 2).

Some basic tools and methods

Many of the tools and methods that we recommend for exploring the socioeconomic and cultural issues associated with rodent management have a long history of use in the field of participatory research (see Box 10.1). A good general introduction to participatory methods is found in another recent ACIAR monograph (see Further reading: Horne and Stur 2003).

An important aspect of participatory methods is that they allow community members to contribute both to the recognition of problems and to the development of solutions. This creates a sense of ownership and understanding that builds their confidence and capacity for learning. The participatory methods also may help build a close relationship among the team members, such that improved communication can take place in an atmosphere of mutual trust and respect.

Community resource maps

These are a good way to begin in a new project area. You will need large pieces of paper and pens or crayons. Invite a small, representative group of local community members (a 'focus group') to draw a map of the important physical features and resources used by their community. This would normally include infrastructure, such as buildings, roads and canals, and the location of major cropping areas. It should

also include features that are particularly important to rodent ecology, such as food storage areas and any areas that people regard as significant breeding habitat. A number of different people should be asked to contribute to the map in order to achieve a balanced representation of local resources (Figure 10.3). The process of compiling the map itself may help you understand how local resources are structured and accessed by different groups within the community and how different people perceive the nature of their rodent problems. The completed map also can be used as a reference point for subsequent activities such as construction of the seasonal calendar and in problem diagnosis.

Figure 10.3 Participants conferring on their community resource (village) map during a focus-group session in Cambodia.

Box 10.1 Participatory approaches to research

Participatory approaches to agricultural research and development (R&D) arose in the 1980s as it became clear that the adoption of various new technologies by farming communities—especially in traditional smallholder farming systems—was not always as rapid or as high as expected by those who developed them. Researchers started to question whether the traditional R&D approach, where scientists develop new crops or associated methods on research farms and then 'release' them into the wider world, was really the most effective way to help the rural poor. Might it not be better to first consult with farmers about their problems and priorities, and perhaps even to explore the appropriateness of possible solutions before investing time and effort into their development?

From these early steps, a whole new area of research methodology has developed within which we can distinguish various contrasting approaches, such as farmer participatory research (FPR), action research (AR), adaptive management (AM), and even active adaptive management (AAM). These methods share a common emphasis on interaction between the developers and the potential users of proposed new technology or practices, but they differ in two main respects. The first is the nature and extent of the interaction among the various stakeholders (researchers, extension staff, users), which ranges from a process of consultation through to a true partnership or collaboration. The second is the nature of the research process itself, which follows fairly traditional lines (i.e. hypothesis formulation and testing) under FPR, but leans towards the immediate implementation and progressive readjustment of management actions under AR, AM and AAM. However, it is generally agreed by those working in the area that there is no one 'right' way to do participatory research and that the choice of method should depend on both the goals and objectives of the project and the particular socio-cultural context.

Seasonal calendars

A seasonal calendar is a simple graphical representation of the important environmental, agricultural and social events that take place during the course of a typical year. The same group of community members that produces the village resource map often produces the seasonal calendar. The same basic materials are required.

A good way to start is to ask when the new 'year' is thought to begin—this may be the planting time of a particular crop, or it may some astrological event such as Lunar New Year. Using this as a starting point, draw a matrix with months along the top. Then invite the focus group to identify the major crop types and write these down the margin. For each of the crop types, the growing phases and

associated activities should be recorded month-by-month across the page. For example, in a lowland rice crop production area, the major growth phases are tillering, panicle initiation, ripening etc., and the key activities would typically include seed-bed construction, land preparation, transplanting, weeding, harvest, and threshing. At this stage, it may be useful to ask participants to indicate the timing and severity of rodent and other pest damage in relation to each crop. Because these problems are often more severe in some places than others, it may be useful to relate these observations back to the community resource map (Figure 10.4). At the same time, you might ask general questions about the methods that people are currently using to manage rodents in the various habitats.

Figure 10.4 Cambodian focus-group participants relating their seasonal calendar back to their community resource map.

Major environmental events such as the start of the wet season and likely periods of flooding or water shortage should also be recorded on the calendar. Finally, the calendar should record other key activities that might require significant investment of labour or cash (e.g. fishing, craft activities) or periods of involvement in social activities such a festivals or community work.

Historical calendars

A historical calendar attempts to document some of the major events or changes that have affected a community's livelihood in the recent past. A first draft is often produced in the context of a focus-group meeting, but the calendar can be revisited many times on the basis of new information from as many different individuals as possible. Individual interviews should be sought with the oldest men and women in the community.

A good way to begin is to ask about the visible infrastructure. When did the community come to occupy its current location? When was the school built? By referring to the community resource map, you could ask when certain resources were developed (e.g. when a canal was built, when a particular cropping area was established). A next step could be to ask about major environmental events such as major floods, serious droughts, or particularly extensive forest fires. In many upland areas of Southeast Asia, people will often identify major

rodent outbreaks as a kind of historical disaster, but at least in the first instance, you should not prompt such observations but rather allow them to emerge. It is natural for people to emphasise what the particular researcher wishes to hear, hence it is important to avoid leading questions.

In many areas, major political events and associated displacement or movement of people may also have played a major role in shaping the present cultural landscape. You may need to explore these factors in a sensitive manner and perhaps through individual interviews rather than in a group context.

Once the general history of the community is established, you might then inquire about some of the more subtle changes that may have affected people's livelihoods. How has their access to markets changed over the years? Have they been placing more emphasis on certain crops at the expense of others? Have they changed their residential pattern or style of housing or storage of foods? What kinds of rodent control activities did people practise in the past as compared to now? For each of the important changes or trends, you should try to establish a general time frame for the events.

Decision analysis matrices

A decision analysis matrix is simple tool for obtaining an overview of the factors that influence decisions by farmers on their current actions of rodent

management. This activity is best done at a focus-group meeting. Ask the farmers to list the type of management actions they use to control rodents. This list should include occasional actions, including those that are only used in years when rat numbers are very high.

Once a basic action list is developed, ask the farmers—for each action—when it is taken, where it is taken (including scale of action), by whom is it done (individual male and/or female farmers;

groups; the whole community), whether it is affordable (in terms of economic benefits versus cost), whether it is feasible (e.g. labour available at the right time; water available for early planted crops to attract rats), whether it is socially and politically acceptable (likely response of neighbours, the wider local community and the government), and whether it has any environmental impact (beneficial or adverse). An example is shown in Table 10.1. Enter the information into a large-format table that

everyone can read. Encourage people to comment on the information at any stage during the process.

Once the table is complete, ask the focus-group participants to prioritise the current management actions. Which ones do they consider the most important overall for rodent management, and which ones are less important? The ensuing discussion about priorities will often provide important insights into why certain decisions are made by individuals or by the community as a whole.

Table 10.1   Decision analysis for San Jacinto/San Jose, Pangasinan Province, the Philippines, of current actions plus proposed use of a community trap–barrier system (CTBS). Note that the scale of most actions is currently at the individual farmer level.

Actions (what)	Timing (when)?	Who?	Where?	Feasible?	Affordable?	Socially OK?	Politically OK?	Environmentally OK?	Priority
1. Maintain cleanliness (banks, villages etc.)	Year-round	Farmer	Whole village	Yes	Yes	Yes	Yes	Yes	High
2. Rat hunt (dig/flood burrows)	Oct/Nov	Farmer	Major banks	Yes	Yes	Yes	Yes	Yes	Medium
3. Rat drive	Oct/Nov & Mar/Apr	Community	Major banks, long grass	Yes	Yes	Yes	Yes	Yes	Medium
4. Small dikes	Land preparation	Farmer	Small banks	Yes	Yes	Yes	Yes	Yes	High
5. Zinc phosphide	Before harvest	Farmer	Rice fields	?	If >5% loss	?	Yes	No	Medium
6. Racumin	Before harvest	Farmer	Rice fields	?	If >5% loss	?	Yes	No	Low
7. Biological control	Year-round	Farmer	Rice fields	Yes	Yes	Yes	Yes	Yes	High
8. Rat traps	Year-round	Farmer	Rice fields	Yes	Yes	Yes	Yes	Yes	Medium
9. Fumigation	Dry season after harvest	Farmer	Banks	Yes	Yes	No	Yes	Yes	Low
10. Crop timing	Planting	Community	Whole village	Yes	Yes	Yes?[a]	Yes	Yes	High
11. CTBS	2–3 weeks before main crops	Community	Whole village	Yes[b]	Yes	Yes	Yes	Yes	High

[a] Price of rice could drop if everyone harvests at the same time.

[b] Depends on the availability of early irrigation water.

Social mapping and wealth analysis

In many societies, there are obvious differences in livelihood status between individuals or between families. If your goal is to improve the livelihood security of all, or most, members of a community, then it is important that you try to understand the basis of these differences. Knowledge about the social and economic structure of a community should help you to develop new approaches that are appropriate to the resources of the wider community and which do not further disadvantage those who are already worst off.

The pattern of social organisation of rural communities is often highly complex, with a number of parallel systems based on ethnicity, systems of familial descent through either male or female lines, and systems of official accreditation based on government appointments (e.g. village headship, teachers). In addition, communities that have received people from other regions as a result of dislocation sometimes have an added historical element (poorer families have often arrived most recently). Many, though not necessarily all, of these factors may influence a family's degree of access to particular resources such as land, water or labour.

Wealth is generated and controlled within a traditional social system, but wealth can also alter the traditional balance. Increasing access to market economies can sometimes allow people to gain access to external funds (e.g. through the sale of craft materials) which can then be used to gain access to new resources such as hired labour and improved quality seed. In many societies, the systems of social and economic influence are going through a process of rapid change.

Social mapping and wealth analysis are two tools that can help you to understand the complex socioeconomic relationships within and between communities. The challenge in a new project is to quickly identify the most critical opportunities and constraints, but to do so without impinging on sensitive issues.

A good way to begin with social mapping is to ask specific questions about the community resource map. If a large canal passes through the cropping areas, it would be worth asking an open-ended question about usage of the irrigation water. For example—Who uses water from the canal to irrigate their crops? If the answer is that only some people do, then you can follow up with questions that are more probing—What percentage of farmers use the water? What is the relationship among the farmers in that group? A series of general questions about access to key resources should help you to build a general impression of how the community is structured. If possible, it is a good idea to test your ideas through a series of individual interviews, ideally with people representing the full socioeconomic spectrum.

Wealth analysis is a tool that can help you to understand the economic circumstances and capacity of various 'wealth groups' within a community. What is it, in terms of possessions or access to resources, that distinguishes the poorest members of the community from those who are moderately well off and those who are considered to be best off? A wealth analysis can also begin with small group discussions. You could start by asking the participants to each write down the economic attributes of the poorest and the richest families within the community. If possible, this should be done without reference to individual families. You can then assemble these results onto a larger sheet as a series of hypothetical gradients (e.g. possesses no livestock versus owns herd of water buffalo). Each of the key parameters can then be discussed in turn to identify what pathways might exist for someone who would wish to improve their livelihood status. Hypothetical discussions of this kind can reveal much about the socioeconomic dynamics of the community.

Problem-cause diagrams

A problem-cause diagram is a graphical representation of the causes and effects of a particular problem, as perceived by the members of the community. The diagram is typically developed by a focus group, with assistance from a facilitator.

You will need a large board, some cards and marker pens or crayons. The process starts with identification of a specific problem. Try to avoid making this too general (e.g. 'Rats'). In our experience, a more specific problem makes a better starting point. For example, 'Rats attack our dry season crop' or 'Rats eat our stored grain'. Write the problem on a card and stick it to the middle of the board. Then ask the focus group to identify the causes of this problem. Write each cause on a card and pin it above the problem. Oftentimes, focus-group members will be aware that the various causes are interrelated; these linkages should be discussed and indicated by connecting arrows (Figure 10.5a). When this discussion starts to become repetitive, ask the focus group to think about the effects or impacts of the problem. Write these on cards in the same manner and attach them below the problem (Figure 10.5b), again indicating any cross-links that the focus group are able to identify. Remember that this should be a representation of local perceptions of the problem, so be careful to avoid leading questions or adding your own causes, effects or linkages.
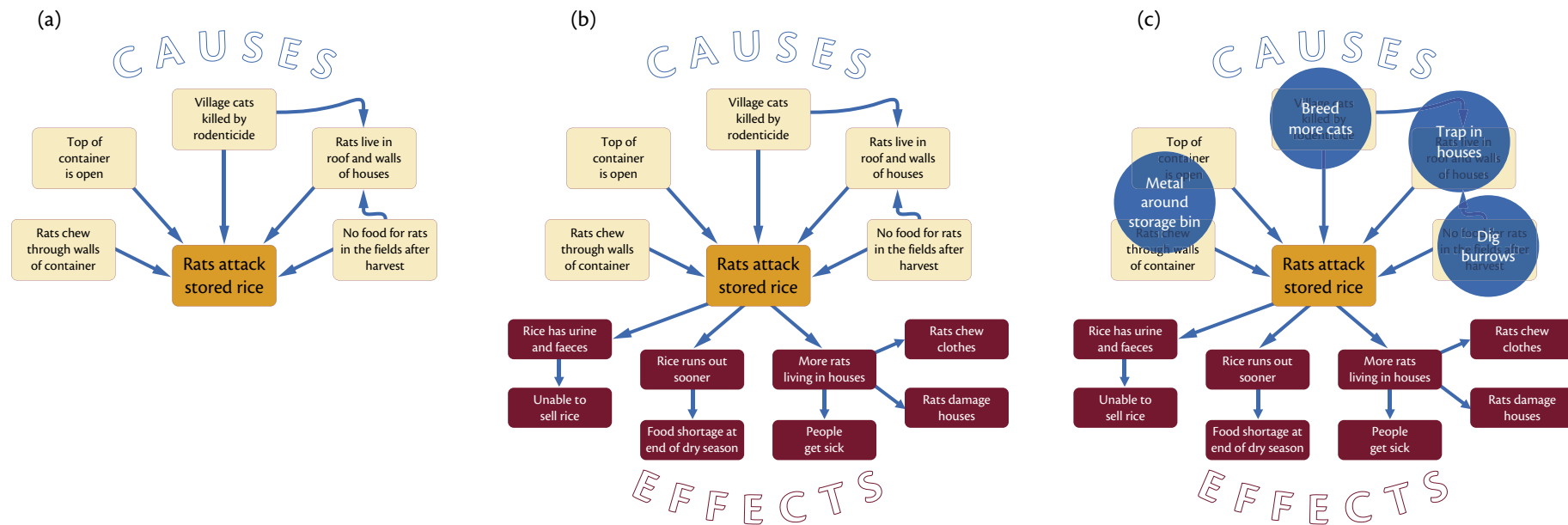
Figure 10.5 An example of a problem-cause diagram created by a focus-group around the problem 'Rats attack stored rice'. As a first step, the focus group have identified five possible causes of the problem (a), some of which are thought to be interlinked. Next, a range of impacts are identified (b), again with some perceived links. In (c), current actions are added to the diagram, the placement indicating the rationale behind each action.

Next, ask the focus group to indicate what they are currently doing to combat the specific problem. Write the current actions on cards and place these over the top of relevant causes (Figure 10.5c). As a final step, you could ask the focus group to think about any other possible actions that might have been tried but abandoned, or discussed but not tried. You could also ask the focus group to speculate as to why these other actions may not be appropriate. The completed problem-cause diagram can be shown to other groups and individual people within the community to gauge the level of representativeness of the focus-group perceptions.

Problem-cause diagrams are a useful method for exploring local knowledge and perceptions of how the agricultural and natural system works, and of finding out about current practices. They can also form a good starting point for subsequent discussions about the project. For example, you could use the diagrams to explain why a particular piece of ecological research is needed, or why certain experimental trials are being conducted. In the later stages of a project, the diagrams might also form a starting point for discussions about the possible benefits and pitfalls of potential new approaches to rodent management.

Individual, structured interviews and KAP questionnaires

The methods discussed above all begin with focus-group discussions as a way of gathering general information and forming a broad impression of the socioeconomic dynamics of a community. Although these activities can be run in ways that reduce the potential influence of one or two dominant individuals, they nonetheless rely on limited and possibly non-representative sampling of opinions within a community. One common means of increasing the sampling of community opinions is to use individual, structured interviews, based on a number of carefully framed but pre-set questions contained within a questionnaire.

One particular kind of questionnaire that has been used with some success in the field of rodent pest management explores the 'knowledge, attitudes and practices' or 'KAP' of a target community (see case studies under Further reading). The first group of questions in a typical KAP survey is designed to establish the basic socioeconomic profile of the respondent (sex, age, some basic wealth parameters). This is followed by questions that explore the respondent's knowledge of the scale and possible causes of rodent problems. Subsequent sections document the kinds of actions that are currently taken to combat these problems and the financial and other costs (actual and perceived) of these actions.

Finally, attitudes towards rodent problems and control measures are explored through questions that range from individual attitudes through to societal norms. Although all KAP surveys tend to follow a similar format, cropping systems are too diverse and cultural sensitivities too variable across the Asia–Pacific region to employ a standard questionnaire in all areas.

The information from KAP surveys can be used to assess various parameters—such as the severity of existing rodent problems, the perceived efficacy of current management actions, and the society's preparedness to try new kinds of actions. Because of the quantitative nature of the information, data from KAP surveys also can be used to compare the impact of rodent management actions on individual and societal attitudes—either by doing a 'before and after' comparison within treatment communities (where new rodent management practices are implemented) or comparing treatment communities with control communities (no change in practices).

KAP surveys usually aim to sample 100 or more respondents, with unbiased representation of males and females, and a good cross-section of 'wealth groups'. These ideals may not be possible in all societies. Wide consultation is needed before the design and implementation of a KAP survey and it is always advisable to do a 'pre-test' of a new questionnaire to make sure that the questions are appropriate in both subject matter and wording.

Pre-tests are also useful in determining how long the survey will take to conduct. Wherever possible, the survey questions and possible responses should be translated into the respondents' first language to reduce any potential for misinterpretation.

Some useful lessons already learned

Several recent rodent management studies in Southeast Asia have included an economic assessment of the various inputs (costs or investments) and outputs (benefits or outcomes), as well as a sociological assessment of the implementation of various methods by community members. These studies have produced some useful insights that you might wish to keep in mind throughout the development of a new project.

Key socioeconomic factors that affect adoption of new methods

Some of the key socioeconomic factors that are likely to influence the economic viability and sustainability of a particular rodent management strategy are listed in Table 10.1. This list is not exhaustive, but it might be a good starting point for consideration.

Examples of short-term and longer-term costs, benefits and constraints are given in Table 10.2. Short-term costs and benefits are relatively easy to quantify. Moreover, by assigning a monetary value to produce and labour, these factors usually can be expressed in terms of a common 'currency'. Similarly, the short-term constraints are usually easy to identify through many of the methods discussed above (e.g. seasonal calendars, wealth analysis).

The long-term costs and benefits of any action are far more difficult to assess. In part, this is due to the difficulty of predicting the long-term or large-scale impacts of rodent management. For example, it may be reasonable to suggest that a reduction in rodents within the fields and village environment would lead to a reduction in rodent-borne diseases such as leptospirosis, and to an increase in the health and fertility of livestock. However, it may be difficult to be assign a monetary value to improvements in human health and even more so for improvements in wider environmental health.

Another very important issue in the assessment of costs and benefits is the concept of **risk**. This in turn relates to the twin concepts of variability and

Table 10.2   A typical list of short and longer-term costs, constraints and benefits of rodent control. Not all of these factors are equally important in every situation and there may be other significant factors apart from those listed here.

	Short-term	Longer-term
Potential costs	• Financial cost of materials • Labour required for actions • Time invested in any associated social activity ('transaction' costs)	• Environmental costs (e.g. impact on non-target species)
Potential constraints	• Other demands on money • Other demands on time • Inability to coordinate actions	• Changing economic or political context • Inability to maintain necessary social structures
Potential benefits	• Increase in agricultural production • Improvement in the quality and value of harvested produce • Reduction in the postharvest loss of stored foods • Reduction in the level of contamination of stored foods • Value that can be assigned to captured rats	• Long-term benefits to human or livestock health (e.g. reduction in the impact of rodent-borne diseases) • Long-term benefits to environmental health (e.g. reduction in chemical use)

predictability. **Variability** is a normal element of all ecological systems, although both the scale or size and the degree of regularity of the changes differ greatly between systems. **Predictability**, the degree to which such variations can be forecast, can relate to either the scale or the regularity of the changes. For example, ecological changes associated with monsoonal flooding are probably quite predictable in terms of timing, but highly irregular in terms of severity. On the other hand, ecological changes that relate to wildfire activity might be predictable in terms of scale but much less predictable in terms of timing.

The economic importance of these concepts can be appreciated from an Australian rodent management example (see Box 10.2). In this case study, farmers can choose from a range of strategies that vary in their degree of associated risk.

The importance of community action and common property resources

In most situations, rodent management will be most effective if appropriate actions are taken over large areas and in a coordinated manner. Where this is not done, there is a real danger that any local impact on rodent numbers will be rapidly and literally overrun by dispersal of excess animals from any adjacent area where numbers remain high.

The application of rodent management over large areas is relatively straightforward in broadacre crop production systems where one farm owner or manager is not only responsible for deciding how and when to act but also has control over all the necessary equipment and budgets. However, across most of Southeast Asia and the Pacific, the situation is very different—the land is typically owned and managed by numerous smallholders and there may be various social and historical factors that make it difficult for people to work collectively towards effective rodent management.

Box 10.2 The economics of house mouse management in Australia

The introduced house mouse (*Mus musculus*) undergoes periodic outbreaks or 'plagues' in some wheat-growing regions of south-eastern Australia, with an average interval of seven years between these events. During plague years, mice cause huge crop losses and other damage to property. However, at other times, mouse numbers are low and they do little damage. Both the severity of plagues and the interval between them vary at any one location.

In normal years, the cost of mouse control outweighs any potential benefit in terms of damage reduction. During plague years, the reverse is always true, but the best benefit-to-cost ratios are achieved when mouse control is started early, before mouse numbers get too high. Farmers now have access to predictions based on probabilistic models, but there is a significant element of risk associated with following these predictions—the models give wrong results (either false alarms or failure to predict a plague) around 30% of the time.

Farmers who live in areas affected by mouse plagues have three main options:

- they can apply mouse control every year—this may well avoid all future plagues, but in six years out of seven they will be wasting their money
- they can apply mouse control only during the predicted plague years, but in the knowledge that these will be in error approximately 30% of the time
- they can reject mouse control altogether and hope that the losses during plague years are offset by the money saved by not applying any control.

The economic consequences of each risk management strategy can be calculated using an individual farmer's potential costs and losses.

The situation may be further complicated where the rodent management system either involves the use of a shared materials or equipment, or else depends on farmers making a contribution of money or labour to activities that will lead to shared benefits. Sociologists use the term **common property resource** in such cases where users share the 'rights' and 'benefits' of resource use, and also share the 'duties' of resource management.

One such system that has been tested in various socio-cultural contexts in Southeast Asia is the community trap–barrier system (CTBS; see Box 10.3). This system was designed and tested in lowland irrigated rice-growing systems in several Southeast Asian countries. Typically, in these regions, farmers own or manage landholdings of 1 ha or less. However, a CTBS unit set up within the boundaries of one farmer's field can be effective in reducing rat numbers and crop damage over a total surrounding area of around 10–15 ha. Hence, many families potentially share the benefits of a CTBS and might be reasonably expected to share in the material and labour costs of installing and maintaining the CTBS.

One study of the CTBS as a common property resource in the Mekong Delta region of Vietnam identified a range of sociological constraints and opportunities for sustainable application of the

CTBS. Foremost among these were the social relationships and associated systems of obligation among CTBS participants, and the nature of existing institutions that emphasise cooperation, such as integrated pest management (IPM) clubs.

The role of these social and institutional factors need to be considered in each new socio-cultural context and with a keen awareness of the wider political and economic environment, including the likelihood of change.

Box 10.3 The community trap–barrier system (CTBS)

The CTBS is a physical method of rodent control that was developed to control rat damage in lowland irrigated rice systems in Indonesia, Malaysia and Vietnam. The major pest species in these systems is the rice-field rat (*Rattus argentiventer*) which times its breeding activity in these systems to match the growth and maturation stages of the rice crop. The efficiency of the CTBS system is currently being evaluated in Indonesia, Vietnam, Philippines and Cambodia, with good results to date.

The CTBS consists of a square or rectangular barrier system (typically with each side measuring 50–100 m) that encloses a lure crop (typically, a rice crop planted 2–3 weeks ahead of the surrounding cropping area). Rats are attracted to the early maturing lure crop and are captured in multiple-capture traps placed at entry points along the barrier. By drawing adult rats out of the local population before they start breeding, the rate of

population increase of the remaining population is lowered, thereby avoiding the high rat densities typical of unmanaged fields. Empirical studies of crop damage around CTBS units suggest that each unit may be effective in protecting a surrounding area of 10–15 ha. In a large, uniform cropping area, CTBS units ideally would be positioned in a way that achieves overlap between the individual 'halos of protection' of each unit.

CTBS units require regular maintenance to ensure that the fence and traps are not compromised. Ideally, the cost of materials and the tasks of constructing and managing each CTBS unit are shared by all of the people who derive benefit from the unit. In some parts of Southeast Asia, the commercial value of captured rats provides added incentive for daily checking and regular maintenance of the CTBS.

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CHAPTER 11

Review of the major pest species

Introduction

All of the major pest rodent species found in South and Southeast Asia belong to only a handful of genera, among which *Rattus*, *Bandicota* and *Mus*, all members of the family Muridae, are pre-eminent. Other minor pests are found within the murid genera *Berylmys* and *Millardia*, and among members of the family Rhizomyidae, the bamboo rats. Various squirrel species are also regarded as agricultural pests, especially in South Asia, but these are not covered here. A list of the species documented and their general geographical distribution is given in Table 11.1.

The information summarised in these accounts draws upon a combination of published data and the authors' combined field experience in many localities between Bangladesh and Papua New Guinea. As a rule, more detail is available for those species that cause the greatest overall damage. For each species, a few key references only are given. The two species of *Millardia* recorded from Bangladesh (*M. meltada*) and Myanmar (*M. kathleenae*) are omitted from this account because of a lack of information on their habits or pest status in these areas. *Millardia meltada* is a significant pest in parts of India, although mainly in semiarid to arid regions.

The measurements given for each species usually represent the adult range, as measured on fresh specimens. In some cases, only a maximum value is available (e.g. to 35 g), or a mean (sometimes with standard deviation). Individual adult specimens may well be slightly bigger or smaller than the values given. Unless otherwise stated, information on litter size is based on counts of live embryos.

The distribution maps are somewhat generalised. While a species is unlikely to occur far outside of the indicated range, it may not be present at all localities within the range.

Using a taxonomic key

The most common type of key used in taxonomy is a **dichotomous key** in which the user works systematically through a series of predefined steps to achieve a reliable identification. At each step, the user is prompted to choose between contrasting pairs of characters or 'couplets' (e.g. 'pes black' or 'pes white') that serve to progressively narrow down the range of possibilities. In general, the complexity and usefulness of any key will depend on the number of species that it attempts to cover.

Dichotomous keys work best where the character states are discrete (e.g. 'black' versus 'white', rather than 'mostly black' versus 'mostly white') and where there is little or no variation within species. Naturally, for many groups of rodents, this type of key does not work very well. However, their performance can be improved by allowing highly variable species such as *Rattus rattus* to appear multiple times within the key and by allowing some couplets to contain two or more different characters (thereby providing a check on each individual character).

A dichotomous key to the major rodent pests of Southeast Asia and the Pacific is given at the end of this chapter. When using this key, you must keep in mind that it does not cover all of the rodents that occur through this diverse region and will give a spurious identification for any species not included within the key. Accordingly, it is strongly recommended you check any species identification obtained using this key carefully against the descriptive, distributional and ecological information provided for each of the pest species in this chapter.

The terminology and full explanations of the characters used in the dichotomous key are provided in Chapter 4. We recommend that you read that chapter before using the key and species accounts.

Table 11.1   Distribution of pest rodent species by country or region.

Species	Bangladesh	Southern China	Myanmar	Thailand	Cambodia	Laos	Vietnam	Malay Peninsula	Sumatra, Java, Borneo	Nusa Tenggara Fl = Flores Su = Sumba	Sulawesi	Philippines Mi = Mindanao Pa = Palawan	New Guinea	Island Pacific	Distribution map
Bandicota bengalensis	+	−	+	−	−	−	−	*	S*	−	−	−	−	−	124
Bandicota indica	+	+	+	+	+	+	+	*	S*J*	−	−	−	−	−	128
Bandicota savilei	−	−	+	+	+	?	+	−	−	−	−	−	−	−	132
Berylmys berdmorei	−	−	+	+	+	+	+	−	−	−	−	−	−	−	134
Berylmys bowersi	−	+	+	+	?	+	+	+	S	−	−	−	−	−	134
Cannomys badius	+	+	+	+	+	+	−	−	−	−	−	−	−	−	137
Mus booduga	?	−	+	−	−	−	−	−	−	−	−	−	−	−	139
Mus caroli	−	+	+	+	+	+	+	*	SJ	Fl	−	−	−	−	142
Mus cervicolor	?	−	+	+	+	+	+	−	−	−	−	−	−	−	144
Mus cookii	−	+	+	+	?	+	+	−	−	−	−	−	−	−	146
Mus musculus Group	+	+	+	+	+	+	+	+	+	+	+	+	+	+	148
Mus terricolor	+	−	−	−	−	−	−	−	S*	−	−	−	−	−	139
Nesokia indica	+	−	−	−	−	−	−	−	−	−	−	−	−	−	150
Rattus argentiventer	−	−	−	+	+	?	+	+	+	Su	+	Mi	*	−	152
Rattus exulans	+	−	+	+	+	+	+	+	+	+	+	+	+	+	157
Rattus losea	−	+	−	+	+	+	+	+	−	−	−	−	−	−	160
Rattus mordax	−	−	−	−	−	−	−	−	−	−	−	−	+	−	163
Rattus nitidus	−	+	+	+	?	+	+	−	−	−	+	−	*	−	166
Rattus norvegicus	+	+	+	+	+	+	+	+	+	+	+	+	+	+	169
Rattus praetor	−	−	−	−	−	−	−	−	−	−	−	−	+	*	163
Rattus rattus Complex (multiple species)	+	+	+	+	+	+	+	+	+	+	+	+	+	+	172
Rattus sikkimensis	+	+	+	+	+	+	+	−	−	−	−	−	−	−	177
Rattus steini	−	−	−	−	−	−	−	−	−	−	−	−	+	−	163
Rattus tiomanicus	−	−	−	−	−	−	+	+	−	−	−	Pa	−	−	179
Rattus turkestanicus	−	+	+	−	−	−	−	−	−	−	−	−	−	−	181
Rhizomys pruinosus	−	+	+	+	+	+	+	+	−	−	−	−	−	−	184
Rhizomys sinensis	−	+	+	−	−	?	+	−	−	−	−	−	−	−	184
Rhizomys sumatrensis	−	−	+	+	+	+	+	+	S	−	−	−	−	−	184

Note: ? = possibly found in this region, but yet to be confirmed; * = restricted to small areas within this region.

Bandicota bengalensis

(Gray and Hardwicke, 1804)

COMMON NAMES: lesser bandicoot, Bengal bandicoot, Indian 'mole rat'

Bandicota bengalensis is a major agricultural and urban pest across much of South Asia, east to central Myanmar. It occupies communal burrows and can reach very high local population densities. The habitual hoarding of large quantities of cereal grain in subterranean caches is also noteworthy.

MORPHOLOGICAL FEATURES: a medium-size rat with a stocky build and distinctively blunt, slightly upturned snout. The general dorsal fur colour is a pale grey–brown, grizzled with black. The belly fur is pale grey, tipped with cream or buff. The tail is usually 20–30 mm shorter than head+body and is uniformly dark above and below. The pes is clothed in dark hairs and bears long, straight claws. The plantar pads are small and low. The incisors are broad, with orange or cream enamel, and the upper pair projects slightly forwards.

MAMMAE: highly variable, usually 7–9 pairs in Bangladesh but as many as 14–17 pairs in India.

Bandicota bengalensis is distinguished from the similarly sized *Bandicota savilei* by its shorter, upturned snout, its paler forward-projecting incisors, its larger number of teats, and its shorter, broader pes.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Gunomys bengalensis*, *Bandicota varius*.

DISTRIBUTION: South Asia, including Sri Lanka, north to Nepal and Bhutan, and east to central Myanmar where it is sympatric with *B. indica* and *B. savilei*. Introduced populations are found on Penang Island, off the west coast of the Malay Peninsula, in the Aceh region of Sumatra and in East Java, Indonesia.

TAXONOMIC ISSUES: there is some variation in chromosome morphology, molar size and fur texture across the geographical range of *B. bengalensis*. Specimens from Penang Island were described as *Bandicota varius*; they are slightly larger than typical *B. bengalensis*.

HABITAT USE: *B. bengalensis* is common in both villages and towns, and in associated cropping areas. It is usually most abundant in higher rainfall areas, and less so in arid regions of Bangladesh where

Adult measurements	Bangladesh	Myanmar
Weight (g)	to 310	to 400
Head+body (mm)	75–254	195–228
Tail length (mm)	44–177	139–188
Pes length (mm)	19–41	33–43
Ear length (mm)	–	21–24

Nesokia indica appears to occupy the same ecological niche. In Pakistan, *B. bengalensis* is recorded from intertidal mangrove forest. There are few estimates of population density, but one study in Andra Pradesh, India, gave figures of 0.5–2.4 individuals/ha. Very high populations of almost 1 individual/m² have been recorded in urban grain stores ('godowns') in India.

This species takes a wide range of food items, including molluscs and crabs. However, in feeding trials, it prefers cereal grain over animal foods, and rice over wheat. Stomach contents of wild-caught adults weighed an average of 24.2 g. Daily consumption in captivity ranges from 4–8% of body weight. In a rice-growing area of Pakistan, *B. bengalensis* consumed all stages of the rice plants, including young tillers, the flowers and the ripening grain. Some insects were also consumed.

Bandicota bengalensis is a good swimmer and causes significant damage to deep-water rice crops where it is reported to occupy platforms constructed from cut tillers. However, in most habitats it constructs and occupies elaborate burrow systems. These burrows are occupied for considerable periods and individuals generally only construct new burrows to avoid flooding or when local food resources are depleted. Males relocate their burrows more frequently than do females.

Studies of *B. bengalensis* movement found that most feeding activity was confined to small areas of 12–40 m² immediately around a burrow complex. However, individuals with their burrow located in village habitat generally moved greater distances (of 130 m or more) to visit preferred feeding areas in the fields. Most movements occurred at times of little or no moonlight.

NESTING BEHAVIOUR: burrows are constructed in field bunds, in vegetable gardens and orchards, and in the floors and walls of buildings. In Bangladesh, mudbrick houses or stores infested by *B. bengalensis* are liable to suffer serious structural damage, to the point of collapse.

Individual burrow systems are often very complex, with multiple chambers and entrances (often as many as 12–16 per burrow). The average length of burrows in India is around 5.5 m, with the largest measured being 45 m in total length. Most burrow entrances are sealed during the day but their location is usually obvious from the piles of excavated soil. However, other entrances may be kept clear of soil and used as escape holes. Burrow systems may be used over several generations.

Burrows are usually occupied by one adult male or female, or by a female with her young. However, multiple occupancy is reported in areas of high population density. Breeding chambers are lined with straw and are often accompanied by caches of wheat or rice panicles. Only females seem to cache food but the quantities can be significant. Burrows excavated after harvest in India contained an average of 3.7 kg of stored wheat.

BREEDING BIOLOGY: breeding activity in rural populations is seasonal, with peak activity coinciding with crop maturation. In urban grain stores in India, breeding occurs year-round but with a peak in the dry season. Pregnancy rates in these contexts peak at more than 70% but fall to 13% during the middle of the wet season.

The oestrus cycle is 3–5 days and the gestation period is 21–25 days. The young weigh 3.5–5.0 g at birth. The eyes open on day 14–18 and weaning commences around day 25–28. Sexual activity commences as early as three months of age among females and slightly later among males. In an urban Indian population, sperm production began around 90–170 days after birth. In a rural, rice-growing area of Pakistan, females showed vaginal perforation at body weights of 40–79 g, and the smallest pregnant female was 89 g. Males in this population developed scrotal testes at body weights of 70–159 g, depending on the season.

Reports of litter size from urban contexts in South Asia range from 1–19, with means of 6.2 in the Punjab, 8 in Bombay, and 7.4 in Yangon. The average interval between pregnancies in these populations ranges from 30–35 days in Calcutta to 62 days in

Yangon. The average life span in a Calcutta godown is around 200 days and only 3% of individuals live for longer than one year. In a study in rural Pakistan, the average litter size varied seasonally from 6.7 to 10.2. The pregnancy rate in this population fell to 2–15% during periods of food shortage but rose to a maximum of 44% around rice harvest time. The estimated number of young produced per female in this population is 28.2 each year. In a nearby, sugarcane-growing area, mean litter size was never higher than 6.5 in any month, but due to more continuous breeding activity, the annual productivity was higher, at 43.6 young per female per year. Five pregnant females collected in central Myanmar in March 2003 had 6–9 embryos (mean of 8.0).

POPULATION DYNAMICS: populations in rural habitats in India show fluctuations that are directly related to cropping cycles. In West Bengal, where a single wet-season rice crop is grown, *B. bengalensis* shows a single peak in abundance around harvest. In Mysore, with two rice crops, the rats show two matching peaks in abundance.

DAMAGE TO CROPS: *B. bengalensis* damages all kinds of field crops and also attacks stored grain. In one study, damage to wheat crops in Bangladesh was confined to within a 6–8 m radius of the centre of the burrow system. Yield loss in this system was estimated at 2.5–12%.

Losses due to consumption and hoarding were estimated at 261 kg/ha for wet-season rice crops in West Bengal, India. Yield loss information from wheat-growing areas in Madhya Pradesh, India, put the quantity of grain lost to caches alone at 261–388 kg/ha. These figures equate to around a 10–15% total loss in production.

Adult *Bandicota bengalensis* from Bangladesh.

Pes of adult from Bangladesh; upper surface (left) and lower surface (right).

Adult from central Myanmar.

Adult from Bangladesh, showing forward-projecting incisors.

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Adult female *Bandicota bengalensis* from Myanmar, showing large number of teats.

Portion of tail; adult from Bangladesh.

A pup from Bangladesh: the eyes are about to open.

Bandicota indica (Bechstein, 1800)

COMMON NAME: giant bandicoot

Bandicota indica is known throughout South and Southeast Asia for its great size and ferocity when captured, and for its large burrow systems that cause damage to buildings, dams and roadways. It is found in both field and village to urban habitats, and appears to be especially common around permanent water sources. In most areas, population densities of *B. indica* are quite low and it can be regarded as a minor pest. However, in parts of South Asia, it reaches much higher densities and is responsible for significant damage to field crops (grains and vegetables), to poultry, and to stored foodstuffs.

MORPHOLOGICAL FEATURES: the largest murine rodent in South or Southeast Asia, with adults often weighing 500–1000 g. The dorsal fur is distinctly shaggy and blackish-brown on the back and flanks, sometimes with a reddish tinge. Numerous black guard hairs project through the dorsal fur, especially down the middle of the back and on the rump. The belly fur is usually dark grey, sometimes with a cream or pale-buff wash. The tail is usually shorter but sometimes slightly longer than the head+body and is uniformly dark. The manus and pes are clothed in black hairs and bear strong claws adapted for digging. The pes is broader and heavier in *B. indica* than in the other *Bandicota* species. The ears are large and sparsely furred in Southeast Asian populations but distinctly smaller and better furred in animals from Bangladesh. Juvenile *B. indica* are readily distinguished from all other murids by their proportionally very large feet and their extremely broad incisors.

Newly captured subadults and adults are highly vocal, making a peculiar noise somewhere between hissing and braying. At the same time, the guard hairs are erected to produce a threatening appearance.

MAMMAE: usually 1+2+3; individuals with 1+2+7 teats are reported from Bombay.

Adult measurements	Bangladesh	Thailand	Cambodia	Southern Vietnam	Northern Vietnam
Weight (g)	to 760	to 545	to 830	to 870	to 830
Head+body (mm)	150–309	to 276	175–285	160–300	to 285
Tail length (mm)	115–267	to 244	140–270	135–248	to 270
Pes length (mm)	42–60	to 56	40.5–54	39–58.5	to 54
Ear length (mm)	–	to 30	24–29	19–29	to 35

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:
Bandicota gigantea, Bandicota maxima, Bandicota indica nemorivaga, Bandicota indica setifera.

DISTRIBUTION: widely distributed across South (including Sri Lanka) and Southeast Asia, through to southern China and Taiwan. *Bandicota indica* is generally absent from peninsular and insular Southeast Asia, except where it has been introduced during prehistoric or historic times—to the Kedah and Perlis regions of the Malay Peninsula and the island of Java in Indonesia. The population on Taiwan may also be a recent introduction.

TAXONOMIC ISSUES: populations of *B. indica* vary in both chromosome number and morphology across its extensive geographical range and this has led several taxonomists to suggest that two or more species are present. Our preliminary genetic studies also point to high levels of divergence between populations within Southeast Asia (e.g. lowland Cambodia and Vietnam versus upland Laos). A full-scale taxonomic revision of the *B. indica* group is clearly needed.

HABITAT USE: found in all parts of the human landscape, including varied cropping systems and village to urban environments. *Bandicota indica* is also reported from uncultivated marshy areas and from patches of forest, although these are never far from human populations. In villages or towns, it is usually found close to ponds or riverside habitats.

In Southeast Asia, it is present in both lowland and upland environments.

Bandicota indica is an excellent swimmer and it has been observed diving to retrieve prey items from bottom sediment. This allows it to exploit a wide range of both aquatic and terrestrial foods, including molluscs, crustaceans, water lily fruit, water hyacinths, insects, earthworms, and field crops such as rice, vegetables (including tubers), fruits, and nuts. Attacks on nestling birds and snakes are also reported.

Burrow systems range from short tunnels (to 72 cm) used as feeding retreats, through to elaborate and extensive complexes with multiple chambers and entrances (the largest covering an area of 300 m²). Burrow entrances are usually left open and are sometimes marked by piles of faeces or food refuse; however, they are sometimes concealed and can even open below water level. Large burrow complexes sometimes contain numerous adults along with their young. Grain hoarding behaviour is reported from localities in India, but it does not seem to be habitual, as it is in *Bandicota bengalensis*. The few studies of movement suggest that *B. indica* individuals usually move only short distances from their burrow systems. However, one study reported nightly movements of around 250 m between a daytime retreat in a village and a feeding area within crops.

There are few estimates of population density, but one study in India recorded an average of 456

individuals per ha in rice fields, with an average of 38 active burrows per ha. This high number presumably included many juveniles. In lowland irrigated systems in Southeast Asia, *B. indica* usually makes up less than 5% of captures either from live-trapping on grids or along transects, or from trap–barrier systems (TBSs). In rainfed lowland rice systems in Binh Thuan Province, Vietnam, the capture rate for *Bandicota* spp. (*B. indica* and *B. savilei*, not distinguished) in TBSs (with trap crops) is much higher—often more than 50% during the early part of the growing season, but falling to around 20–30% by harvest time.

NESTING BEHAVIOUR: pups are born in a 'brood chamber' constructed within the general burrow complex. These chambers are lined with leaves or other soft material, such as paper or cloth. More than one female can bear their young within a single burrow system; indeed, one report from Bangladesh notes eight separate litters within a single, interconnected burrow system.

BREEDING BIOLOGY: a two-year study of breeding activity in natural marshland habitat on Sagor Island, off the coast of India, found breeding in all months of the year. The overall adult pregnancy rate was 27%, but this peaked at 50% in October–April and fell to 10% in May–September. Mean litter size did not vary between months.

Litter size is variously reported as 1–8 (mean of 4.8), 1–4, and 'up to 10' for Indian populations, and 2–12

for northern Vietnam. A sample collected in the southern Vietnamese province of Binh Thuan in March 2001 gave live embryo counts of 4–8 with a mean of 5.8.

Wild-caught Indian animals have an oestrus cycle of 4–8 days and a gestation period of 23 ± 1.2 days. Vaginal perforation occurs at 190–210 days after birth, at body weights of 287–345 g. The largest imperforate female in the Binh Thuan Province sample weighed 520 g, while the smallest perforate female weighed only 145 g.

POPULATION DYNAMICS: nothing is reported. Capture rates in the Mekong Delta of Vietnam and West Java, Indonesia, are consistently low at all times of the year.

DAMAGE TO CROPS: at low population densities, *B. indica* may feed primarily on invertebrates and cause little damage to crops. Indeed, the benefits of its regular predation on molluscs and crabs may outweigh any crop damage. However, at high densities, it is reported to cause heavy damage, both in rice crops (individuals cutting 1–4 m² of tillers per night) and in potato (damaging 1–3 kg per night) and peanut fields.

Mean daily food intake over long periods in captivity is 95 g of mollusc flesh or 35 g of paddy. This tallies well with the stomach content yield for wild-caught Indian adults of 23–58 g.

Adult *Bandicota indica* from the uplands of Laos.

Adults from Bangladesh (top) and Cambodia (bottom).

Juvenile from the uplands of Laos.

Incisors of adult from the uplands of Laos.

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Tien, D.V. and Cu, H.T. 1964. Données éécologiques sur le bandicote forestier (*Bandicota indica nemorivaga* (Hodgson, 1836 Muridae)). Zietschrift für Säugetierkunde, 30, 185–189.

Bandicota indica: tail, adult from Vietnam.

Upper surface of manus; adult from Bangladesh.

Pes of an adult from southern Vietnam: upper surface (left) and lower surface (right).

Ear of an adult from Bangladesh.

Bandicota savilei (Thomas, 1916)

COMMON NAME: lesser bandicoot

This species takes the place of *Bandicota bengalensis* in Southeast Asia. It is apparently restricted to lowland areas and is often found together with *Bandicota indica*. *Bandicota savilei* can be locally abundant and is presumably a significant pest species, especially in areas of rainfed rice crops that do not experience any major flooding.

MORPHOLOGICAL FEATURES: a medium-size, terrestrial rat with shaggy, reddish-brown fur on the back and sides, and buff-tipped, grey-based belly fur. The fur is spiny and contains numerous long, black guard hairs that are most conspicuous on the lower back. The tail is usually 20–30 mm shorter than the head+body and is uniformly dark above and below. Rarely, the tail has a short, all-white tip. The ears are relatively large and well furred. The pes of adult *B. savilei* is narrow and 'gracile' compared with similar-size (immature) *B. indica*. As in the other *Bandicota*

species, the incisors are broad (>3.5 mm in combined width), the claws on the manus and pes are long and relatively straight, and the plantar pads are small and low compared with all species of *Rattus*.

MAMMAE: 1+2+3.

Bandicota savilei was recently found living alongside *B. bengalensis* near Yezin in central Myanmar. The two species are of similar size and colouration, but *B. bengalensis* is readily distinguished by its more forward-projecting upper incisors, smaller ears, shorter and broader pes, shorter tail and more numerous teats in females. Indeed, *B. savilei* is more similar to *B. indica* in most regards, but can be distinguished by its smaller, narrower feet as noted above, and by its overall smaller size.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Bandicota bengalensis, Bandicota bengalensis hichensis, Bandicota bengalensis giaraiensis, Bandicota banchakensis, Bandicota varius, Bandicota indica savilei.*

DISTRIBUTION: lowlands of central Myanmar, Thailand and Vietnam; probably present in lowland areas of Laos, within the valley of the Mekong River.

Adult measurements	Myanmar	Thailand	Southern Vietnam
Weight (g)	233–318	to 199	to 292
Head+body (mm)	208–227	145–225	102–228
Tail length (mm)	170–195	75–178	90–185
Pes length (mm)	35–41	33–40	26–40
Ear length (mm)	23–26	20–30	19–26

TAXONOMIC ISSUES: this species has been frequently confused with *B. bengalensis* or treated as a subspecies of *B. indica*. It is morphologically and genetically distinct from both, and occurs together with *B. indica* in many areas. In March 2003, we encountered all three *Bandicota* species living together in a single field complex near Yezin in central Myanmar. The fields held a standing crop of the pulse known as 'black gram', grown as an intercrop for rainfed rice.

HABITAT USE: *B. savilei* is abundant in rainfed rice cropping systems in Binh Thuan Province, Vietnam, where it occurs together with *Rattus argentiventer* and *B. indica*. It appears to be absent from those parts of the Mekong Delta in Vietnam that experience widespread flooding, and may thus be relatively intolerant of prolonged inundation. If so, this would be a point of distinction with the other species of *Bandicota*, both of which might be characterised as semi-aquatic in habits.

An apparently 'natural' population of *B. savilei* is reported from Thailand, living in "grass beneath teak forest".

NESTING BEHAVIOUR: burrows are constructed in large bunds and small 'upland' areas. One report from Thailand mentions "runways through grass" leading to a burrow "only about 18 inches [46 cm] deep".

BREEDING BIOLOGY: little known. In Binh Thuan Province, a high proportion of adult females (48%, $n = 27$) were breeding in March 2001. This was near the end of the dry season, after a prolonged fallow period and with little or no standing crop of any kind. The smallest pregnant females weighed only 75 g, but most pregnancies were in females above 160 g. Embryo counts were 3–10 with a mean of 5.7. Nearly two-thirds of the pregnant females also had uterine scars indicative of one previous litter. A sample of pregnant females collected in central Myanmar in March 2003 had 5–11 embryos.

POPULATION DYNAMICS: nothing known.

DAMAGE TO CROPS: damage to maize is specifically mentioned for Thailand. In Myanmar, ripening seed pods of the pulse 'black gram' were found scattered around conspicuous burrow entrances in low bunds, and fragments of the pulse were observed in the stomach of several individuals of *B. savilei*.

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Musser, G.G. and Brothers, E.M. 1984. Identification of bandicoot rats from Thailand (*Bandicota*, Muridae, Rodentia). American Museum Novitates, No. 3110, 1–56.

Adult *Bandicota savilei* from southern Vietnam.

Adult from Cambodia.

Adult from southern Vietnam.

Lower surface of pes; adult from southern Vietnam.

Berylmys berdmorei (Blyth, 1851)
Berylmys bowersi (Anderson, 1879)

COMMON NAME: white-toothed rats

Berylmys berdmorei

Berylmys bowersi

The genus *Berylmys* contains four or more species, all of which have their primary populations in forest habitat. However, at least two species of this group appear to warrant inclusion as occasional agricultural pests. In northern Laos, *B. berdmorei* constructs its burrow systems within the upland agricultural landscape and the species is identified by farmers as a minor pest. In the same area, *B. bowersi* is said to be confined to forest habitat. However, elsewhere in Laos and across wider Southeast Asia, *B. bowersi* has been trapped within cropping areas and is presumably responsible for some crop damage.

MORPHOLOGICAL FEATURES: species of *Berylmys* can be recognised by their pale-cream or white incisor enamel and their short, crisp-grey or brownish-grey dorsal fur, which is sharply demarcated from a pure-white belly. The ears are moderately large and thinly furred. More specifically,

for each of the two pest species:
- *B. berdmorei* has steel-grey dorsal fur, the tail is usually 30–40 mm shorter than the head+body, and the feet are clothed in pure-white or grey hairs
- *B. bowersi* has brownish-grey to dull-tan dorsal fur, and the tail is usually slightly longer than the head+body, is usually slightly darker above than below, and is either plain to the tip or ends in an all-white section.

MAMMAE: 1+2+2 in *B. berdmorei*; 1+1+2 in *B. bowersi*.

Adult measurements	B. berdmorei	B. berdmorei	B. bowersi	B. bowersi
	Thailand	Laos	Thailand	Vietnam
Weight (g)	to 235	118–205	to 420	to 292
Head+body (mm)	to 255	175–207	to 245	240–285
Tail length (mm)	to 192	134–170	to 256	260–310
Pes length (mm)	to 46	36–39	to 55	54–57
Ear length (mm)	to 29	24–26	to 30	30–37

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *B. berdmorei = Rattus berdmorei*; *B. bowersi = Rattus bowersi*.

DISTRIBUTION: *B. berdmorei* is recorded from scattered localities in southern Myanmar, northern and south-eastern Thailand, Cambodia (Kampot), northern Laos and southern Vietnam (including Con Son Island). It was recently collected in Luang Prabang Province, northern Laos. *Berylmys bowersi* is known from many more localities, from north-eastern India to southern China (to Fukien Province) and ranging through northern Thailand, Laos and Vietnam. It is also found on the Malay Peninsula and on Sumatra, Indonesia.

TAXONOMIC ISSUES: both species show considerable variation across their ranges and may include more than one species.

HABITAT USE: both species are most commonly encountered in upland regions—the main exception being the record of *B. bowersi* from Con Can Island off southern Vietnam. Both are essentially terrestrial and live in burrows, although the longer-tailed *B. bowersi* is a capable climber.

In Luang Prabang Province, Laos, we recently found *B. berdmorei* to be moderately abundant in valley floor and slope habitats, with burrows located both in small bamboo thickets and in a cleared field planted with cassava and sweet potato. Individuals were trapped in these areas but also in an irrigated rice-field complex, suggesting nightly movements of several hundreds of metres.

Berylmys bowersi in Laos appears to be more strictly forest dwelling, although it is occasionally trapped in cropping areas. It is said to be an occasional pest in gardens and orchards in Malaysia. Ecological studies of this species in Malaysian forests suggest a diet of fruit and vegetable matter, with occasional insects and molluscs. Although a capable climber, *B. bowersi* spends most of its time foraging on the ground.

NESTING BEHAVIOUR: burrows of *B. berdmorei* in Luang Prabang Province were several metres in length with two entrances and one central chamber. In three of four cases, the main burrow entrances were concealed beneath leaf litter and lacked any associated mound of soil. However, in each case, an obvious entrance with a conspicuous mound of soil was located several metres away; these all led into short, blind tunnels. Whether these represent aborted tunnels, feeding retreats or decoy tunnels is not known, but the difference in visibility might favour the latter interpretation.

Two of the three excavated *B. berdmorei* burrows contained a large bundle of freshly cut leaf material in the central chamber. No pups were found, but one burrow contained two adults (a female and a probable male that escaped), and two subadults. The other burrows contained solitary, young males.

In Malaysia, nests of *B. bowersi* were located in fallen logs and in burrows situated in drier, more elevated sites.

BREEDING BIOLOGY: in Malaysia, female *B. bowersi* have been found with 2–5 live embryos. Maturation of both sexes is very slow in this population, with males developing scrotal testes and showing epipidymal sperm at body weights between 150–300 g. The smallest pregnant female weighed 290 g.

One adult female of *B. berdmorei* (body weight 203 g) collected in Luang Prabang showed 14 placental scars, apparently belonging to two sets. Six other females between 118–182 g all showed perforate vaginas but none had commenced breeding.

POPULATION DYNAMICS: nothing recorded.

DAMAGE TO CROPS: Lao farmers in Luang Prabang Province claim that *B. berdmorei* damages all crops but especially tubers such as sweet potato. One burrow was indeed located in a sweet potato field and the farmers were able to produce tubers that had been eaten out from below, presumably by *B. berdmorei*. The same farmers gave a credible description of *B. bowersi* but said that it stayed within the forest, where it burrowed but ate fallen fruits.

Nothing specific is reported regarding the damage attributed to *B. bowersi* in Malaysia.

KEY REFERENCES:

Harrison, J.L. 1955. Data on the reproduction of some Malayan mammals. Proceedings of the Zoological Society, London, 125, 445–460.

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Manus of adult *Berylmys bowersi* from Thailand: upper (left) and lower (right) surfaces.

Pes of adult *B. bowersi* from Thailand: upper (left) and lower (right) surfaces.

Tail of adult *B. bowersi* from Thailand.

Adult *Berylmys berdmorei* from the uplands of Laos.

Adult *B. berdmorei* from the uplands of Laos.

Incisors; adult *B. berdmorei* from the uplands of Laos.

Lower surface of pes; adult *B. berdmorei* from the uplands of Laos.

Cannomys badius (Hodgson, 1841)

COMMON NAME: lesser bamboo rat

Cannomys badius is widespread across the upland regions of Tibet and north-eastern India to southern China and Cambodia. It is a moderately large, stockily built animal with lush orange–brown fur and obvious adaptations to fossorial life. Its deep and extensive burrow systems are often located in slash-and-burn gardens and it inflicts some damage on upland rice crops. There are some also reports of bamboo rats damaging sugarcane, cassava and orchard trees. Farmers excavate many *Cannomys* burrows for their tasty contents and they are often traded along roadsides.

MORPHOLOGICAL FEATURES: in common with the other bamboo rats, *Cannomys badius* has a massively broadened head, a plump body with short limbs, strong claws on both pes and manus, and a short, sparsely haired tail which lacks scales (it is instead covered in soft, wrinkled skin). Other diagnostic features include massive incisors, and small eyes and ears.

Cannomys badius is the smallest bamboo rat and it is easily distinguished from the species of *Rhizomys* by its reddish-brown fur colour, extremely small ears (less than 10 mm) that are hidden in the fur, and smooth rather than granulated plantar pads on the manus and pes.

MAMMAE: 1+1+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rhizomys badius, Cannomys badius castaneus.*

DISTRIBUTION: *C. badius* ranges through the uplands of Nepal, eastern Bangladesh and India, Myanmar, Thailand, Laos and Cambodia.

TAXONOMIC ISSUES: subspecies of *C. badius* are sometimes recognised but their validity is untested. The bamboo rats as a whole are in urgent need of revision, especially on account of the heavy exploitation of these species in some areas as commercial food items.

HABITAT USE: the bamboo rats are probably most abundant in the natural bamboo forests that still cover large areas of the uplands of Southeast Asia. Their presence in such areas is always obvious from their large, poorly concealed burrow systems

Adult measurements	Thailand	Vietnam
Weight	0.5–0.8 kg	–
Head+body (mm)	147–265	191–259
Tail length (mm)	60–75	43–73
Pes length (mm)	30–35	30–35
Ear length (mm)	7–10	8–11

in which they shelter through the day. In northern Laos, burrows of *C. badius* are often located within or around the margins of swidden gardens. Local farmers claim that these burrows are present at the time when the garden site is prepared from forest or fallow regrowth, but the concentration of burrows makes it seem likely that some are newly dug, at least around the garden edges. Lao farmers also claim that *C. badius* tunnels below rice plants and consumes the plants from below. The remains of plants destroyed in this way have been observed in upland fields in Luang Prabang Province.

NESTING BEHAVIOUR: in north-eastern India, the burrows of *C. badius* consist of a single tunnel running at a depth of 60 cm or so below the ground and ending in a large chamber. A second tunnel usually runs partway to the surface; perhaps as an incomplete emergency exit. When the burrow is occupied, the active entrance is closed with freshly piled earth.

BREEDING BIOLOGY: litter size is reported as 3–5 for Thailand.

POPULATION DYNAMICS: nothing known.

KEY REFERENCE:
Marshall, J.T. 1977. Family Muridae. In: Lekagul, B. and McNeely, J.A., ed., Mammals of Thailand. Bangkok, Kurusapha Press, 397–487.

Adult *Cannomys badius* from the uplands of Laos. The white patch on the forehead is absent in some individuals.

Pes of an adult from Laos: upper surface (left) and lower surface (right).

Mus booduga (Gray, 1837)
Mus terricolor (Blyth, 1851)

COMMON NAME: pygmy mice

Mus booduga

Mus terricolor

These are small-bodied mice that sometimes occur together in the agricultural landscape of South Asia. Unfortunately, the two species were only recently confirmed as separate species, hence much of the ecological literature is probably based on mixed observations of the two.

MORPHOLOGICAL FEATURES: both species are small mice with soft fur, pure-white manus and pes, and tails that are distinctly darker above than below. *Mus booduga* has bright, yellowish-brown dorsal fur and a pure-white belly. The tail is usually around 10 mm shorter than the head+body. *Mus terricolor* is even smaller than *M. booduga* and has dull, brownish-grey dorsal fur and belly fur that is light grey with white tipping. The fur on the cheek and lower part of the snout is pale. The tail is usually slightly shorter than the head+body. The pes of *M. terricolor* shows two distinctive features that suggest

a strong digger with poor climbing ability—low plantar pads and forward-projecting claws, similar to those of *Bandicota* spp. The inner and outer metatarsal tubercles are positioned close together, unlike the condition in *M. musculus*. The ears of *M. terricolor* are distinctly smaller than those of sympatric *M. musculus*.

MAMMAE: 1+2+2 in both species.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:
(of *Mus terricolor*) *Mus booduga, Mus dunni.*

Adult measurements	M. booduga, India	M. booduga, Myanmar	M. terricolor, India	M. terricolor, Bangladesh
Weight (g)	to 14	16–17.5	to 11	7–11
Head+body (mm)	to 80	85–93	to 70	66–68
Tail length (mm)	to 70	55–66	to 70	60–62
Pes length (mm)	to 17	15–17	to 16	14–15
Ear length (mm)	to 12.5	16–17.5	to 11.5	–

DISTRIBUTION: not yet fully documented. *Mus booduga* is found in Sri Lanka and peninsular India north to Jammu and Kashmir, and southern Nepal. There is an isolated population in central Myanmar. *Mus terricolor* is widespread on peninsular India, west to Pakistan and north to Nepal. We have collected this species in eastern Bangladesh. An isolated population of *M. terricolor* in northern Sumatra, Indonesia, is probably due to human introduction.

TAXONOMIC ISSUES: the distinction between the two Indian species of pygmy mice was confirmed in 1968 by studies of their chromosomes. There has been no recent review of the morphological differences between the species, and the status of various populations remains uncertain. A population from central Myanmar (described as *Mus lepidoides*) is tentatively associated with *M. booduga*.

HABITAT USE: pygmy mice are found in a variety of agro-ecosystems, including irrigated rice and mixed vegetable cropping. Although they are generally not reported as true commensals, *M. terricolor* has been caught inside village houses in eastern Bangladesh.

Home-range estimates of around 800–1275 m² are reported from various localities in India, but it is unclear which species is represented.

NESTING BEHAVIOUR: burrows are located in bunds between damp or flooded fields, but in the floor of dry fields. The burrows in bunds are deep and branched, with 2–4 entrances and 1–2 nest chambers. Those in fields are shallow and have a simple tunnel leading to a single chamber. The entrances of both kinds of burrow are small compared with those of other rodents, rarely exceeding 1 cm in diameter, and they typically feature a small pile of excavated soil pellets.

There are several reports of hoarding behaviour. One study found rice grain in 8 of 10 excavated burrows, and 36–85 individual grains per burrow. Another study reported caches of up to 7 g.

BREEDING BIOLOGY: in Kerala State, India, pygmy mice breed in all months of the year. The pregnancy rate averaged through the entire year is around 20%, but there is a peak in pregnancies (to >70% of adult females) during the monsoon season. The smallest pregnant female recorded weighed 6.5 g.

The gestation period in captivity is given as 19–22 days. Estimates of litter size range from 1–6 to 6–13, suggesting a possible interspecific difference. The interval between litters in one population was found to be 45 days.

POPULATION DYNAMICS: no information available.

DAMAGE TO CROPS: rice grain is often observed in and around the burrows of pygmy mice. Analysis of stomach contents shows a seasonal shift from predominantly leafy material early in the rice cropping season to mixed leaf/grain during the seed-ripening phase of the crop cycle. High population densities are also reported in vegetable fields, but the extent of any damage is not reported.

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Rao, A.M.K.M. 1979. Monthly variation in the numbers and reproduction activity of the Indian field mouse, *Mus booduga* Gray around Tirupati. Indian Journal of Rodentology, 1, 35–45.

Rao, A.M.K.M. 1992. The mice, *Mus* spp. In: Prakash, I. and Ghosh, P.K., ed., Rodents in Indian agriculture, volume 1. Jodhpur, Scientific Publishers, 147–164.

Rao, A.M.K.M. and Rajabai, B.S. 1979. Morphological variation in the little Indian field mouse, *Mus booduga booduga* (Gray, 1837), inhabiting two different habitats. Säugetierkundliche Mitteilungen, 27, 317–320.

Adult specimens of *Mus booduga* from central Myanmar.

Adult specimens of *Mus terricolor* from Bangladesh.

Mus caroli (Bonhote, 1902)

COMMON NAMES: long-tailed rice-field mouse, Ryukyu mouse

Mus caroli can be quite common in and around rice fields and it presumably causes some damage to crops. It is often found together with *Mus cervicolor*.

MORPHOLOGICAL FEATURES: a small mouse with a relatively long, distinctly bicoloured tail (darker above than below), and dark-orange upper incisors. The back is brownish-grey, and the belly is white, with grey bases in the north of its range, but pure white in central and south-eastern Thailand and on Java, Indonesia. The chin and lips are white in all populations. The fur on the back and flanks contains narrow spines and varies in texture from soft to moderately stiff. The pes is relatively large and is either pure white or peppered with dark hairs. The plantar pads on the pes are prominent and the inner and outer metatarsal tubercles are positioned close together.

MAMMAE: 1+2+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Mus formosanus, Mus caroli ouwensi.*

DISTRIBUTION: widespread on mainland Southeast Asia—from central and eastern Thailand, south to the Isthmus of Kra, Laos, Cambodia, and all of Vietnam except the northernmost highlands. Outside this core area, the species is found in widely scattered localities, many of which probably represent human introductions—these include South Kedah State in Malaysia, north-eastern Sumatra, central and eastern Java and Flores in Indonesia, and Hainan Island, Fujian Province, Hong Kong and Taiwan in China, and various islands in the Ryuku Archipelago and in Japan. A population of *Mus caroli* was recently located in central Myanmar.

TAXONOMIC ISSUES: none.

HABITAT USE: common in rice fields and grasslands across its range. *Mus caroli* is also recorded from pine savannah in Loei Province, Thailand.

Adult measurements	Thailand	Cambodia	Southern Vietnam	South Kedah	Central Myanmar
Weight (g)	12	12.5–16	11.5–17.5	to 13	18–19.5
Head+body (mm)	76	72–95	73–86	79	80–87
Tail length (mm)	78	75–89	78–89	84	89–95
Pes length (mm)	18	14.5–19	16–18	18	17–19
Ear length (mm)	14	12.5–14	13	–	12–13

NESTING BEHAVIOUR: burrows are often constructed in rice-field bunds. The burrows typically have two entrances with a central chamber filled with rice straw. The entrances are left open through the day and are marked by small mounds of excavated soil.

BREEDING BIOLOGY: nothing reported.

POPULATION DYNAMICS: nothing reported.

DAMAGE TO CROPS: no specific information. In Cambodia, where *M. caroli* and *Mus cervicolor* occur together, mice as a group are said to climb tillers to feed on individual grains. In Laos, they are said to clean up fallen grains and panicles left behind by larger pests such as *Rattus* and *Bandicota* spp.

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Marshall, J.T. 1977. A synopsis of Asian species of *Mus* (Rodentia, Muridae). Bulletin of the American Museum of Natural History, 158, 173–220.

Adult *Mus caroli* from central Myanmar.

Lower surface of pes; adult from Laos.

Adult from central Myanmar.

Mus cervicolor (Hodgson, 1845)

COMMON NAMES: short-tailed rice-field mouse, fawn-coloured mouse

Mus cervicolor is a widely distributed species often found together with *Mus caroli* in rice fields. It is distinguished from *M. caroli* by its shorter tail, softer fur and more delicate feet.

MORPHOLOGICAL FEATURES: a small, soft-furred mouse with a distinctively short tail and pale-orange or yellow incisors. The dorsal fur is orange–brown to brownish-grey, and the belly fur is cream with pale-grey bases. The fur around and behind the eye is a gingery colour. Numerous very fine spines are present in the dorsal fur, but these are not obvious to the touch and the fur feels soft. The tail is usually distinctly bicoloured (darker above than below) but it is sometimes mottled. The pes is relatively slender and delicate, but the plantar pads are moderately prominent. The inner and outer metatarsal tubercles are more widely separated than in *M. caroli*. The upper surface of the manus and pes is clothed in white fur, peppered with occasional dark hairs on the pes.

MAMMAE: 1+2+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAME: *Mus cervicolor popaeus*.

DISTRIBUTION: widespread on mainland Southeast Asia, from eastern Nepal through Myanmar, Thailand, south to the Isthmus of Kra, Laos, Cambodia, and southern and central Vietnam. There are scattered records from Bangladesh. Populations in north-eastern Sumatra and eastern Java, Indonesia, are probably the result of human introductions. *Mus cervicolor* is found across a wide altitudinal range.

TAXONOMIC ISSUES: two subspecies are distinguished in Southeast Asia—*M. cervicolor cervicolor* and *M. cervicolor popaeus* for lowland and upland populations, respectively. Laboratory crosses

Adult measurements	Nepal		Cambodia	Thailand
	Male	Female		
Weight (g)	14.1 ± 3.4	12.4 ± 4.3	8–16	14.6
Head+body (mm)	73.2 ± 7.2	70.8 ± 9.7	63–81	82
Tail length (mm)	81.9 ± 6.2	74.1 ± 7.4	53–65	59
Pes length (mm)	17.9 ± 1.1	16.3 ± 1.9	13.5–16	16.2
Ear length (mm)	14.6 ± 0.7	14.5 ± 1.5	12–16	14.6

between Thai individuals of the two subspecies resulted in no apparent reduction in fertility. The Nepalese population is the true *Mus cervicolor cervicolor*. The subspecies identity of the Bangladesh populations is not yet known.

HABITAT USE: in Thailand, the two forms can be found living in close proximity with typical *M. cervicolor* in rice fields and *M. cervicolor popaeus* in nearby forest habitat.

NESTING BEHAVIOUR: in Cambodia, *Mus cervicolor* has been dug from bunds alongside lowland rice fields. The burrows are relatively short but with multiple entrances, usually including one more-or-less vertical hole that opens on the surface of the bund. One burrow contained two young in a central brood chamber that lacked any nesting material.

BREEDING BIOLOGY: nothing reported

POPULATION DYNAMICS: nothing reported.

DAMAGE TO CROPS: nothing reported.

Adult *Mus cervicolor* from Cambodia.

Adult from lowland Laos (tip of tail is damaged).

Adult from Cambodia.

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Mus cookii (Ryley, 1914)

COMMON NAME: Cook's mouse

This species is often captured in upland rice fields. However, nothing is known regarding its impact on crops and it is probably best regarded as an occasional or minor pest species.

MORPHOLOGICAL FEATURES: *M. cookii* is a moderately large species of mouse with a long, narrow snout, large, broad ears, and a distinctly hairy, weakly bicoloured tail. In upland populations, the dorsal fur is a dark-brown colour and contains numerous broad spines, giving it a distinctly 'stiff' texture. The fur is paler and softer in samples from lower elevations. The belly fur is cream with dark-grey bases in upland Lao populations, but populations with pure-white belly fur are known from elsewhere in the region. The belly fur is said to be dark in juveniles from Thailand. The tail may be slightly shorter or slightly longer than the head+body. The pes is large and hairy, usually with a mixture of white and dark hairs. The plantar pads are prominent and the two metatarsal pads are close together.

MAMMAE: 1+2+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Mus famulus cookii*, *Mus palnica*, *Mus nagarum*.

DISTRIBUTION: widespread across mainland Southeast Asia, from north-eastern India, northern Myanmar, central Thailand, Laos, southern China (Yunnan Province) and central Vietnam.

TAXONOMIC ISSUES: Indian populations sometimes distinguished as *Mus nagarum* and *M. palnica* may be local variants of *M. cookii*.

HABITAT USE: reported from grass beneath pine forest, and from upland rice fields and gardens.

NESTING BEHAVIOUR: in northern Laos, *M. cookii* was flushed from straw piles in upland rice fields. However, it is unclear whether the animals were nesting among the straw or in small burrows observed below the straw piles.

BREEDING BIOLOGY: nothing reported.

Adult measurements	Thailand	Laos
Weight (g)	23	16.5
Head+body (mm)	96	77
Tail length (mm)	83	91
Pes length (mm)	19.5	19.5
Ear length (mm)	15	15

POPULATION DYNAMICS: nothing reported.

DAMAGE TO CROPS: nothing reported.

KEY REFERENCES:

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Marshall, J.T. 1977. A synopsis of Asian species of *Mus* (Rodentia, Muridae). Bulletin of the American Museum of Natural History, 158, 173–220.

Adult *Mus cookii* from the uplands of Laos.

Adult from the uplands of Laos.

Pes of an adult from the uplands of Laos: upper surface (left) and lower surface (right).

Mus musculus Group

COMMON NAME: house mouse group

Members of the house mouse group occur as truly wild animals in the northern temperate zone, from Western Europe through to southern China. In South and Southeast Asia, this group of mice is commonly found around human habitation, and only rarely in cropping areas. In some areas, they reach very high population densities and presumably cause significant postharvest losses. They are also said to damage household items such as clothes and bedding.

MORPHOLOGICAL FEATURES: moderately large mice with moderately long tails, soft fur on the back and flanks and distinctive pes morphology. Although colouration is highly variable across the full range of this group, South and Southeast Asian house mice are usually a plain-brown or grey–brown colour, with the belly fur similar in colour to that on the back. Occasional specimens have cream- or buff-tipped belly fur. The ears are relatively large compared with other Asian *Mus* species. The tail is usually longer than the head+body and is either the same colour above and below or very slightly paler below. The manus and pes are usually covered in dark hairs. All of the plantar pads on the pes are small, and the outer metatarsal pad is sometimes absent. When both metatarsal tubercles are present, they are very widely separated and this feature will distinguish members of this group from other Asian *Mus* species.

MAMMAE: 1+2+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Mus domesticus, Mus castaneus, Mus musculus castaneus, Mus musculus homourus, Mus musculus domesticus.*

DISTRIBUTION: widely distributed throughout the region, with populations in most major towns. In some areas (e.g. eastern Bangladesh), house mice are also common in many of the smaller villages. The mapped distribution may be incomplete as many populations may be undocumented and the range is presumably still expanding.

TAXONOMIC ISSUES: a wild member of this group (generally distinguished as *M. musculus castaneus*) is found in agricultural contexts and in natural grasslands of southern China through to central

Adult measurements	Bangladesh	Thailand
Weight (g)	to 26	13
Head+body (mm)	26–95	75.9
Tail length (mm)	45–117	79.4
Pes length (mm)	13–20	16.5
Ear length (mm)	–	–

Asia. However, across most of South and Southeast Asia, members of this group are truly commensal, with populations restricted to towns and villages. These populations are potentially of very mixed origin, with input from Asian *M. musculus castaneus* and one or both of *M. musculus musculus* and *M. musculus domesticus*, of Eastern and Western European origin, respectively. Here we will refer to them as *'Mus musculus'*, indicating their uncertain status within the *Mus musculus* group.

HABITAT USE: in South and Southeast Asia, *'M. musculus'* is usually confined to houses and other buildings. It is occasionally captured in village gardens or animal pens, but is generally excluded from cropping areas by the presence of other *Mus* species such as *M. caroli*, *M. cervicolor*, *M. booduga* and *M. terricolor*. In a study of farm household rodent populations at Joydebpur, Bangladesh, *M. musculus* accounted for 53.4% of all captures (otherwise captures of *Suncus murinus* > *'Rattus rattus'* > *Bandicota* spp.). All captures of *M. musculus* were made inside buildings.

NESTING BEHAVIOUR: *'M. musculus'* uses a variety of nesting sites, including burrows excavated in the walls and floors of buildings, or under piles of straw. Nests are sometimes also constructed in piles of grain bags or amongst stored cloth. In India, burrows usually have 1–3 entrances with 1–2 brood chambers measuring 5–7 cm in diameter. No evidence has been found of hoarding behaviour.

BREEDING BIOLOGY: in Rajasthan, India, *'M. musculus'* has a gestation period of 18–21 days with an average interval between litters of 50 days. Female breeding activity usually commences at around 45 days. Breeding is seasonal in the semi-arid environment of Rajasthan. In the Comilla District of eastern Bangladesh *'M. musculus'* appears to breed year-round in farm households, with embryo counts ranging from 1–10 (modal values of 5–6).

POPULATION DYNAMICS: the highest capture rates of *M. musculus* in farm households in Joydebpur were obtained in October–January and May–July, corresponding to periods of changing weather followed by major rice harvests.

DAMAGE TO CROPS: at high population densities, house mice presumably cause significant damage to stored grain and other foods. They are also said to damage household items such as clothes and furniture.

KEY REFERENCES:
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Adult *Mus musculus* from Bangladesh.

Adult from Bangladesh, lower surface of pes.

Adult from Australia.

Nesokia indica (Gray and Hardwicke, 1830)

COMMON NAMES: mole rat, short-tailed mole rat

Nesokia indica is a highly fossorial species found in arid regions from South Asia to the Middle East and North Africa. It is a major pest species in north-western Bangladesh, where it causes extensive damage to wheat, rice and sugarcane. The burrow systems of this species are elaborate and it is reported to spend long periods below ground without emerging.

MORPHOLOGICAL FEATURES: a medium-size, stocky rat with a broad head, short snout, very wide incisors, short, rounded ears, and a very short, thinly-furred tail that in adults is ≥50 mm shorter than the head+body. Uniquely among the Asian murid rodents, the lower pair of incisors of *Nesokia* is wider than the upper pair. The fur is thick and shaggy, grey–brown on the back and flanks, but with an orange mantle across the shoulders. The guard hairs are short and inconspicuous, even on the lower back. The belly fur is pale grey. The claws

are strongly developed on both the manus and pes. Unlike all *Bandicota* species, the inner and outer plantar tubercles are both rounded in shape and of approximately equal size (the inner tubercle is longer and elongated in *Bandicota* spp.).

MAMMAE: 1+1+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: numerous subspecies names; the population in Bangladesh may be typical *Nesokia indica indica*.

DISTRIBUTION: a large area of South Asia and Eurasia—from western Bangladesh and northern India to Israel, north-eastern Egypt and Tadzhikistan.

TAXONOMIC ISSUES: numerous regional populations have been assigned species or subspecies names. The genus is in need of revision using modern methods.

HABITAT USE: in India, *N. indica* shows a preference for moist areas with relatively soft soil and some vegetation cover. Burrow systems are usually located in large field bunds and banks of major canals, and only rarely in flat fields.

This species appears to be entirely herbivorous. A significant proportion of feeding is done from below ground.

NESTING BEHAVIOUR: burrow systems are extensive and elaborate, with a pyramidal pile of soil

Adult measurements	Bangladesh
Weight (g)	170 ± 33
Head+body (mm)	to 194
Tail length (mm)	to 128
Pes length (mm)	to 37
Ear length (mm)	to 20

at each opening. Most have multiple entrances (up to 19) and a zigzagging internal arrangement. The longest recorded burrow was 34.5 m in total length, measured in India.

BREEDING BIOLOGY: litter size is relatively low, with estimates in wild populations in South Asia of 3.8 ± 0.25 and 4.1 ± 0.5. The highest recorded litter was eight pups. Vaginal perforation is recorded at body weights below 40 g but the smallest pregnant female weighed 63 g. First pregnancies are more typical in the weight range 100–119 g, corresponding to an estimated age of 120 ± 7 days. The gestation period in South Asia is variously reported as 24 and 30 days; the different estimates perhaps reflect delayed implantation in lactating females (see Chapter 6). Newborn pups have an average weight of 4.6 g for males and 4.1 g for females. The mean interval between births in outdoor pens is 36 ± 2.6 days.

The pregnancy rate of a wild population in the Punjab was 30%, averaged through the year. Wild populations in Pakistan show two peaks in breeding activity, corresponding to cooler months but avoiding periods of extreme cold. In a rice-growing area of Pakistan, the average female productivity was only 10.6 pups per female.

POPULATION DYNAMICS: nothing is reported.

DAMAGE TO CROPS: *N. indica* is reported to cause damage to a wide variety of crops, including cereals (rice, wheat, barley), potato, peanut, sugarcane, melons and tomato. In rice-growing areas of Pakistan, *N. indica* consumes only the ripening grain.

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Adult specimens of *Nesokia indica* from Bangladesh.

Adult, lower surface of pes.

Rattus argentiventer

(Robinson and Kloss, 1916)

COMMON NAME: rice-field rat

Rattus argentiventer is the major agricultural rodent pest across much of island and mainland Southeast Asia. Its apparent natural preference for waterlogged areas with dense grassy cover makes it ideally suited to life in rice fields. Moreover, an unusually high litter size allows the species to undergo rapid population increase at times when food is abundant.

MORPHOLOGICAL FEATURES: a medium-size rat with moderately spiny, orange–brown dorsal fur that is typically flecked with black. The belly fur can vary from silvery-white to a dull grey in colour, and there is often a darker streak along the midline of the belly. The snout is moderately long and the ears are large and lightly furred. A distinct orange fringe of fur is usually present just forward of the ear, although this may fade in older animals. The tail is usually just shorter than the head+body and is dark above and below. The pes is relatively long and narrow, and usually has a broad band of dark hairs on the upper surface, extending forward from the ankle.

MAMMAE: 1+2+3 (compared to 1+1+3 in all *R. losea* and in a variable but often high proportion of *R. rattus*).

This species is most often confused with *R. losea*, '*R. rattus*' and *R. tiomanicus*. The orange ear fringe is uniquely diagnostic for *R. argentiventer*, but absence of this fringe, especially in an old adult, is not informative. In comparison to *R. argentiventer*:

- *R. losea* is a smaller species with softer, grey–brown to orange–brown dorsal fur and a grey-based but cream or white-tipped belly fur. The pes of adult *R. losea* is shorter and narrower than that of an equivalent-size (but younger) *R. argentiventer*. The ears of *R. losea* are distinctly smaller and furrier than in *R. argentiventer* and the tail is relatively shorter and more finely scaled. *Rattus losea* and *R. argentiventer* are often found living together in Vietnam and Cambodia.
- *R. tiomanicus* usually has a longer tail and a slightly shorter, broader pes. The dorsal fur is

Adult measurements	Thailand	Northern Vietnam	Malaysia	Sumba Is, Indonesia
Weight (g)	to 212	52–239	–	–
Head+body (mm)	to 204	136–205	160–194	176–230
Tail length (mm)	to 187	149–195	165–210	172–201
Pes length (mm)	to 39	30–38	34–41	35–40
Ear length (mm)	to 22	16–22	20–24.5	20–24

a plainer brown colour and of sleeker texture, with less prominent guard hairs, especially on the lower back. The pure-white belly fur of *R. tiomanicus* contrasts with the grey or silvery belly of *R. argentiventer*

* the European 'black rat' form of '*R. rattus*' is easily distinguished from *R. argentiventer* by its dark belly fur, much longer tail and broader pes. However, many Asian populations of '*R. rattus*' have relatively short tails, a narrower pes and a high frequency of white-bellied individuals. These populations are much more similar to *R. argentiventer*, but can be distinguished by consulting the following suite of characteristics: the orange ear fringe (never present in '*R. rattus*') pes colour (generally less extensive dark fur in '*R. rattus*'), pes shape and pad morphology (pes slightly broader and pads more prominent and strongly striated in '*R. rattus*'), dorsal fur colour (usually less peppered with black in '*R. rattus*'), belly fur colour (either grey-based or pure creamy white in '*R. rattus*', rarely pure grey or silvery), tail length (not often shorter than head+body in '*R. rattus*'), tail scale size (slightly smaller scales in '*R. rattus*'), and ear furring (less densely furred in '*R. rattus*').

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:
Rattus rattus argentiventer, Rattus rattus brevicaudatus, Rattus rattus bali, Rattus rattus umbriventer.

DISTRIBUTION: lowland areas of southern Thailand, Cambodia and Vietnam, extending along the Mekong River into southern Laos, and along the length of the Malay Peninsula; all of the major islands of Indonesia, east to Sulawesi and Timor. Isolated populations are present in southern New Guinea and on the islands of Cebu, Luzon, Mindanao, Mindoro and Negros in the Philippines. On Timor, a population was found living in terraced rice fields in a narrow valley on the lower slopes of Gunung Mutis, in the central highland area.

The populations on Sulawesi, the Lesser Sunda Islands, the Philippine islands and New Guinea presumably were introduced by people in recent prehistoric or historic times.

TAXONOMIC ISSUES: some slight differences in body proportions are found between major island populations in Indonesia. These populations may represent different subspecies.

HABITAT USE: found in and around rice fields, gardens and orchards, and in adjacent areas of fallow grassland across its entire range; generally absent from village habitats, except as a vagrant. *Rattus argentiventer* is probably most abundant in areas that experience regular and extensive seasonal inundation—a condition that may favour this species over other, less water-tolerant rodent species.

Rattus argentiventer eats a wide variety of food items, including green foliage (e.g. grasses and paddy weeds), grass seeds including cereal grains, and invertebrates (e.g. crabs, molluscs and insects). In captivity, the species survives best when fed on cereal grains or other starchy foods.

Males and females both dig burrows but those dug by breeding females are more extensive. Burrows are often located in low bunds between fields, in the banks of irrigation canals, and in and around raised vegetable gardens or orchards.

Patterns of habitat use are best known from studies in West Java, Indonesia. In this area, during fallow periods and through early stages of crop growth, burrows are concentrated in refuge habitats, such as along canals and in upland garden areas. At this time, individual rats often travel considerable distances between burrows and feeding areas, generally moving along regular trails. At later stages of crop growth, when the rat population is increasing and the crop provides dense cover, more burrows can be found in the low bunds between fields and some rats also take shelter in the field through the day. After a field is harvested, many rats take shelter in piles of straw left in the fields, while others move to exploit nearby unharvested fields. Individual movements decrease during this period, perhaps due to increased predation risk in the more open, postharvest habitat.

NESTING BEHAVIOUR: burrows dug by breeding females tend to be more extensive than those occupied by males or non-breeding females. Communal burrows have not been recorded, but one burrow can house two or more litters from a single female. The burrow is presumably enlarged to accommodate each successive litter.

BREEDING BIOLOGY: breeding activity is closely linked to the growth and maturation of rice crops, with first mating taking place just before maximum tillering. Many females produce their first litter as the rice reaches booting stage, their second litter during the ripening stage, and a third litter shortly after harvest. Members of the first litter usually do not get an opportunity to breed within a single cropping cycle. However, they may do so if adjacent crops are planted 2–3 weeks later, thereby extending the availability of abundant, high-quality food.

Females become perforate and begin ovulating at body weights of 31–40 g, or around 28 days of age. However, most females do not experience their first pregnancy until body weight reaches 60–120 g. Males appear to mature somewhat later, with full testicular descent only in males weighing more than 90 g (at an estimated 59 days old). The gestation period is 20–26 days, with a mean of 21 days. The interval between births in captive animals is usually 20–25 days.

Embryo counts as high as 18 have been recorded for *R. argentiventer*, but the average embryo count is around 7–8 for captive animals. Litter size in wild-caught samples was 5–7 (mode of 6) in Malaysia and 11–12 for West Java, Indonesia. An unusual aspect of reproductive behaviour in *R. argentiventer* is the frequent presence of multiple litter generations within a single breeding burrow. More typically among rodents, the birth of a new litter leads to the obligate dispersal of previous offspring. This behaviour in *R. argentiventer* may partly explain how this species is able to achieve such high local population densities.

Very high adult female pregnancy rates, close to 100%, have been recorded for *R. argentiventer* in West Java. This, together with the high litter size, may also explain the very high seasonal abundances of this species.

POPULATION DYNAMICS: the seasonal breeding activity generally mirrors the cycles of the rice-farming system, with one breeding season in areas with a single rice crop per year and two breeding seasons in areas that practise double cropping. Triple cropping, as practised increasingly in areas with reliable irrigation water, can lead to more-or-less continuous breeding, especially where the crops are grown asynchronously for reasons of water or labour management.

The close link between breeding and cropping cycles generally leads to pronounced seasonal changes in abundance, with a rapid increase in numbers during the crop-ripening stage and a dramatic collapse after harvest. Local population densities as high as 500–600 individuals/ha have been recorded at field sites in Indonesia where double cropping is practised. Local densities may be even higher in areas with triple cropping. Annual, deep flooding also seems to promote very large fluctuations in population size in *R. argentiventer*, presumably as a result of high mortality caused by the flooding itself and by the long period of enforced fallow.

DAMAGE TO CROPS: damage occurs at all stages of growth of the rice plant, but is perhaps most intense around the booting and milky stages. Crop losses in rice-growing areas where the rodent community is dominated by *R. argentiventer* are typically in the order of 10–20%. In areas with double cropping, losses are typically higher during the second crop.

Chronic losses in the order of 30–50% are reported for fields positioned close to refuge habitat such as major canals or extensive 'upland' areas. Very high chronic losses are also reported in areas where triple cropping is practised and rat densities are especially high.

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Adult *Rattus argentiventer* from Indonesia.

Adult tail; specimen from Vietnam.

Adult from Cambodia.

Juvenile *Rattus argentiventer* from Vietnam.

Pes of adult from Indonesia: upper surface (left) lower surface (right).

Adult from Vietnam.

Adult from Indonesia.

Rattus exulans _(Peale, 1848)

COMMON NAMES: Pacific rat, Polynesian rat, kiori

Rattus exulans is the major field and village rat in many parts of Melanesia, Micronesia and Polynesia. In Indonesia through to mainland Southeast Asia, it is usually restricted to village houses and gardens, and is less commonly found in the major cropping areas. In New Zealand and on some other islands, *R. exulans* has declined in abundance and geographical range following the introduction of *Rattus rattus* and *R. norvegicus*.

MORPHOLOGICAL FEATURES: a small, reddish-brown to grey–brown rat with spiny, reddish-brown dorsal fur and cream- or white-tipped belly fur with grey bases. The facial vibrissae are very long and typically reach beyond the ears when folded back. The tail is usually longer than the head+body and is uniformly dark above and below. The upper surface of the pes is white, but often with a strip of dark hairs along the outer edge. This species is much smaller than any other pest *Rattus* and is often misidentified as a mouse. The presence of an elongated inner metatarsal pad on the pes is one feature that distinguishes *R. exulans* (even juveniles) from all species of *Mus* (all with a rounded pad).

MAMMAE: 1+1+2.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus concolor*, *Rattus browni*.

DISTRIBUTION: mainland Asia from eastern Bangladesh to central Vietnam, the Malay Peninsula, Taiwan and the southern Ryukus; all major and most small islands of Indonesia and the Philippines; the island of New Guinea and its satellites, and beyond into Island Melanesia, Micronesia, Polynesia and New Zealand; Adele Island, off the north-western coast of Australia.

The exact place of origin of *R. exulans* is unknown, but the species is probably of mainland Southeast Asian origin. It was introduced into eastern Indonesia and the wider Pacific region with early seafarers, mostly within the last 2000–3000 years.

TAXONOMIC ISSUES: there appears to be no significant geographical variation, except that populations on small islands tend to be larger.

Adult measurements	Bangladesh	Southern Vietnam	Pacific Islands
Weight (g)	34–40	23–42	–
Head+body (mm)	105–120	91–130	75–165
Tail length (mm)	120–135	105–146	102–197
Pes length (mm)	22–28	21–26	23–30
Ear length (mm)	–	15–18	–

HABITAT USE: highly arboreal, often seen climbing around in tall grasses or low trees, and on the walls and roofs of houses. *Rattus exulans* is usually confined to villages and household gardens, but is also present in areas of disturbed forest and regrowth vegetation. In New Guinea, it is very common in tall grassland habitat, such as *Imperata* and canegrasses.

In lowland areas of Bangladesh, Laos, Cambodia and Vietnam, *R. exulans* coexists in village houses with various members of the *R. rattus* Complex, all of which are substantially larger-bodied. In the uplands of Laos, *R. rattus* is dominant in village habitat and the smaller species is rarely, if ever, present.

In areas that lack any surviving native rodent species, such as the mountains of Timor, Indonesia, *R. exulans* can be found in primary forest, far from human habitation.

NESTING BEHAVIOUR: usually constructs a leaf or grass nest, most often in dense grass and positioned 20 cm or more above the ground. Inside buildings, nests are usually located in roof thatch, less often in piles of straw or other material on the ground.

BREEDING BIOLOGY: studies in Malaysia and Papua New Guinea (PNG) suggest year-round breeding but with reduced output during the cooler months in PNG. In Hawaii, where the climate is more strongly seasonal, breeding is restricted to the wetter months.

The reproductive life span of females in Hawaii is less than one year. The gestation period for non-lactating females is 23 days, but 3–7 days longer for a lactating female.

Litter size in PNG and Hawaii ranges from 1–7, with a modal value of 4 in both populations. In Malaysia, the maximum recorded litter size is 10, but with a mean of 3.8 and a mode of 4. In Hawaii, females develop a perforate vagina from 32–44 g, and males develop scrotal testes at 41–57 g. In Malaysia, females with a body weight above 30 g are commonly pregnant. Estimates of reproductive output (number of young per female per year; based on litter size and the average proportion of adult females pregnant) range from 9.8 on Ponape in Micronesia, to 17.2 in Hawaii and 25.7 in Malaysia.

POPULATION DYNAMICS: population density estimates are available for various Pacific islands, including 1–3/ha in coconut plantation on Guam; 7–12/ha in grassland and 11–24/ha in coconut plantation on Ponape; and 7–30/ha for various habitats on Tokelau.

DAMAGE TO CROPS: on mainland Southeast Asia and through Indonesia, this species is usually confined to village habitats, where it attacks household gardens and damages stored food. However, in New Guinea and on many of the smaller Pacific islands, *R. exulans* is the major agricultural pest rodent, causing damage to root crops, coconuts, fruits and vine vegetables such as beans. In some parts of Thailand, Malaysia and the Philippines (including Palawan), *R. exulans* is reported as a significant field pest of rice crops.

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Adult *Rattus exulans* from highlands of Papua New Guinea.

Adult from Vietnam.

Inner metatarsal tubercle

Lower surface of pes; adult from Vietnam.

Upper surface of pes; adult from Vietnam.

Rattus losea (Swinhoe, 1871)

Common name: lesser rice-field rat

Rattus losea appears to be discontinuously distributed across mainland Southeast Asia and East Asia north to Taiwan, and it displays regional variation in its morphology. Although it is often mentioned as an agricultural pest species, the role of *R. losea* in this regard is overshadowed by the fact that it often occurs together with larger-bodied species, such as *R. argentiventer* in Vietnam and Cambodia, and *R. rattus* in Thailand and Laos. Accordingly, little is known of its basic biology.

MORPHOLOGICAL FEATURES: there appear to be two major populations of *R. losea* that differ in morphology and genetics. All populations are distinguished from other Southeast Asian *Rattus* species by their smaller ears, softer dorsal fur that lacks any obvious spines, and shorter, more finely scaled tail.

Typical *R. losea* from Taiwan through to the north of Vietnam is a medium-size, terrestrial rat with dull, grey–brown dorsal fur and grey-based but white- or cream-tipped belly fur. The tail is usually 5–15 mm shorter than the head+body and is weakly 'bicoloured' (darker above than below), especially in younger animals. The pes is usually white, but sometimes with a narrow dark band of hairs down the outer side.

Populations currently referred to *R. losea* from the south of Vietnam, Cambodia, Thailand and

lowland areas in Laos are smaller and more richly coloured, with brown to reddish-brown dorsal fur, darker grey but buff-tipped belly fur, a shorter tail that is dark above and below, and darker feet. Cambodian specimens are a rich, reddish-brown colour, contrasting with plainer, brown-coloured populations in surrounding areas.

MAMMAE: 1+1+3 in all populations.

Adult measurements	Thailand	Northern Vietnam	Southern Vietnam	Taiwan
Weight (g)	to 77	22–90	38–92	–
Head+body (mm)	128–160	120–177	120–160	to 185
Tail length (mm)	75–163	128–165	113–140	to 170
Pes length (mm)	28–33	24–32	28–32	to 32
Ear length (mm)	15–20	16–20	15–18	to 21

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:
Rattus exiguus, possibly *R. hoxaensis* (usually treated as a synonym of *R. argentiventer*).

DISTRIBUTION: found across much of lowland Southeast and East Asia. As indicated above, there appear to be two distinct forms of *R. losea*. True *R. losea* (described from Taiwan) appears to be distributed across southern China (including Hainan Island) through to northern and central Vietnam. The second form is found in the Mekong Delta region of southern Vietnam through to Cambodia and Thailand, and north to Vientiane Province in Laos. An apparently isolated population of *R. losea* occurs on the Malay Peninsula, just south of the Isthmus of Kra. The relationship of this population to the two groups mentioned above is currently unclear.

TAXONOMIC ISSUES: the populations of *R. losea* from northern and southern Vietnam to Cambodia are genetically distinct. To date, Thai samples have not been included in any genetic analysis and their affinities remain uncertain.

HABITAT USE: reported from across its range as an inhabitant of rice fields and associated vegetable gardens and orchards. In most localities, *R. losea* appears to be less abundant than larger-bodied, co-occurring *Rattus* species (either *R. argentiventer* or '*R. rattus*', depending on location). In Vinh Phuc Province, northern Vietnam, the abundance of *R. losea* in any habitat appears to be inversely proportional to that of *R. argentiventer*, perhaps the result of active competition between these species. In the Mekong Delta, southern Vietnam, *R. losea* is most abundant in areas that experience heavy flooding but where there are significant areas of upland habitat. Conversely, it appears to be least abundant in areas that experience more widespread and uniform inundation.

Rattus losea is recorded as the dominant pest species (90% of captures) in Prachin Buri Province, Thailand, where deep-water ('floating') rice is grown once a year, from June to December. This population of *R. losea* shows a clear cyclical pattern, with numbers peaking just after harvest and then falling steadily through until the following planting season. The fact that animals were captured in floating live-traps indicates an ability to move about freely in the deep-water habitat.

In Chaiyaphum Province, Thailand, *R. losea* occurs in natural grassland beneath pine forest at an altitude of 850 m. While this may represent a natural habitat for the species, much caution is needed to distinguish genuinely natural from feral populations of any widespread pest species.

NESTING BEHAVIOUR: *R. losea* has been dug from burrows in rice-field bunds in both northern and southern Vietnam and in Thailand. In Prachin Buri Province, breeding takes place at a time when the fields are deeply inundated; unfortunately, it is not reported where *R. losea* builds its nests under such conditions.

BREEDING BIOLOGY: in Prachin Buri Province, breeding of *R. losea* commences in September and lasts through until harvest of the deepwater rice in December. In the north of Vietnam, breeding activity is linked to rice cropping cycles, starting around maximum tillering and ending a few weeks after harvest. The average litter size is 7.5 in this population.

POPULATION DYNAMICS: population cycles have been studied in both northern and southern Vietnam. In both areas, *R. losea* numbers fluctuate in response to the availability of field crops, especially rice. However, the amplitude of the fluctuations is not as high as for co-occurring populations of *R. argentiventer*.

DAMAGE TO CROPS: damage caused by *R. losea* has not been distinguished from that caused by *R. argentiventer* or *R. rattus*.

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Adult specimens of *Rattus losea* (top) and *Rattus argentiventer* (below) from Cambodia showing differences in body size and relative tail length.

Pes of adult *R. losea* from southern Vietnam; upper surface (left) and lower surface (right).

Adult *Rattus losea* from Cambodia.

Rattus New Guinean species:

Rattus mordax (Thomas, 1904), *Rattus praetor* (Thomas, 1888) and
Rattus steini Rümmler, 1935

COMMON NAMES: New Guinean spiny rats (*R. mordax* and *R. praetor*),
Stein's rat (*R. steini*)

R. mordax

R. praetor

R. steini

Three native *Rattus* species (*R. mordax*, *R. praetor* and *R. steini*) cause significant crop damage in New Guinea and nearby islands, with impacts on both subsistence gardens and plantations. All are probably disturbance specialists that were advantaged by the development and spread of agricultural practices in Melanesia over the last few thousand years. One of the three species (*R. praetor*) was carried during prehistoric times into more remote parts of the Pacific.

MORPHOLOGICAL FEATURES: all three species are medium-size, terrestrial rats with grizzled, reddish- to dark-brown dorsal fur and unicoloured tails that are 20–30 mm shorter than the head+body. In addition:

- *R. mordax* is a spiny-furred rat with few projecting guard hairs. The dorsal fur is rust-brown with yellowish-brown tipping, and the belly fur is a dull-cream colour. The pes is moderately broad and covered with short, brown hairs

- *R. praetor* also has coarse, spiny dorsal fur but with the addition of long, black guard hairs. The dorsal fur is brown, sometimes with reddish tipping, and the belly fur is pale-grey or ivory coloured, often with a pure-white chest patch. The pes is narrow and has a covering of light-brown hairs

- *R. steini* has soft dorsal fur with few, if any, spines but with fine, projecting guard hairs. The belly fur is grey with buff tipping, and the pes is narrow, with a covering of cream or light-buff hairs.

Adult measurements	R. mordax	R. praetor	R. steini
Weight (g)	–	164–228	110–220
Head+body (mm)	142–254	157–245	140–193
Tail length (mm)	115–203	144–181	136–155
Pes length (mm)	28–44	34–39	33–37
Ear length (mm)	–	18–20	16–21

MAMMAE: 0+1+2 or 0+2+2 in *R. steini*; 0+2+2 in *R. praetor* and *R. mordax*.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:

- for *R. mordax*: *Rattus ringens mordax, Rattus leucopus mordax, Rattus ruber mordax, Rattus ruber*
- for *R. praetor*: *Rattus leucopus praetor, Rattus ringens praetor, Rattus ruber praetor, Rattus ruber*
- for *R. steini*: *Rattus ringens steini, Rattus ruber steini, Rattus ruber.*

DISTRIBUTION: *R. mordax* is confined to the Huon Peninsula and south-eastern 'tail' of New Guinea. Most records are from elevations below 600 m. A distinctly larger subspecies (*R. m. fergussoniensis*) is found on many of the islands of the D'Entrecasteaux group and the Louisade Archipelago, and on Woodlark Island of the Trobriand group. *Rattus praetor* occurs in the northern lowlands of New Guinea and on New Britain and the Solomon Islands to the east. The species is also recorded from recent fossil remains from Vanuatu and Fiji, far outside of its current range. All of the island populations probably resulted from prehistoric human introductions. *Rattus steini* is widely distributed at mid-altitudes (approximately 500–2500 m) along the central mountain chain of New Guinea, with isolated populations in the northern ranges and on the Huon Peninsula. Three subspecies of *R. steini* are distinguished on the basis of slight morphological differences and variation in mammary formulae.

TAXONOMIC ISSUES: each of the New Guinean pest species of *Rattus* was formerly included under the species *R. leucopus, R. ringens* and *R. ruber*. *Rattus leucopus* (with subspecies *ringens*) is a distinct, forest-dwelling species found in the lowlands of southern New Guinea and northern Australia. The name *Rattus ruber* was based on a New Guinean example of *R. nitidus*.

HABITAT USE: all three species occur in low numbers in primary rainforest but they are more abundant in subsistence gardens and plantation areas, where they are usually the most commonly caught species of rat. *Rattus steini* and *R. mordax* are also common in anthropogenic grasslands created by gardening and burning. None of these species is reported as a true resident of village habitat. Indeed, everywhere across New Guinea, this niche is occupied by the introduced species *R. exulans* and *R. rattus*. However, *R. mordax* will occasionally enter village houses to attack stored rice and sweet potato.

NESTING BEHAVIOUR: all three species are reported to dig burrow systems. Females of *R. steini* and *R. praetor* are known to raise litters in this context.

BREEDING BIOLOGY: breeding is recorded at all times of year in *R. steini* and *R. praetor*. Estimates of mean litter size are 4.5 (range 2–7) for *R. praetor*, 2.7–3.4 (range 2–5) for *R. steini*, and 2.3 (range 2–4) for *R. mordax*.

POPULATION DYNAMICS: in the Southern Highlands of Papua New Guinea, *R. steini* remains common in abandoned gardens for the first nine months of successional regeneration. After that, its numbers decline as a shrub layer develops and various forest-dwelling species of rodents become re-established.

DAMAGE TO CROPS: there are few specific reports of crop damage, but all three species are identified by subsistence farmers as significant pests. Damage commonly occurs to tuber crops, especially to sweet potato.

Damage to stored food by *R. mordax* was noted above.

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Adult *Rattus mordax.*

Adult *Rattus steini.*

Adult *Rattus praetor.*

Rattus nitidus (Hodgson, 1845)

COMMON NAME: Himalayan rat

Rattus nitidus is a major agricultural pest species in rice- and wheat-growing areas of Sichuan Province, southern China. In some of the more mountainous parts of Southeast Asia, it is said to be a common village pest. Although this species has been introduced in recent prehistoric or historic times to various Philippine and Indonesian islands, its pest status in these areas remains undocumented.

MORPHOLOGICAL FEATURES: a medium-size rat with soft, woolly fur that is brown dorsally, and cream but grey-based on the belly. The snout is long and broad, and the ears are large and lightly furred. The tail is approximately equal in length to the head+body and is weakly 'bicoloured' (dark above, paler below). The pes is relatively long and narrow, and clothed in pure white hairs. The manus and lower fore-limb are also pure white.

MAMMAE: 1+2+3.

Measurements from published sources suggest that more northerly populations (Sichuan and Gansu Provinces of China) grow to a considerably larger size than those from Southeast Asian localities, e.g. maximum body weights recorded for Sichuan are 273 g for males and 320 g for non-pregnant females, compared with 122 g for the Thai population.

OTHER RECENTLY APPLIED SCIENTIFIC NAME: *Rattus rattus nitidus*.

DISTRIBUTION: mountainous and hilly regions of northern India and Nepal, through northern Myanmar, northern and central Thailand, northern Laos and southern China to Hainan Island and Fukien Province; extending down the central mountain chain of Vietnam. *Rattus nitidus* is also found in four widely scattered localities in island Southeast Asia, presumably as a result of human introductions: Benguet Province, Luzon Island, in the Philippines; central Sulawesi, Indonesia; Seram in Maluku Province, Indonesia; and the Bird's Head Peninsula, Province of Papua, Indonesia. On Seram

Adult measurements	Thailand	Southern Vietnam	Sichuan, China (male)	Sichuan, China (female)
Weight (g)	to 122	–	135.9	114.4
Head+body (mm)	to 177	173–177	163.4	157.6
Tail length (mm)	to 168	168–191	170	166
Pes length (mm)	to 37	35–40	36.0	34.0
Ear length (mm)	to 21	20–24	21.3	21.0

and the Bird's Head Peninsula of Papua, *R. nitidus* has successfully invaded montane forest habitat.

HABITAT USE: abundant in all cropping systems in Sichuan Province, including rice, wheat, maize and potato fields and orchards, but uncommon in associated village habitat where 'R. rattus' is dominant. Results of a mark–recapture study suggest that individuals regularly shift their exploitative focus between habitats, presumably following the availability of food.

In the uplands of Thailand, *R. nitidus* is said to occur exclusively in village houses, where it is trapped in about equal numbers with 'R. rattus' and *R. exulans*. In a village near Luang Prabang in the uplands of northern Laos, small numbers of *R. nitidus* were trapped in irrigated rice fields situated between a village and regrowth forest. At this locality, *R. nitidus* is either rare or absent in the village habitat, which instead supports a high-density population of 'R. rattus'.

NESTING BEHAVIOUR: nothing is reported. *Rattus nitidus* presumably nests in or around houses in upland villages in Thailand, but may dig burrows where it occurs as a field pest in southern China.

BREEDING BIOLOGY: breeding activity in Sichuan Province occurs in all months except December–February. Pregnancy rates peak at 50% in March–April, with a second, more variable peak (25–50%) in August–September. These peaks correspond to periods of crop growth and maturation for wheat and rice, respectively.

Litter size in Sichuan Province ranges from 4–15, with an overall average of 8.25. There is little variation in litter size through the course of the extended breeding season. Captive females can produce four litters per year with an interval between births of 34.7 ± 10.3 days. However, most wild-caught pregnant females have only one set of scars, indicating a lower reproductive output under natural conditions. Females in Gansu Province, China, produce 2–3 litters per year, with litter sizes of 2–7. A wild-caught female from Thailand was reported to have produced and raised a litter of 6.

Captive-born pups from the Chinese population are reported to weigh 7 g at birth, which is very large in comparison with other pest species (e.g. 4.3 –6.2 g in *R. norvegicus*; see Chapter 6, Table 6.1). The pups are weaned after 25–30 days. Males reach sexual maturity at 63–80 days; females considerably later at an average of 119 days. The average life span is estimated at around 12 months for males; slightly less for females.

POPULATION DYNAMICS: *R. nitidus* makes up 67% of rodent captures in Sichuan Province, with average densities through the year estimated at around 3–4 individuals/ha. Peak abundances (around 10/ha) are recorded in May–June (during the ripening phase of wheat) and September–October (just after the rice harvest), reflecting the two major periods of juvenile recruitment at those times.

DAMAGE TO CROPS: levels of damage to crops are not reported for Sichuan Province.

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Fore-limbs of adult *Rattus nitidus* from Laos.

Upper surface of pes; adult from Laos.

Lower surface of pes.

Rattus norvegicus (Berkenhout, 1769)

COMMON NAMES: Norway rat, sewer rat, brown rat

Rattus norvegicus is a major urban pest worldwide and it has successfully invaded many Asian cities and towns. It is also reported as a field pest in scattered locations in Thailand, Vietnam and the Philippines, but generally with lower population densities than other local pest rodents.

MORPHOLOGICAL FEATURES: a large, terrestrial rat with short, grey–brown to plain-brown dorsal fur and a pale-brown or grey belly. Black (melanistic) individuals are moderately common in some populations. The snout is long and broad, and both the eyes and the ears are small. The tail is almost always shorter than the head+body. It is usually weakly 'bicoloured' (dark above, paler below) but sometimes appears mottled or blotched. The pes is proportionally longer and heavier than in other species of *Rattus* and is usually clothed in pure white hairs (but with dark hairs in melanistic individuals).

Rattus norvegicus is sometimes mistaken for species of *Bandicota*. However, the bandicoot rats have larger ears, a darker manus and pes, and broader incisors.

MAMMAE: 1+2+3 (some eastern European populations have 1+1+3).

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus rattus norvegicus, Rattus norvegicus socer.*

DISTRIBUTION: original distribution is thought to be south-eastern Siberia and northern China, but now found on all continents except Antarctica. In South and Southeast Asia and in the wider Pacific region, *R. norvegicus* is most commonly found around ports and the major towns. However, in several areas it appears to be established as an agricultural pest.

TAXONOMIC ISSUES: the name *norvegicus* was originally proposed for the population occupying Great Britain. East Asian populations, including those found in a presumed wild state, are generally referred to the subspecies *R. norvegicus socer*. Because this species is often found around wharves and ships, many local populations may be of mixed origin.

HABITAT USE: a terrestrial, burrowing species with poor climbing skills and often found close to water, such as along rivers and major irrigation canals. It

Adult measurements	Russia	Thailand	Northern Vietnam
Weight (g)	–	to 300	230–510
Head+body (mm)	150–248	to 233	205–260
Tail length (mm)	100–220	to 201	190–250
Pes length (mm)	27–44	to 44	38–50
Ear length (mm)	17–22	to 21	19–26

occurs in many major cities and towns, where it lives in and around buildings and animal yards, feeding on refuse and stored food. Less commonly, it is found in cultivated areas removed from human habitation, including rice fields in Vietnam, Thailand and Luzon Island in the Philippines, and rice–wheat fields in Sichuan Province, southern China.

Rattus norvegicus lives communally and constructs large and complex burrow systems that may be occupied for many years. A typical burrow complex has multiple points of entry and exit, and numerous interconnected tunnels and chambers. Food storing or 'caching' behaviour is reported for this species in North America. Detailed behavioural studies of wild *R. norvegicus* have documented a strongly hierarchical social system in which high-ranked individuals (males and females) enjoy privileged access to food and other resources. This allows them to forage less often and for shorter periods each night, and for high-ranked females to breed at a younger age and with considerably greater success. Behavioural differences related to social rank become especially pronounced under high population densities.

NESTING BEHAVIOUR: nests lined with leaves or other soft material are constructed within the communal burrow complex. Food caches are often added immediately before the weaning of pups— presumably to allow the immature rats to remain longer in the safe, burrow environment.

BREEDING BIOLOGY: in Sichuan Province, breeding activity is restricted to the warmer months (May– October). Year-round breeding is likely in warmer regions, provided adequate food is available.

No litter size estimates are available from Southeast Asia. Elsewhere in the region, a mean litter size of 8.1 is reported for India and 10 (range 5–13) for central Asia. In North America, estimates of mean litter size range from 8.4–9.9, with similar estimates (8.7–9.3) for populations in Europe. Overall pregnancy rates for adult females are estimated at 15% for India, and in the range of 11–29% for North America and 17–31% for Europe. The pregnancy rate peaks at 30% in June in Sichuan Province, China. The gestation period in wild populations is reported as 22–24 days.

In one wild North American population in Baltimore, females entered oestrus for the first time at around 40 days of age, but first conception was often delayed by several months, especially in socially low-ranked females. Females continued to produce litters through to a maximum of 420 days of age, but with longer intervals between litters after the first year. Females generally outlived males, but few individuals lived longer than two years.

In wild European and North American populations, males generally develop scrotal testes at a head+body length of 93–190 mm. Females generally show vaginal perforation at 72–123 mm, but rarely produce their first litter below 180 mm.

POPULATION DYNAMICS: low population densities of 0.5–3 individuals/ha are reported from cropping areas in Sichuan Province. Slight fluctuations in abundance occur through the year, with minor peaks in June and in September–October. Experimental removal of *Rattus nitidus*, the major co-occurring pest species, led to an increase in the abundance of *R. norvegicus*.

The historical introduction of both *R. norvegicus* and *R. rattus* to many different parts of the world has had various outcomes. In Great Britain, *R. norvegicus* arrived later than *R. rattus* and led to the local decline, almost to extinction, of the latter species. The reverse has occurred in New Zealand where *R. norvegicus*, once common and widespread, has been largely displaced by *R. rattus*.

DAMAGE TO CROPS: the level of damage to crops caused specifically by *R. norvegicus* in South and Southeast Asia is not recorded. Elsewhere in the world, the species is responsible for extensive agricultural damage. It is also known to harm domestic fowl.

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Adult *Rattus norvegicus* from Vietnam.

Pes of adult from Vietnam: upper surface (left) and lower surface (right).

Adult specimen from Cambodia.

Adult from Vietnam.

Rattus rattus Complex

COMMON NAMES: house rat, black rat, roof rat

This group or 'complex' includes a number of closely related species that presumably arose in discrete geographical areas but are now intermingled across at least part of their ranges. This has probably resulted in some local interbreeding and gene flow (hence the term 'complex'), and resulted in much confusion over the true number of species in the group. The group probably originated in Southeast Asia and still has its main diversity there. Today, the most widely distributed member of the group is the 'black rat' which spread initially to Europe and from there to many other parts of the world. In many countries, members of the *Rattus rattus* Complex are confined to village or urban habitats. However, in some parts of Southeast and South Asia, these animals are the dominant agricultural rodent pests, causing extreme damage in a wide range of crops, including cereals.

MORPHOLOGICAL FEATURES: medium-size rats that are equally at home on or off the ground. Most populations are highly variable in external appearance. In Asia, the dorsal fur is usually some shade of brown (greyish to reddish). Black individuals are rare in Asia, in contrast to Europe where the 'black rat' is the more typical form. The belly fur in Asian populations is equally variable, with some individuals having pure creamy-white belly fur and others having a grey-based fur with cream to buff tipping. A contrasting chest patch or mid-belly line is quite common, either white against a dark belly, or dark against a white belly. In adults, the dorsal fur is moderately spiny, especially on the flanks. Long, black guard hairs project through the dorsal fur; these are most conspicuous on the lower back. The snout is moderately long and narrow, and the ears are large and thinly furred. The tail is usually slightly longer than the head+body, but in some populations it is either slightly shorter or much longer than the head+body. The tail is always dark above and below, but very occasionally

Adult measurements	India (arid zone)	Bangladesh	Thailand	Northern Vietnam	Malaysia	Sumba Is, Indonesia
Weight (g)		to 213	–	to 219	–	–
Head+body (mm)	138–185	73–225	to 182	105–215	150–205	172–230
Tail length (mm)	163–216	55–234	to 188	120–213	175–231	176–237
Pes length (mm)	22–33	18–39	to 33	26–35	32–39	35–43
Ear length (mm)	19–22	–	to 23	17–23	19–25	22–28

ends in a short, all-white tip. The fur on the upper surface of the manus is dark over the wrist but white on the digits. The pes is moderately broad and has prominent plantar pads, usually with obvious striations. The upper surface of the pes is occasionally pure white, but is more often partially clothed in black or orange hairs. The fur on the toes is white.

MAMMAE: 1+1+3 or 1+2+3; some individuals have contrasting numbers of postaxillary teats on the left and right sides of the body.

Members of the *R. rattus* Complex can be difficult to distinguish from the similar-size pest species *R. tiomanicus*, *R. argentiventer* and *R. nitidus*, and from *R. sikkimensis*—a highly arboreal, forest-dwelling species. In comparison to *R. rattus*:

- *R. tiomanicus* is always white-bellied and adults have 'sleeker' fur with shorter guard hairs that barely project through the fur, even on the lower back
- *R. argentiventer* usually has a conspicuous fringe of orange hairs just forward of the ear, a slightly longer and relatively narrower pes that is more boldly marked with dark hairs, a relatively shorter tail with slightly larger tail scales, more densely furred ears, and a more 'peppered' (orange and black) appearance to the dorsal fur
- *R. nitidus* has darker, woollier fur, a pure-white manus (the white fur often extends partway up the fore-limb), and a relatively long and narrow,

pure-white pes. The tail is often slightly darker above than below (weakly 'bicoloured')

- *R. sikkimensis* has larger plantar pads on the pes, longer and thicker facial vibrissae that extend past the ears when folded back, very prominent guard hairs along the entire length of the back, and a proportionally longer tail. This is a highly arboreal species.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus tanezumi*, *Rattus flavipectus*, *Rattus germaini*, *Rattus molliculus*; also various subspecies within *R. rattus* (e.g. *R. r. alexandrinus*, *R. r. arboreus*, *R. r. diardii*, *R. r. frugivorous*, *R. r. mindanensis*, *R. r. sumbae*).

DISTRIBUTION: members of the *R. rattus* complex are found throughout mainland Southeast and South Asia, including all large and most small islands. They also are widely distributed through the Pacific region, where they were introduced during prehistoric (Micronesia) and historic times (Melanesia and Polynesia).

TAXONOMIC ISSUES: members of the *R. rattus* Complex display a variety of chromosomal rearrangements, some of which result in reduced fertility between hybrids. In some recent works, the names *Rattus rattus* and *Rattus tanezumi* have been used to distinguish the European 'black rat' with 38 chromosomes (*R. rattus*, with possible wild populations in India) from the Asian 'house rats' with 42 chromosomes (*R. tanezumi*). However,

genetic studies currently in progress show two major genetic groups among the Asian house rats, with partially overlapping ranges. Because we are not confident that *R. tanezumi* is the earliest available name for either of the two Asian lineages, we prefer to group them all as the *Rattus rattus* Complex pending completion of a comprehensive taxonomic study. In the following sections, they are referred to collectively as '*R. rattus*'.

HABITAT USE: most commonly found in and around human dwellings, livestock yards and storage facilities. However, in Asia and the Pacific region, '*R. rattus*' also commonly enter gardens and cropping areas, including rice fields. In Indonesia, Malaysia, Vietnam and Cambodia, '*R. rattus*' usually accounts for less than 10% of captures in these habitats, presumably as a consequence of competition with *R. argentiventer*. In areas where *R. argentiventer* is absent, such as in Bangladesh, Laos, parts of Thailand and on many of the Philippine islands, capture rates of '*R. rattus*' in field areas are typically much higher, and it sometimes assumes the role of dominant agricultural pest.

In the uplands of Laos, '*R. rattus*' is the dominant pest in both village and field habitats. The species is also abundant in adjacent forest-edge habitat. A recent radio-tracking study in Luang Prabang Province, undertaken immediately postharvest, found many individuals sheltering in piles of rice straw and cut Job's tear stalks. Others were using

burrows and arboreal nests in trees, located both in field and fallow habitats, and in adjacent forest. Large-scale movements were observed between forest and field habitats, with rats using the forest–field interface for at least some of the more extensive movements. Females with pups were found occupying burrows in the field habitats.

In southern Laos, '*R. rattus*' is said by farmers to dislike entering water. This statement is supported by observations of rice tiller damage only around the edges of flooded fields, adjacent to the bunds. Dry fields in the same area had more extensive damage in patches throughout the crop. This observation is seemingly contradicted by a report from the Philippines of exceptionally high densities of '*R. rattus*' (800–2200 individuals/ha) in a large area of flooded marshland alongside rice paddy. These estimates are based on rats flushed from arboreal leaf nests in this habitat and are presumably reliable. One possible explanation is that the emergent marshland sedges were sufficiently robust for '*R. rattus*' to occupy this habitat without entering the water. Alternatively, the apparent aversion to swimming among Lao '*R. rattus*' may be a peculiarity of that population or member of the species complex.

NESTING BEHAVIOUR: nests are constructed in almost any convenient place. They usually consist of leaves or other dry, soft material drawn together in a bundle and placed in a confined space—in a burrow, among rocks, in a tree hollow or a fallen log,

in the fork of a tree, in the foliage of tall grasses or dense shrubs, inside the stump of a cut banana leaf, in roof thatch, in a wall cavity or inside a mud-brick wall, in a pile of cut wood or brush, in a straw-pile in a harvested field, among stored sacks of grain or jute etc. Numerous different nesting sites are often used within a single population. For example, in one area of northern Laos, different individuals were found raising litters in burrows, in straw piles, in tree hollows, in large, arboreal leaf nests, and in a house roof. Only one litter was ever present in any burrow or nest, so any previous young may disperse upon or before the birth of a new litter; alternatively, the female may herself relocate to a new nest for each successive litter.

BREEDING BIOLOGY: breeding data are available from many parts of the world but most studies refer to urban populations of the European 'black rat'. In South and Southeast Asia, urban or village populations of '*R. rattus*' generally breed more-or-less year-round, probably feeding on refuse and stored food. However, populations living in field habitat generally show cycles of breeding activity and population abundance linked specifically to cropping cycles (see below). For example, in upland areas of Laos, breeding is probably continuous in village habitats but appears to cease altogether during the long dry season in adjacent field habitats.

Estimates of mean litter size fall mostly in the range of 4–8, with the highest mean litter sizes (up to 10)

found in rice-producing regions in the Philippines. Both in the Philippines and in Sekong Province of southern Laos, individual females have been found with as many as 14 live embryos, but such high counts are unusual.

Pregnancy rates averaged across the entire year usually fall around 15–20% of adult females. Individual monthly values typically peak at around 25–30%, but monthly values up to 70% have been recorded in both field and village populations in upland Laos.

Vaginal perforation is reported in individuals as small as 25 g, but many females of 50 g body weight remain imperforate. Pregnancy is recorded in females with body weights as low as 50 g, but first pregnancies are more common above 80–100 g. The gestation period for European '*R. rattus*' is 20–22 days, longer in lactating females. Males typically develop scrotal testes at body weights of 60–100 g.

POPULATION DYNAMICS: in rice-growing areas of the Philippines, the abundance of '*R. rattus*' fluctuates in direct relation to cycles of crop maturation and harvest. A single peak in abundance was reported for Cotabato, Mindanao, with a single, rainfed rice season, but where double cropping is practised in Siniloan, Laguna, two distinct peaks in abundance were observed.

Refuge habitats probably play an important role in population cycles of 'R. rattus' as a field pest. In many agricultural areas, the most likely refugia are villages, where population levels may remain fairly constant through the year. Individuals from these areas presumably colonise the adjacent fields as food and cover for nesting become available.

Upland areas with extensive areas of remnant forest may also act as an important refuge and source of emigrants. Little is yet known about the ecology of 'R. rattus' in such habitats in Asia. However, in various parts of the world including Madagascar, New Zealand and some parts of Indonesia, 'R. rattus' is known to have successfully invaded primary or only minimally disturbed forests. In both Madagasar and in the Galapagos Islands, invasion by 'R. rattus' has apparently precipitated the extinction of native species. In New Zealand, the earlier invader *R. exulans* has been largely displaced from all habitats by the larger and more aggressive 'R. rattus'.

DAMAGE TO CROPS: 'R. rattus' is responsible for major postharvest losses in many countries. Where 'R. rattus' is the dominant field pest, it also causes extensive damage to a wide variety of cereal, vegetable and fruit crops, including coconuts. The rats cut whole tillers at all stages of growth but are also sufficiently agile to climb and directly attack the panicles of mature plants.

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Adult *Rattus rattus* from the uplands of Laos.

Adult from Bangladesh.

Lower surface of pes: adult *Rattus rattus* from Bangladesh.

Lower surface of pes: adult from southern Vietnam.

Adult from the uplands of Laos.

Adult from the uplands of Laos.

Upper surface of pes; adult from Bangladesh.

Upper surface of pes; adult from southern Vietnam.

Adult from the uplands of Laos: note dark fur of fore-limb.

Incisors of adult from Bangladesh.

Juvenile from the uplands of Laos.

Lower surface of manus; adult from Bangladesh.

Rattus sikkimensis (Hinton, 1919)

COMMON NAME: Sikkim rat

Rattus remotus
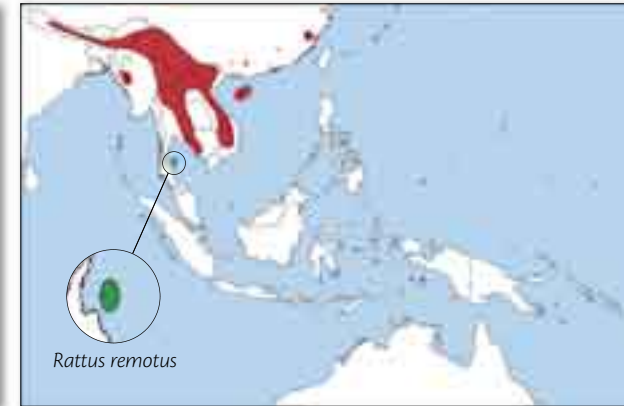
Rattus sikkimensis is widespread in upland forest habitats of mainland Southeast Asia. It is a highly arboreal species and closely resembles *Rattus rattus* in appearance; indeed, the two are often confused. However, unlike the members of the house rat group, *R. sikkimensis* is not known either as a village or as an agricultural pest.

MORPHOLOGICAL FEATURES: a moderately large, arboreal rat, closely resembling white-bellied varieties of the house rat, *Rattus rattus*. It differs from these in a number of features including its larger and more prominent plantar pads, its proportionally longer tail (commonly 20–30 mm longer than the head+body), its larger ears and longer, more inflated snout, and its longer, thicker vibrissae and more prominent guard hairs. The dorsal fur is orange–brown and distinctly shaggy, with conspicuous, black guard hairs all the way down the centre of the back. The belly fur is either pure white or cream in colour, sometimes with a reddish-brown chest patch. The pes is densely furred with a mixture of white and black hairs—the dark hairs often extending to the base of the toes or beyond. The ears are relatively larger than in any other Southeast Asian *Rattus* species and the vibrissae are also exceptional both for their thickness and their length (extending well past the ears when folded back). A short, white tail-tip is reported in 20% of specimens from Hainan Island, southern China. One specimen from Hong Kong had an extensive white patch covering the snout and cheeks.

MAMMAE: always 1+2+3.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus rattus sladeni, Rattus rattus koratensis, Rattus koratensis, Rattus remotus.*

DISTRIBUTION: widely distributed across the upland regions of Southeast Asia, from Nepal in the west, through the Sikkim region of India and northern Myanmar and Laos, to the uplands of northern and central Vietnam in the east. It is recorded from various localities in southern China including Hainan and Hong Kong Islands, north to Fujian Province.

TAXONOMIC ISSUES: *R. sikkimensis* is sometimes treated as a geographical variant of *Rattus remotus*

Adult measurements	Thailand	Laos	Vietnam	Hong Kong
Weight (g)	to 129	–		105–50
Head+body (mm)	to 173	200	to 185	156–200
Tail length (mm)	to 209	212	to 204	185–240
Pes length (mm)	to 33.5	31	to 36	32–37
Ear length (mm)	to 22.5	–	–	23–25

(Robinson & Kloss, 1914), a morphologically similar species found on Koh Samui and some nearby islands in the Gulf of Thailand (see map on previous page). If this were proven correct, *remotus*, being the earlier name, would apply to the entire group.

HABITAT USE: in Thailand, *R. sikkimensis* is said by Marshall (1977) to be "common in evergreen forest of mountains". In Laos, we have trapped the species in upland gardens adjacent to forest, and in clumps of giant bamboo growing along a major river flowing alongside a large village. Despite this close proximity to human activity, no individuals of *R. sikkimensis* were trapped in village houses, perhaps on account of competition from *R. rattus*. In Vietnam, this species (as *R. remotus*) is said by Lunde and Son (2001) to be "often trapped in agricultural areas, scrub habitats and around houses". However, during our work in Vietnam, we have never encountered *R. sikkimensis* in densely settled, lowland agricultural habitat in either of the Mekong Delta in the south or the Red River Delta in the nort.

NESTING BEHAVIOUR: nothing reported.

BREEDING BIOLOGY: nothing reported.

POPULATION DYNAMICS: nothing reported.

DAMAGE TO CROPS: although there are no reports of crop damage, the potential for confusion with *R. rattus* should be kept in mind.

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Adult *Rattus sikkimensis* from Laos.

Subadult from Hong Kong.

Pes of adult from Laos; upper surface (left) and lower surface (right).

Rattus tiomanicus (Miller, 1900)

COMMON NAME: wood rat

This highly arboreal species is found in secondary forests and plantations of the Malay Peninsula and the surrounding islands of the Sunda Shelf. It is a minor pest in gardens and orchards but reaches high densities and causes significant damage in oil palm plantations. In some parts of Malaysia, *R. tiomanicus* is found in villages, but generally only where its close relative, *R. rattus* is absent.

MORPHOLOGICAL FEATURES: a medium-size, arboreal rat with grizzled brown dorsal fur and a pure-white to off-white belly. The snout is short and broad, and the ears are large and thinly furred. The tail is usually slightly longer than the head+body and is dark above and below. Overall, *R. tiomanicus* is very similar in appearance to *R. rattus* but with shorter guard hairs that barely project through the fur. The fur is described as 'sleek' compared with 'coarse' or 'shaggy' in *R. rattus*.

MAMMAE: 1+1+3.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus jalorensis, Rattus rattus jalorensis.*

DISTRIBUTION: the Malay Peninsula and the surrounding Sunda Shelf islands (Sumatra, Borneo, Java, Palawan and many smaller islands). Off the Sunda Shelf, it is found on Bali, Enggano Island (south-west of Sumatra) and the islands of the Maratua Archipelago (east of Borneo).

TAXONOMIC ISSUES: some morphologically distinct island populations are currently recognised as subspecies (e.g. *R. tiomanicus mara* of the Maratau Archipelago).

HABITAT USE: primarily arboreal and said to feed mainly on fruits. In oil palm plantations,

R. tiomanicus often shelters in piles of cut fronds and, less frequently, in cut stumps or fallen logs. Very occasionally it is found in terrestrial burrows, but these are probably excavated by other species. Individuals generally have small home ranges, consisting of one or a few adjacent palms. Occasional, larger-scale movements are undertaken to establish new feeding areas.

NESTING BEHAVIOUR: nests are said to be off the ground, presumably in crowns of palms and in hollow stumps and logs.

Adult measurements	Malaysia
Weight (g)	91 ± 34
Head+body (mm)	154–176
Tail length (mm)	155–198
Pes length (mm)	27–35
Ear length (mm)	16–22

BREEDING BIOLOGY: studies in a Malay oil palm plantation found year-round breeding. Litter size ranged from 2–7, with a mean of 4.4. The average female pregnancy rate, taken over several complete years, was 17.6%, or 27.9% if restricted to sexually mature individuals.

Males show rapid testicular enlargement at a body weight of around 60 g. Females show vaginal perforation from body weights below 30 g, but a few are still imperforate at 50 g. The smallest recorded pregnant female weighed 65 g.

POPULATION DYNAMICS: estimates of population density in unbaited oil palm plantations range from 183–539/ha, with an average of 306/ha.

DAMAGE TO CROPS: *R. tiomanicus* damages the maturing and ripe fruit of the oil palm, and can cause losses of up to 5% in oil production if uncontrolled.

KEY REFERENCES:

Harrison, J.L. 1952. Reproduction in rats of the subgenus *Rattus*. Proceedings of the Zoological Society of London, 121, 673–694.

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Adult *Rattus tiomanicus* from peninsular Thailand (museum specimen).

Rattus turkestanicus (Satunin, 1903)

COMMON NAME: Turkestan rat

Rattus turkestanicus is an important field pest in parts of south-eastern China. In the western part of its range, it is known mainly as a forest rat.

MORPHOLOGICAL FEATURES: a medium-size, primarily terrestrial rat with reddish-brown dorsal fur and a tail that is approximately the same length as the combined length of the head+body. In the western part of its range, there are two colour 'forms' that may represent distinct subspecies or species. The 'typical' form of *R. turkestanicus* has a yellowish-white belly and a weakly 'bicoloured' (dark above, paler below) tail. The snout is short and broad, and the ears are relatively small and densely furred with a mixture of white and dark hairs. In the *vicerex* form, the belly fur is grey-based with cream tipping, the fur on the manus and pes is pure white, the ears are larger and the tail is strongly bicoloured. The eastern population most closely resembles typical *turkestanicus*.

MAMMAE: 1+2+3.

Overall, this species most closely resembles *Rattus norvegicus* but adults are smaller and have a relatively longer and more densely furred tail.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES: *Rattus rattoides*, *Rattus vicerex*.

DISTRIBUTION: highlands of the Middle East to Central Asia, extending along the southern flanks of the Himalayan massif, including parts of northern India, Nepal and south-western China (Yunnan Province). A possibly isolated population occurs in south-eastern China (Guandong, Xiamen and Fujian Provinces).

TAXONOMIC ISSUES: the south-eastern Chinese populations of this species are generally reported as *R. rattoides*. It is unclear whether these populations are continuous with typical *R. turkestanicus* of the Central Asian highlands. There is some variation in chromosome morphology among the western populations but it is unclear how this relates to the morphological variations noted above.

HABITAT USE: in the western part of its range, this species occupies natural forests at moderate to high altitudes. To the east, the species occupies the major

Adult measurements	Turkestan
Weight (g)	100–200
Head+body (mm)	168–215
Tail length (mm)	167–213
Pes length (mm)	31–38
Ear length (mm)	19–25

rice-producing area of the South China Plain, along the lower reaches of the Yangtze and Pearl Rivers. Here, *R. turkestanicus* is the dominant agricultural rodent pest, making up 62–84% of trap captures in rice-field habitat.

NESTING BEHAVIOUR: nests are located in burrows. Areas with dense groundcover are preferred as burrow sites, with burrow densities averaging almost 50 holes per 100 m transect through dense (>60%) groundcover. In rice fields, burrows are located in bunds and are most numerous along weedy bunds.

BREEDING BIOLOGY: in Guandong and Fujian Provinces, there are two peaks in breeding activity, in June and October—both linked to periods of rice maturation. The lowest pregnancy rate occurs during winter (December–January). Two peaks are also reported in Xiamen Province, but in March–May and August–October.

Mean litter size (from embryo counts) is 6.78 (range 2–11) in the Pearl River Delta and 7.0 (range 4–11) in Fujian Province. In both areas, litter size varies seasonally, with slightly higher litters in late winter–autumn (August–October) than in spring (March–May).

POPULATION DYNAMICS: in Fujian Province, high densities occur in November–December through to April, with a dramatic decline during May. In Xiamen Province, population densities remain fairly stable in the irrigated rice fields, but fluctuate markedly in vegetable and dry-land plots. Seasonal migration is reported in Guandong Province, with rats moving from the rice fields to nearby orange and banana plantations after each harvest.

DAMAGE TO CROPS: in southern China, the greatest damage is inflicted on early ripening rice crops, with reported yield losses of 5.3%. Damage is also reported to vegetables and fruits (oranges, bananas) but it is unclear whether damage is inflicted to fruiting bodies (which would imply some climbing ability) or to the plants themselves.

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Adult *Rattus turkestanicus* (*vicerex* form) from Nepal (museum specimen).

Adult *Rattus turkestanicus* (*vicerex* form) from Nepal: dorsal view (left) and ventral view (right) (museum specimen).

Adult ('typical' form) from Nepal: dorsal view (left) and ventral view (right) (museum specimen).
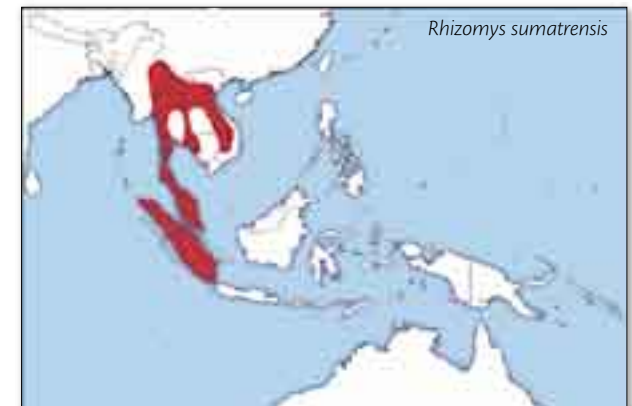
Rhizomys species

COMMON NAME: bamboo rats

Rhizomys pruinosus

Rhizomys sinensis

Rhizomys sumatrensis

Bamboo rats of the genus *Rhizomys* are widespread across the upland regions of northern Myanmar to southern China. They are moderately large to huge, stockily built animals with obvious adaptations to fossorial life and their diet of bamboo rhizomes and shoots. Although their burrows are sometimes located in slash-and-burn gardens, it is unclear how much damage they inflict on crops. There are some reports of bamboo rats damaging sugarcane and cassava.

MORPHOLOGICAL FEATURES: all species share a common body form, with a massively broadened head, a plump body with short limbs, strong claws on both the pes and manus, small eyes and ears, and a short, sparsely haired tail that lacks scales (it is instead covered in soft, wrinkled skin).

Rhizomys species have grey–brown to dull orange–brown fur, rounded ears that just project through the fur, and granulated plantar pads on the manus and

Adult measurements	*R. pruinosus* Thailand	*R. pruinosus* Vietnam	*R. sinensis* China	*R. sumatrensis* Thailand
Weight	–	645–690 g	–	2–4 kg
Head+body (mm)	256–350	280–290	230–450	280–480
Tail length (mm)	100–124	108–120	50–90	102–200
Pes length (mm)	45–61	47–48	35–60	46–67
Ear length (mm)	18–26	18–21	11–20	18–36

pes (compared to smooth pads in *Cannomys badius*). *Rhizomys sumatrensis* (Raffles, 1821) grows to a much greater size than the other species, the tail is relatively longer, and the top of the head bears a distinctive, triangular patch of dark fur. The two posterior plantar pads on the pes of *R. sumatrensis* are united (compared to separate in other *Rhizomys* spp.). *Rhizomys pruinosus* Blyth, 1851 is a smaller species with a shorter tail and numerous white-tipped hairs that gives the fur a frosted appearance. *Rhizomys sinensis* Gray, 1831 is similar to *R. pruinosus* but has lush, pale-brown fur that lacks any frosting. The top and sides of the face in *R. sinensis* are darker than the back or flanks.

MAMMAE: 1+1+3 for all three species;
  1+0+3 in some *R. pruinosus*.

OTHER RECENTLY APPLIED SCIENTIFIC NAMES:
of *Rhizomys pruinosus* = *Rhizomys pannosus*
of *Rhizomys sinensis* = *Rhizomys vestitus*,
  Rhizomys senex
of *Rhizomys sumatrensis* = *Nyctocleptes sumatrensis*.

DISTRIBUTION: *R. sumatrensis* is found in the uplands of eastern Myanmar, western, central and peninsular Thailand, south-western Cambodia, Laos and Vietnam. It occurs across a wider elevational range in peninsular Malaysia and on the island of Sumatra, Indonesia. *Rhizomys pruinosus* has a similar distribution to *R. sumatrensis* but it extends further west into Assam, India, and north to cover large

parts of southern China. It is absent from Sumatra. *Rhizomys sinensis* occurs in the uplands of southern China and northern Vietnam, through to northern Myanmar. It has not yet been recorded in Laos but might be expected in the northern provinces.

TAXONOMIC ISSUES: each species shows geographical variation and subspecies are sometimes recognised. The bamboo rats as a whole are in urgent need of revision, especially on account of the heavy exploitation of these species in some areas as commercial food items.

HABITAT USE: bamboo rats are probably most abundant in the natural bamboo forests that still cover large areas of the uplands of Southeast Asia. Their presence in such areas is usually obvious from their large, poorly concealed burrow systems in which they shelter through the day. In Malaysia, *R. sumatrensis* emerges in the early evening and roams widely within the bamboo thickets, feeding on fallen fruit and other herbivorous matter. This species also has the habit of climbing bamboo culms to cut out sections of woody stem; these are carried back to the burrows for unknown purpose.

NESTING BEHAVIOUR: the burrow systems of *Rhizomys* spp. are possibly more complex than those of *Cannomys badius* but no details are available.

BREEDING BIOLOGY: litter size is reported as 3–5 in *R. sumatrensis*, which has a gestation period of "at least 22 days".

The young of *R. sumatrensis* grow hair at about 10–13 days, open their eyes at 24 days and are weaned over an extended period from 1–3 months after birth. Life span in captivity is about 4 years.

POPULATION DYNAMICS: nothing known.

DAMAGE TO CROPS: damage to sugarcane and cassava has been reported.

KEY REFERENCE:
Marshall, J.T. 1977. Family Muridae. In: Lekagul, B. and McNeely, J.A., ed., Mammals of Thailand. Bangkok, Kurusapha Press, 397–487.

Tail of adult *Rhizomys pruinosus* from northern Vietnam, showing absence of scales and very sparse fur.

Adult *Rhizomys pruinosus* from Laos.

Adult *Rhizomys sinensis* from China (museum specimen).

Adult *R. pruinosus* from northern Vietnam.

Pes of adult *R. pruinosus*; specimens from northern Vietnam (left) and Laos (right).

Adult *Rhizomys sumatrensis* from Malaysia (museum specimen).

# Key to the pest rodents of South and Southeast Asia and the Pacific

1a. Tail covered with soft skin, lacking obvious scales **2**

1b. Tail covered with scales, arranged in rings **5**

2a. Plantar pads on pes with smooth surfaces; ears completely hidden in fur . . . . *Cannomys badius*

2b. Plantar pads on pes with granular surfaces; ears projecting through fur **3**

3a. Inner and outer metatarsal tubercles on pes fused; adult body size can exceed 1.5 kg . . . . *Rhizomys sumatrensis*

3b. Inner and outer metatarsal tubercles on pes separate; adult body size not exceeding 1.5 kg **4**

4a. Fur on back and flanks plain gingery-brown, lacking projecting guard hairs . . . . *Rhizomys sinensis*

4b. Fur on back and flanks grey with long, silvery tipped guard hairs . . . . *Rhizomys pruinosus*

5a. Head+body length less than 110 mm **6**

5b. Head+body length more than 110 mm **12**

6a. Inner and outer metatarsal tubercles (IMT, OMT) on pes of nearly equal size (diagram A, below); females with mammae 1+2+2 . . . . **7**

6b. Inner metatarsal tubercle elongated, much larger than outer metatarsal tubercle (diagram B, below); specimen is juvenile (proceed with extreme caution!) **12**
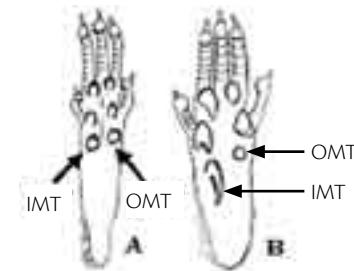
7a. Inner metatarsal tubercle positioned close to outer metatarsal tubercle (as in diagram A, above) **8**

7b. Inner and outer metatarsal tubercles widely separated (gap between them far exceeds diameter of either pad) **11**

8a. Fur very stiff due to presence of numerous, broad spines; tail weakly bicoloured . . . . *Mus cookii*

8b. Fur soft, without spines or with few, narrow spines; tail distinctly bicoloured **9**

9a. Plantar pads on pes distinctly raised; tail usually longer than head+body . . . . *Mus caroli*

9b. Plantar pads on pes low; tail usually shorter than head+body **10**

10a. Belly fur pure white; tail usually 10 mm or more shorter than head+body . . . . ***Mus booduga***

10b. Belly fur grey-based with white or cream tipping; tail usually less than 10 mm shorter than head+body . . . . ***Mus terricolor***

11a. Tail distinctly bicoloured and usually 10 mm shorter than head+body . . . . ***Mus cervicolor***

11b. Tail uniformly dark or weakly bicoloured and usually longer than head+body . . . . ***'Mus musculus'***

12a. Upper incisors in adult greater than 3.5 mm in combined width across tips; vibrissae on sides of snout short (barely reach ears when folded back) **13**

12b. Upper incisors in adult less than 3.5 mm in combined width; vibrissae on sides of snout long (overlapping ears when folded back) **16**

Upper incisor width
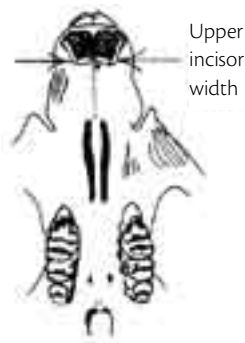
13a. Upper incisors wider than lower incisors; inner metatarsal tubercle elongated; tail with conspicuous hairs **14**

13b. Upper incisors narrower than lower incisors; inner metatarsal tubercle rounded; tail hairs indistinct . . . . ***Nesokia indica***

14a. Upper incisors projecting forward (diagram A, below); head+body usually 20 mm or more longer than tail; females with at least seven mammae on each side . . . . ***Bandicota bengalensis***

14b. Upper incisors curving backward (diagram B, below); head+body usually less than 20 mm longer than tail; females with six mammae on each side **15**

Upper incisors
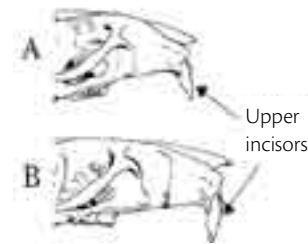
15a. Pes longer than 40 mm, even in juveniles; ear longer than 26 mm in adults; adult body weight commonly exceeding 350 g . . . . ***Bandicota indica***

15b. Pes usually shorter than 40 mm; ear not exceeding 26 mm; adult body weight usually less than 350 g . . . . ***Bandicota savilei***

16a. Specimen is adult female (able to accurately count mammae) **17**

16b. Specimen is male or young female (unable to count mammae) **33**

17a. Pectoral teat present **18**
17b. Pectoral teat absent **31**

Pectoral teat
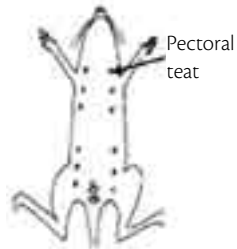
18a. Two inguinal teats on each side **19**
18b. Three inguinal teats on each side **21**

Two inguinal teats
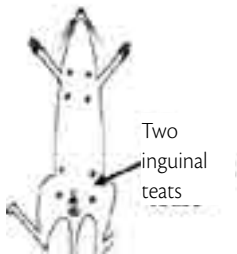
19a. Two postaxillary teats on each side . . . . **Berylmys berdmorei**
19b. One postaxillary teat on each side **20**

Two postaxillary teats
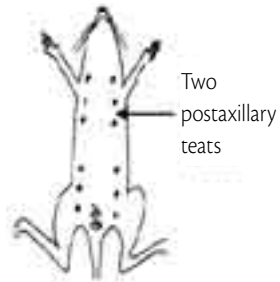
20a. Belly fur grey-based; pes shorter than 30 mm;
incisors with orange or yellow enamel . . . . **Rattus exulans**
20b. Belly fur pure white; pes longer than
30 mm; incisors with white or pale yellow enamel . . . . **Berylmys bowersi**

21a. One postaxillary teat on each side **22**
21b. Two postaxillary teats on each side **24**

22a. Tail slightly shorter or longer than head+body;
fur on back and flanks with obvious spines **23**
22b. Tail 15 mm or more shorter than head+body;
fur on back and flanks soft, lacking obvious spines . . . . **Rattus losea**

23a. Guard hairs conspicuous on lower back; belly
fur pure white to cream or grey-based . . . . **Rattus rattus**
23b. Guard hairs barely visible on lower back; belly
fur always pure white or cream . . . . **Rattus tiomanicus**

24a. Belly fur distinctly grey-based, with cream or
buff tips **25**
24b. Belly fur pure white, cream or silvery grey **27**

25a. Large rat, adult body size commonly exceeding
250 g; pes length usually exceeding 40 mm, even
in juveniles . . . . **Rattus norvegicus**
25b. Smaller rat, adult body size rarely exceeding 250 g;
pes length usually less than 40 mm **26**

26a. Fur very soft; manus and fore-limb covered in white fur; tail weakly bicoloured ....*Rattus nitidus*

26b. Fur with obvious spines; fore-limb covered in dark fur, contrasting with manus; tail uniformly dark ....*Rattus rattus*

27a. Tail bicoloured (darker above than below) **28**

27b. Tail uniformly dark above and below **29**

28a. Large rat, adult body size commonly exceeding 250 g; pes usually longer than 40 mm, even in juveniles .... *Rattus norvegicus*

28b. Smaller rat, adult body size rarely exceeding 200 g; pes usually shorter than 40 mm .... *Rattus turkestanicus*

29a. Fringe of orange fur just forward of ear; tail usually shorter than head+body .... *Rattus argentiventer*

29b. No fringe of orange fur just forward of ear; tail usually longer than head+body **30**

30a. Vibrissae on sides of snout reach ears when folded back; all plantar pads on pes moderately large but well-separated .... *Rattus rattus*

30b. Vibrissae on sides of snout extend beyond ears when folded back; all plantar pads on pes very large and close together .... *Rattus sikkimensis*

31a. Fur on back and flanks with obvious broad spines **32**

31b. Fur on back and flanks lacking obvious spines .... *Rattus steini*

32a. Fur on back with numerous projecting guard hairs; belly fur grey or ivory .... *Rattus praetor*

32b. Fur on back without projecting guard hairs; belly fur cream .... *Rattus mordax*

33a. Tail bicoloured (darker above than below) **34**

33b. Tail uniformly dark above and below **38**

34a. Large rat, adult body size commonly exceeding 250 g; pes usually longer than 40 mm, even in juveniles **35**

34b. Smaller rat, adult body size rarely exceeding 200 g; pes shorter than 40 mm **36**

35a. Tail shorter than head+ body; belly fur grey or brown; incisors with yellow or orange enamel .... *Rattus norvegicus*

35b. Tail longer than head+body; belly fur cream or white; incisors with white or pale yellow enamel ...... *Berylmys bowersi*

36a. Belly fur pure cream or yellowish-white colour **37**

36b. Belly fur distinctly grey-based, with cream or buff tipping .... *Rattus nitidus*

37a. Dorsal fur plain grey; incisor enamel cream or white .... *Berylmys berdmorei*

37b. Dorsal fur brown or reddish brown; incisor enamel yellow or orange .... *Rattus turkestanicus*

38a. Belly fur distinctly grey-based, with cream or buff tips **39**

38b. Belly fur pure white, cream or silvery grey **44**

39a. Large rat, adult body size commonly exceeding 250 g; pes usually longer than 40 mm, even in juveniles . . . . *Rattus norvegicus*

39b. Smaller rat, adult body size rarely exceeding 250 g; pes shorter than 40 mm **40**

40a. Fur on back and flanks very soft, lacking obvious spines; tail equal in length or shorter than head+body **41**

40b. Fur on back and flanks with obvious spines; tail equal in length or longer than head+body **43**

41a. Manus and pes covered with pure white hairs; fur on lower fore-limb also pure white, forming long 'glove' . . . . *Rattus nitidus*

41b. Manus and pes with a few to many dark hairs; fur on lower fore-limb dark **42**

42a. Dorsal fur reddish-brown; specimen from Melanesia . . . . *Rattus steini*

42b. Dorsal fur grey–brown to reddish–brown; specimen from mainland Southeast Asia . . . . *Rattus losea*

43a. Pes not longer than 30 mm, even in adult . . . . *Rattus exulans*

43b. Pes usually longer than 30 mm, even in juveniles . . . . *'Rattus rattus'*

44a. Large rat, adult body size commonly exceeding 250 g; pes usually longer than 40 mm, even in juveniles . . . . *Rattus norvegicus*

44b. Smaller rat, adult body size rarely exceeding 250 g; pes shorter than 40 mm **45**

45a. Tail much shorter (at least 15 mm) than head+body **46**

45b. Tail slightly shorter than or longer than head+body **48**

46a. Fur on lower back and flanks soft, lacking obvious spines . . . . *Rattus losea*

46b. Fur on lower back with obvious spines **47**

47a. Fur on lower back of adult with many long guard hairs . . . . *Rattus praetor*

47b. Fur on lower back of adult with few projecting guard hairs . . . . *Rattus mordax*

48a. Most vibrissae on sides of snout extending beyond ears when folded back; all plantar pads on pes very large and close together . . . . *Rattus sikkimensis*

48b. Most vibrissae on sides of snout not extending beyond ears when folded back; all plantar pads on pes well-separated **49**

49a. Fringe of orange fur just forward of ear; tail usually 10 mm or so shorter than head+body . . . . . . *Rattus argentiventer*

49b. No fringe of orange fur just forward of ear; tail usually equal in length or longer than head+body **50**

50a. Fur on lower back of adult with many long, projecting guard hairs . . . . *Rattus rattus*

50b. Fur on lower back of adult with few projecting guard hairs . . . . *Rattus tiomanicus*

APPENDIXES

APPENDIX 1: Trapping data sheet and coding system

TRAPPING DATA SHEET

Site Name:	Census No.:	District:	Date:	Name of Trapper & Data Recorder:	Entered by:	Verified by:	Page No: /

Crop Stage:	Water Depth:	Recent Farmer Control Activities:

CEN	YR	JUL	SITE	LINE	TRP	HAB	CS	RAT	SP	SX	V	T	P	TAIL LTH	EAR LTH	FOOT LTH	Total WGT	BAG	WGT	CL	CN	FA	COMMENTS

COMMENTS:

Codes for trapping data sheet

CEN   **census number**

YR   **year** (e.g. 2003 or 2004)

JUL   **Julian date** (day of the year—see Appendix 3)

SITE   **site number**

LINE   **trap-line number**
1 = field
2 = channel/bank
3 = sugarcane
4 = village

TRP   **trap number**

HAB   **habitat code**
1 = local variety rice crop
2 = improved variety rice crop
3 = sugarcane
4 = groundnut
5 = sunflower
6 = mung bean
7 = black gram
8 = vegetable
9 = village (house)
10 = village (stores)
11 = village (garden)

CS   **crop stage of rice**
1 = fallow
2 = tillering
3 = booting
4 = flowering
5 = harvesting
6 = stubble

RAT   **rat number** (individual ear-tag number)

SP   **species number***
1 = *Rattus argentiventer*
2 = *Rattus rattus* (European)
3 = *Rattus rattus* (Asian)
4 = *Rattus norvegicus*
5 = *Rattus exulans*
6 = *Bandicota indica*
7 = *Bandicota bengalensis*
8 = *Bandicota savilei*
9 = *Mus cervicolor*
10 = *Mus caroli*
11 = *Mus booduga*
12 = *Suncus murinus*
13 = other species (write species in comments)

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*   This is an example. A list such as this would need to be compiled for each country.

SX **sex**
  1 = male
  2 = female
  −1 = not determined

V **vagina**
  1 = not open (membrane intact)
  2 = open (membrane broken)

T **teats**
  1 = barely visible—never lactated before
  2 = prominent but not currently lactating
  3 = prominent and currently lactating

P **pregnancy** (by feeling)
  1 = no or not sure
  2 = pregnant

TAIL LTH **tail length**
  length (mm) from middle of anus to tip of tail (−1 = tail with tip lost)

EAR LTH **ear length**
  length (mm) from tip of ear to base of cartilage

FOOT LTH **pes (hind-foot) length**
  length (mm) from tip of longest toe to heel

TOTAL WGT **weight (g) of bag + rat**

BAG **weight of the bag without the rat (g)**

WGT **weight of the rat (g)**

CL **capture class**
  1 = first capture
  2 = recapture within current census
  3 = recapture from previous census
  4 = recapture but tag lost

CN **capture number**
  the number of times has the animal been caught over all censuses

FA **fate of the animal**
  1 = released at site of capture
  2 = died in trap
  3 = escaped without tag
  4 = taken to laboratory
  5 = taken as voucher specimen
  (write voucher number in comments)

COMMENTS any observation about the rat, trap or change in normal procedure
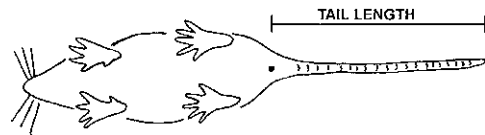
Measure tail length from the middle of the anus to the tip of the tail.
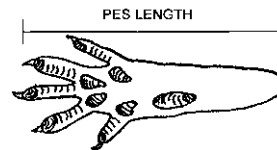
Measure the pes length from the base of the heel to the end of the toe pad on the longest toe (not including the claw).
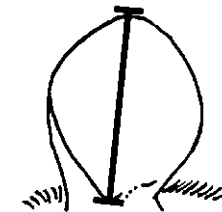
Measure the length of the ear from the bottom of the ear notch to the furthest point along the rim.

APPENDIX 2:   Breeding data sheet and coding system

BREEDING DATA SHEET

Site Name:	Census No.:	District:	Date:	Name of Trapper & Data Recorder:	Entered by:	Verified by:	Page No.: /

CEN	YR	JUL	SITE	HAB	CS	RAT	SP	SX	V	T	P	TAIL LTH	EAR LTH	FOOT LTH	HB	WGT	EMB	E STAGE	SCAR	UT STAGE	VN	Comments

Codes for breeding data sheet

CEN **census number**

YR **year** (e.g. 2003 or 2004)

JUL **Julian date** (day of the year—see Appendix 3)

SITE **site number**

HAB **habitat code**
  1 = local variety rice crop
  2 = improved variety rice crop
  3 = sugarcane
  4 = groundnut
  5 = sunflower
  6 = mung bean
  7 = black gram
  8 = vegetable
  9 = village (house)
  10 = village (stores)
  11 = village (garden)

CS **crop stage of rice**
  1 = fallow
  2 = tillering
  3 = booting
  4 = flowering
  5 = harvesting
  6 = stubble

RAT **rat number** (individual ear-tag number)

SP **species number***
  1 = *Rattus argentiventer*
  2 = *Rattus rattus* (European)
  3 = *Rattus rattus* (Asian)
  4 = *Rattus norvegicus*
  5 = *Rattus exulans*
  6 = *Bandicota indica*
  7 = *Bandicota bengalensis*
  8 = *Bandicota savilei*
  9 = *Mus cervicolor*
  10 = *Mus caroli*
  11 = *Mus booduga*
  12 = *Rattus losea*
  13 = *Rattus tiomanicus*
  14 = *Rattus nitidus*
  15 = *Suncus murinus*
  16 = other species

SX **sex**
  1 = male
  2 = female
  −1 = not determined

V **vagina**
  1 = not open (membrane intact)
  2 = open (membrane broken)

T **teats**
  1 = barely visible—never lactated before
  2 = visible but with fur at base—not currently lactating but has lactated previously
  3 = visible and obvious, with no fur at base—currently lactating

P **pregnancy** (by feeling)
  1 = no or not sure
  2 = pregnant

---

* This is an example. A list such as this would need to be compiled for each country.

TAIL LTH   **tail length**
length (mm) from middle of anus to tip of tail (−1 = tail with tip lost)

EAR LTH   **ear length**
length (mm) from tip of ear to base of cartilage

FOOT LTH   **pes (hind-foot) length**
length (mm) from tip of longest toe to heel

HB   **head+body length**
length (mm) from tip of nose to middle of anus measured with the animal of its back

WGT   **weight of the rat (g)**

EMB   **number of embryos in uterus**

E STAGE   **embryo stage**
1 = first trimester (early pregnancy)
2 = second trimester (mid pregnancy)
3 = third trimester (late pregnancy)

SCAR   **number of SETS of scars in the uterus**
(if you can count the number of scars in any set, write the number in the comments column)

UT STAGE   **condition of uterus**
1 = very thin (like a thread) and with indistinct blood supply
2 = thin (like a string) but with distinct blood supply
3 = thick but not pregnant
4 = with embryos

VN   **voucher number**
number attached to voucher specimen

COMMENTS   any observation about the rat, trap or change in normal procedure
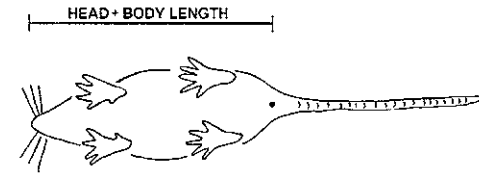
Measure the head+body length along the spine of the rodent from the tip of the nose to the middle of the anus.
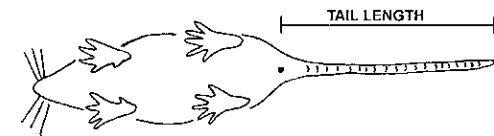
Measure tail length from the middle of the anus to the tip of the tail.
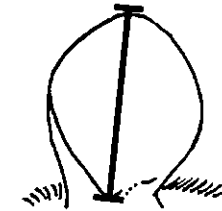
Measure the length of the ear from the bottom of the ear notch to the furthest point along the rim.
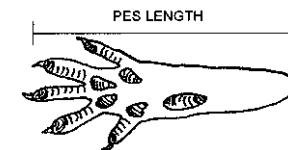
Measure the pes length from the base of the heel to the end of the toe pad on the longest toe (not including the claw).

Appendix 3: Tables of Julian dates

Julian dates for non-leap years and leap years

NON-LEAP YEAR

Date	1 Jan	2 Feb	3 Mar	4 Apr	5 May	6 Jun	7 Jul	8 Aug	9 Sep	10 Oct	11 Nov	12 Dec
1	1	32	60	91	121	152	182	213	244	274	305	335
2	2	33	61	92	122	153	183	214	245	275	306	336
3	3	34	62	93	123	154	184	215	246	276	307	337
4	4	35	63	94	124	155	185	216	247	277	308	338
5	5	36	64	95	125	156	186	217	248	278	309	339
6	6	37	65	96	126	157	187	218	249	279	310	340
7	7	38	66	97	127	158	188	219	250	280	311	341
8	8	39	67	98	128	159	189	220	251	281	312	342
9	9	40	68	99	129	160	190	221	252	282	313	343
10	10	41	69	100	130	161	191	222	253	283	314	344
11	11	42	70	101	131	162	192	223	254	284	315	345
12	12	43	71	102	132	163	193	224	255	285	316	346
13	13	44	72	103	133	164	194	225	256	286	317	347
14	14	45	73	104	134	165	195	226	257	287	318	348
15	15	46	74	105	135	166	196	227	258	288	319	349
16	16	47	75	106	136	167	197	228	259	289	320	350
17	17	48	76	107	137	168	198	229	260	290	321	351
18	18	49	77	108	138	169	199	230	261	291	322	352
19	19	50	78	109	139	170	200	231	262	292	323	353
20	20	51	79	110	140	171	201	232	263	293	324	354
21	21	52	80	111	141	172	202	233	264	294	325	355
22	22	53	81	112	142	173	203	234	265	295	326	356
23	23	54	82	113	143	174	204	235	266	296	327	357
24	24	55	83	114	144	175	205	236	267	297	328	358
25	25	56	84	115	145	176	206	237	268	298	329	359
26	26	57	85	116	146	177	207	238	269	299	330	360
27	27	58	86	117	147	178	208	239	270	300	331	361
28	28	59	87	118	148	179	209	240	271	301	332	362
29	29		88	119	149	180	210	241	272	302	333	363
30	30		89	120	150	181	211	242	273	303	334	364
31	31		90		151		212	243		304		365

LEAP YEAR

Date	1 Jan	2 Feb	3 Mar	4 Apr	5 May	6 Jun	7 Jul	8 Aug	9 Sep	10 Oct	11 Nov	12 Dec
1	1	32	61	92	122	153	183	214	245	275	306	336
2	2	33	62	93	123	154	184	215	246	276	307	337
3	3	34	63	94	124	155	185	216	247	277	308	338
4	4	35	64	95	125	156	186	217	248	278	309	339
5	5	36	65	96	126	157	187	218	249	279	310	340
6	6	37	66	97	127	158	188	219	250	280	311	341
7	7	38	67	98	128	159	189	220	251	281	312	342
8	8	39	68	99	129	160	190	221	252	282	313	343
9	9	40	69	100	130	161	191	222	253	283	314	344
10	10	41	70	101	131	162	192	223	254	284	315	345
11	11	42	71	102	132	163	193	224	255	285	316	346
12	12	43	72	103	133	164	194	225	256	286	317	347
13	13	44	73	104	134	165	195	226	257	287	318	348
14	14	45	74	105	135	166	196	227	258	288	319	349
15	15	46	75	106	136	167	197	228	259	289	320	350
16	16	47	76	107	137	168	198	229	260	290	321	351
17	17	48	77	108	138	169	199	230	261	291	322	352
18	18	49	78	109	139	170	200	231	262	292	323	353
19	19	50	79	110	140	171	201	232	263	293	324	354
20	20	51	80	111	141	172	202	233	264	294	325	355
21	21	52	81	112	142	173	203	234	265	295	326	356
22	22	53	82	113	143	174	204	235	266	296	327	357
23	23	54	83	114	144	175	205	236	267	297	328	358
24	24	55	84	115	145	176	206	237	268	298	329	359
25	25	56	85	116	146	177	207	238	269	299	330	360
26	26	57	86	117	147	178	208	239	270	300	331	361
27	27	58	87	118	148	179	209	240	271	301	332	362
28	28	59	88	119	149	180	210	241	272	302	333	363
29	29	60	89	120	150	181	211	242	273	303	334	364
30	30		90	121	151	182	212	243	274	304	335	365
31	31		91		152		213	244		305		366

APPENDIX 4: Cereal crop damage data sheet and example of calculations

CEREAL CROP DAMAGE DATA SHEET

Crop type:_____

Transect No.	Site Name:	District:	Date:	Name of Data Recorder:	Entered by:	Verified by:	Page No: ....../......

Distance	Number of tillers	Hill Number										Total
		1	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**10**	**Total**
Edge of field	Cut tillers (damaged)											
	With mature grain (undamaged)											
	With growth but not mature (short)											
	Total tillers											
20% in	Cut tillers (damaged)											
	With mature grain (undamaged)											
	With growth but not mature (short)											
	Total tillers											
30% in	Cut tillers (damaged)											
	With mature grain (undamaged)											
	With growth but not mature (short)											
	Total tillers											
40% in	Cut tillers (damaged)											
	With mature grain (undamaged)											
	With growth but not mature (short)											
	Total tillers											
Centre of field	Cut tillers (damaged)											
	With mature grain (undamaged)											
	With growth but not mature (short)											
	Total tillers											

Example of calculations

Example of rat damage to rice crop in Vietnam, where there were 72 rat damaged tillers out of 1569 tillers (from 200 plants):

Size of field in square metres $(N) = 2000$

Area of one set of transect samples (40 plants) in m² (size of stratum h $(N_h)) = 0.50$

Total area in units of samples $= 2000/0.50 = 4000$

Strata	Number of the sampled tillers damaged by rodents								Average proportion (Damgd/n_h=$\hat{p}_h$)	Stratum size (N_h)
	Transect 1		Transect 2		Transect 3		Transect 4			
	Damgd	Total	Damgd	Total	Damgd	Total	Damgd	Total		
Edge of field	3	116	1	68	2	60	1	62	7/306 = 0.0229	800
20% in	5	97	2	83	4	85	5	62	16/327 = 0.0489	800
30% in	7	78	5	58	1	73	9	68	22/277 = 0.0794	800
40% in	3	86	2	49	5	98	1	84	11/317 = 0.0347	800
Centre of field	4	112	1	89	6	68	5	73	16/342 = 0.0468	800

Damgd = Number of damaged tillers.

Estimated mean proportion damaged averaged over all strata $(\hat{p}_{ST})$:

= sum of stratum size × average proportion

$$= \frac{(0.0229 \times 800) + (0.0489 \times 800) + (0.0794 \times 800) + (0.0347 \times 800) + (0.0468 \times 800)}{4000}$$

$$= 0.0465$$

The stratifed mean proportion damaged by rodents $(\hat{p}_{ST})$ is given by:

$$\hat{p}_{ST} = \frac{\sum N_h \hat{p}_h}{N}$$

where:

N_h = Size of stratum h (in number of sample units)

$\hat{p}_h$ = Estimated proportion damaged for stratum h

N = Total field size (in number of sample units)

Calculation continued…

Calculation of the standard error of the stratified mean proportion, $\text{SE}(\hat{p}_{ST})$:

$$\text{SE}(\hat{p}_{ST}) = \frac{1}{4000} \sqrt{\sum \left[\begin{array}{l} \dfrac{800^2 \times (800 - 306) \times 0.0229 \times (1 - 0.0229)}{(800 - 1) \times (306 - 1)} + \\[2em] \dfrac{800^2 \times (800 - 327) \times 0.0489 \times (1 - 0.0489)}{(800 - 1) \times (327 - 1)} + \\[2em] \dfrac{800^2 \times (800 - 277) \times 0.0794 \times (1 - 0.0794)}{(800 - 1) \times (277 - 1)} + \\[2em] \dfrac{800^2 \times (800 - 317) \times 0.0347 \times (1 - 0.0347)}{(800 - 1) \times (317 - 1)} + \\[2em] \dfrac{800^2 \times (800 - 342) \times 0.0468 \times (1 - 0.0468)}{(800 - 1) \times (342 - 1)} \end{array} \right]}$$

The standard error of the stratified mean proportion damaged by rodents ($\text{SE}(\hat{p}_{ST})$) is given by:

$$\text{SE}(\hat{p}_{ST}) = \frac{1}{N} \sqrt{\sum \left[\frac{N_h^2 (N_h - n_h) \hat{p}_h \hat{q}_h}{(N_h - 1)(n_h - 1)} \right]}$$

where:

N_h = Size of stratum h (in number of sample units)

n_h = Sample size in stratum h (= 4 in this case)

$\hat{p}_h$ = Estimated proportion damaged for stratum h

$\hat{q}_h$ = $1 - \hat{p}_h$

N = Total field size (in number of sample units)

$\text{SE}(\hat{p}_{ST}) = 0.0042$

In this example, the confidence limits for the stratified mean proportion were 0.038 to 0.055.

An EXCEL spreadsheet program (Stratified_Damage_Estimates.xls) to do these calculations from raw data is available on request from rodent-inquiries@csiro.au. Note that this spreadsheet will do the correct calculations even if there is only one transect.

Glossary

authority
the author and date of publication of a species name

bicoloured
(of the tail) the upper half of the tail differs in colour from the lower half

breeding season
(of a population) starts with the first successful mating after a period of non-breeding, and ends when the last litter of pups is weaned

common property resource
users share the 'rights' and 'benefits' of resource use, and also share the 'duties' of resource management

control
(in an experiment) an experimental unit that has been given no treatment, or the baseline against which the other experimental outcomes are compared

dorsal/dorsum
(anatomy) technical term for an animal's upper surface or back

effective trap-nights
the total number of traps set, minus any traps that are sprung without making a capture

exclusion plot
a representative area of crop that is protected against rodent damage by a rodent-proof fence or barrier

experimental unit
the smallest division of the experimental material such that any two units may receive different treatments

gestation period
the period from conception to delivery of offspring

guard hairs
long, straight, thick hairs that project some distance beyond the general body hair

head+body
the combined length of the head and body, measured from the tip of the nose to the centre of the anus

home range
the area used by an individual animal in the course of its regular pattern of activities

hypothesis
(plural: hypotheses) an explanation for one or more observations; can be tested by further observations or by an experiment to manipulate one or more factors

hybridisation
interbreeding between members of two different species

imperforate
(vagina) in juvenile rodents, the vagina is sealed off by a thin, shiny layer of skin, hence **imperforate**. As the animal reaches sexual maturity, the vaginal covering breaks down and the vagina is open or **perforate** from then on

intensity of infection
(by a parasite) number of parasites per infected animal

interspersion
(in an experiment) the planned placement of treatments and controls to obtain a good spatial mixture

Julian date
the number of a day within a year from day 1 through to day 365 (366 in a leap year)

mammae
teats of female rodents

manus	technical term for the fore-foot (also known as the fore-paw or hand)	**range overlap**	the proportion of a home range that is used by more than one individual of the same species
murid	rodent of the Family Muridae	**range span**	the largest distance across a home range
necropsy	dissection and examination of a dead animal	**replication**	the repetition of a basic experiment (each repeated version is called a replicate)
neophobia	fear of new objects in the environment	**reproductive potential**	the number of offspring that a typical female is likely to produce during her life
palpation	a technique used to confirm whether a female is pregnant; it involves running a thumb and finger gently down the abdomen to feel the developing embryos	**stratified random sampling**	sampling method whereby a population is first subdivided according to some criterion into non-overlapping subgroups called strata, and then sampled on a random basis within each stratum
pes	technical term for the hind-foot	**transect**	a line of traps, set through an area of uniform habitat type
population	a group of individuals that occupy a single locality and among which all members of one sex could potentially interbreed with all members of the opposite sex	**trap–barrier system (TBS)**	a combination of multiple-capture traps integrated with a drift fence which directs rodents to the traps
population density	number of animals of a given species per unit area	**trap-line**	a series of traps, usually placed at set intervals along a transect
prevalence of infection	percentage of animals infected with a particular parasite or disease agent	**trap-nights**	calculated by multiplying the number of traps by the number of nights of trapping, e.g. 100 traps set for 4 consecutive nights equals 400 trap-nights
radio-tracking	method for observing the movements of animals using a radio-transmitter that is attached to an animal	**trapping effort**	total number of effective trap-nights over a particular trapping period
randomisation	(in experimental design) taking a random sample from the population or assigning treatments at random to experimental units		

trapping grid   traps set in parallel lines that ensure an even density of traps per unit area

trap success   number of rodents caught, divided by the total number of effective trap nights—this value is usually multiplied by 100 to give percentage trap success

treatment   (in an experiment) an experimental unit that has been manipulated in some way (to be compared to control or untreated units)

triangulation   the process by which the location of a radio transmitter can be determined by measuring the direction of the received signal from two or three different points

trimester   a third of the gestation period (the gestation period is divided into first, second and third trimesters)

tubercle   a small knoblike prominence projecting from the plantar or under-surface of the foot (pes)

ventral/ventrum   (anatomy) technical term for an animal's belly or underside

vibrissae   (anatomy) technical term for sensory whiskers, found on the head and limbs

zoonoses   diseases that can be transmitted between animal hosts and humans; also known as zoonotic diseases

Index

Note: on many occasions through this index, readers are referred to 'species accounts'. Species accounts make up the bulk of Chapter 11. Each species account provides a summary of all the information available on the featured rodent species, and includes the scientific and common names for the pest rodent, distribution map, table of adult measurements, and information under the following headings—morphological features; mammae; other recently applied scientific names; distribution; taxonomic issues; habitat use; nesting behaviour; breeding biology; population dynamics; and damage to crops.

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