NSW DEPARTMENT OF
PRIMARY INDUSTRIES

Australian Government
Department of Agriculture, Fisheries and Forestry

Other titles in this series:

Monitoring Techniques for Vertebrate Pests – Rabbits

Monitoring Techniques for Vertebrate Pests – Foxes

Monitoring Techniques for Vertebrate Pests – Cats

Monitoring Techniques for Vertebrate Pests – Feral Goats

Monitoring Techniques for Vertebrate Pests – Feral Pigs

Monitoring Techniques for Vertebrate Pests – Wild Dogs

MONITORING TECHNIQUES FOR VERTEBRATE PESTS

MICE

BRUCE MITCHELL AND SUZANNE BALOGH

NSW DEPARTMENT OF PRIMARY INDUSTRIES

BUREAU OF RURAL SCIENCES

NSW DEPARTMENT OF PRIMARY INDUSTRIES

Australian Government
Department of Agriculture, Fisheries and Forestry

MONITORING TECHNIQUES FOR
VERTEBRATE PESTS

MICE

BRUCE MITCHELL AND SUZANNE BALOGH

NSW DEPARTMENT OF PRIMARY INDUSTRIES

BUREAU OF RURAL SCIENCES
NATURAL HERITAGE TRUST

NSW DEPARTMENT OF
PRIMARY INDUSTRIES

Australian Government
Department of Agriculture,
Fisheries and Forestry

Acknowledgments

The authors would like to thank Glen Saunders, Steve McLeod, John Tracey, Peter Fleming, Michelle Dawson, Brian Lukins, David Croft, Matt Gentle, Peter West, Ben Russell, Quentin Hart, Dave Forsyth, Peter Brown, Bill Atkinson and Nathan Cutter for their help with preparing this manual. Thanks also to the many other people too numerous to mention who gave the benefit of their experience.

The National Feral Animal Control Program funded the project.

Citation:
Mitchell, B. and Balogh, S. (2007) NSW DPI Orange
ISBN 978 0 7347 1880 8

Bureau of Rural Sciences, Canberra
October 2007
ISBN 978 0 7347 1880 8
Written by Bruce Mitchell and Suzanne Balogh

Monitoring Techniques for Vertebrate Pests:
Series ISBN 978 0 7347 1888 4

TABLE OF CONTENTS

WHY MONITOR VERTEBRATE PESTS?

Since 1993, the Bureau of Rural Sciences has produced a series of 'best practice' national guidelines to manage the agricultural and environmental damage caused by vertebrate pests. These publications set down principles and strategic approaches for managing vertebrate pests.

The strategic approach to pest animal management is based on six key steps (Braysher 1993):

1. define the problem in terms of impact
2. determine the objectives and performance indicators
3. identify and evaluate management options
4. implement the program
5. **monitor the management program**
6. evaluate the overall management program.

The focus of this manual is to provide details of the techniques available to researchers, land managers and policymakers for monitoring mice in Australia. The manual covers simple monitoring techniques and analysis as well as highly complex and detailed techniques for specialist areas. It is acknowledged that many techniques described here will be impractical for routine farm-level monitoring, while others will not be precise enough for research. End users are encouraged to develop specific monitoring tools for their own purposes based on the descriptions in this manual.

The management program should be monitored before, during and after control, especially if it is a long-term program.

- Monitoring is done **before** the program to establish a benchmark of vertebrate pest abundance and to identify actual or potential damage. This will allow objectives and performance indicators to be determined.
- Monitoring **during** the program is done to determine how the program is progressing against set objectives. The monitoring may provide an early warning that a change in the management program is required so as to achieve control success. This form of adaptive management is recommended to help achieve outcomes within timeframes and budgets without sustaining too much damage; however, it is rarely suitable for research.
- Monitoring **after** the program finishes is aimed at determining the success of the program against the performance indicators, and finding out whether the program objectives have been achieved.

Monitoring of vertebrate pest impacts and their abundance is critical in determining whether a management program has been successful or not.

A management program that incorporates monitoring of both vertebrate pest abundance and the impacts that the pests have will probably be more successful than one that monitors only one of these factors.

There are numerous research and management reasons for initiating monitoring programs of animal populations. Monitoring plays a fundamental role in conservation, by providing an 'early warning system' to identify problems before they become irreparable, and it can also suggest possible solutions (Goldsmith 1991; Thomas 1996). An example of this

is monitoring the abundance of threatened and endangered native species as part of pest animal control programs that aim to protect them.

When an animal species conflicts with human interests (i.e. becomes a pest to agriculture and the environment) and requires management, the need for monitoring its abundance or impact would seem self-evident (Engeman & Witmer 2000). However, this is often a forgotten component of pest management, although it is an essential function that can guide future management practices and should be an integral and budgeted component of existing and proposed management programs (Braysher 1993; Olsen 1998).

Monitoring in vertebrate pest management has two functions: to provide the necessary information to trigger management action (i.e. to act as an 'early warning system') (Elzinga *et al.* 2001); and to indicate whether a management strategy is achieving its objectives or is in need of alteration (performance monitoring) (Possingham 2001; Edwards *et al.* 2004).

Ideally, it is the damage caused by a particular pest that should be monitored (Hone 1994). However, it is often difficult or impractical to survey pest animal impact and, typically, pest abundance is monitored and used as a surrogate indication of associated damage (Edwards *et al.* 2004). This type of monitoring makes the assumption that there is a known relationship between population size and damage.

The most obvious application for pest animal monitoring is to determine the efficacy of control programs to reduce vertebrate pest abundance. In an ideal world, monitoring should compare treated sites (where the control operation occurs) with untreated sites (where no control has been undertaken), and accurately measure damage and abundance before, during and after control. As already stated, measurements of damage are often not available, so assessments of abundance alone are usually used. However, estimates of the absolute abundance of wild animals are expensive to obtain, and may be unnecessary for many pest management decisions (Caughley 1980). Furthermore, complete counts of all pest animals in an area are rarely practical, and more often than not sample counts are done to provide an index of abundance.

In order for monitoring programs to be effective, efficient and reliable estimates of changes in population or damage need to be obtained (Thomas 1996). In addition, these estimates need to be repeatable, to allow meaningful conclusions to be drawn from any changes. An appropriate way of achieving this is to standardise the methodology. An important component of standardisation is education and training. Two or more people could act on written instructions and get quite different results. Physical demonstration of the monitoring technique and the chance to calibrate measurements against those of experienced operators would be likely to improve the accuracy and precision of any monitoring efforts.

The purpose of this manual is to provide details of the techniques available for monitoring mice in Australia. By providing a step-by-step description of each technique, it will be possible to standardise many monitoring programs and make valid comparisons of abundance and damage across the nation. This is becoming increasingly important for the states, territories and the Australian Government, to help evaluate and prioritise natural resource management investments.

KNOW THE PEST: THE HOUSE MOUSE

History

The house mouse (*Mus domesticus*) became established in Australia around the time of European settlement, and has spread across the entire continent (Singleton & Redhead 1989). They are ever-present in agricultural areas, usually in low numbers, but when conditions are favourable mouse populations may erupt to form plagues. Mouse plagues tend to occur when there is plenty of food and water available, environmental temperatures are not extreme, soil is moist and easy to dig, nesting conditions are favourable, and diseases, parasites and predation are at low levels (Saunders & Giles 1977; Redhead *et al.* 1985; Singleton 1989). Mouse plagues seem to be increasing in frequency, and this may be due to changes in agricultural practice (Caughley *et al.* 1998b). There has been a marked increase in the number of crops grown under irrigation, as well as a change to follow-on summer/winter cropping (Singleton & Brown 1999). In some of the areas with available irrigation, it is not unusual to grow two summer crops that mature at different times and then follow up with a winter cereal crop. The potential impacts of mice in these agricultural systems may therefore become more severe.

Impacts

Mice cause damage to almost all sown crops, by digging into the loose soil immediately after sowing to feed on the seed or newly emerging seedlings (Caughley *et al.* 1998a). Most crops suffer damage before seedling emergence and when the crop is maturing. However, in cereal crops such as wheat, mice chew the growing nodes of the plant and can stop the development of the head or cause the stem to collapse.

In most farm produce storage areas, mice will be present and active (Saunders 2000). When mouse population abundance is low, little damage will occur, but when mice are at plague densities, damage will be high. During a plague it is difficult to maintain the mouse-free status of any facility unless there has been a mouse-proof component incorporated into the initial design and construction. In machinery sheds mice can cause major damage to vehicle wiring, upholstery and electric motors, sometimes causing fires (Caughley *et al.* 1998a).

Mouse plagues cause losses to pig and poultry farmers through increased feed costs, and stress and injuries to stock from attacks by mice (Caughley *et al.* 1998a). They can also transmit a number of diseases to humans and livestock.

Distribution

The house mouse is found throughout Australia in almost all habitats, and has adapted to a wide range of environmental conditions. More importantly, it is common on all agricultural lands, particularly in cereal and summer cropping areas and houses and buildings. Plagues of mice occur predominantly in the grain belts of southern and eastern Australia.

Biology

Diet

Mice eat a wide range of foods, consuming 2–4 g daily (Brown & Singleton 2001) – about 10% of their body weight. In a field situation, mice survive on the seeds of native grasses and thrive on introduced cereal grains. In food storage areas their diet can include cereals, other grains, vegetables, meat, fish, nuts, cheese and non-rancid animal products.

Varying levels of mouse damage to maize

Mouse damage to a sunflower head

Mice will sample all foodstuffs within their range, but may not return to a particular feed type for many days. Mice can successfully live and breed without free water if the moisture content of the food is at least 15% (Brown & Singleton 2001). When mice live in sheds and areas where the food supply has a low moisture content, they need 1–2 g of water daily to survive.

Reproduction

Mice can start breeding at five or six weeks of age, and can produce a litter each month if conditions are good (Caughley 1998). The gestation period is 19–21 days. The female re-mates almost immediately after giving birth, and can become pregnant again within two days of delivering a litter (Brown & Singleton 2001). Litter size is generally 5 or 6, but can be up to 10. Young mice begin eating solid food at 11 days, and are weaned at 21 days of age. They have an average life span of approximately six months, and females generally produce 2–5 litters during their reproductive lives.

In Australia, mice living under field conditions have a seasonal pattern of breeding. This generally begins in early spring and continues until cold or wet conditions develop in late autumn (Caughley 1998). Mice living in unfavourable seasonal conditions may have a shorter breeding period, whereas those with nests in the warmth of buildings or haystacks are likely to have an extended breeding period.

Movements and home range

Mice are most active at night, but can also be seen during the day, particularly around buildings or areas with adequate cover (Caughley 1998), or when densities are high. Their home range is variable according to habitat and season. For example, in the Darling Downs (Queensland) region, home range varies from 0.014 ha in the breeding season to 0.199 ha in the non-breeding season (Krebs *et al.* 1995), whereas in the Mallee of Victoria the variation is 0.037 ha (breeding) to 0.119 ha (non-breeding) (Chambers *et al.* 2000). Young mice are forced to seek new areas during periods of high breeding, and this is one of the factors associated with the development of plagues. Mice tend to follow the same path from refuge to feeding areas. In built structures, paths are often confined to walls, pipes or natural barriers, so telltale smear marks on these structures can be an indication of mouse activity (Brown & Singleton 2001). In the field, distinct tracks through the vegetation become obvious, especially when densities are high.

MONITORING MOUSE ABUNDANCE

Monitoring of the change in relative abundance of mice is essential to allow for pre-emptive mouse control, which will minimise the costs and damage associated with mouse plagues. This section discusses the different methods that can be used to monitor mouse abundance. The summary tables at the end of this handbook summarise these methods and compare them with the methods of monitoring mouse impact presented in the next section.

Trapping

Trapping of house mice in Australia has long been used as a monitoring tool by researchers and some government agencies. There are many trap designs, both lethal (e.g. snap-back traps) and live (e.g. Elliott and Longworth traps), that are useful for capturing mice. The selection of trap design will depend on the resources available for the project and the type of monitoring undertaken. Simple snap-back traps can utilise baits such as bacon rind, peanut butter or pumpkin seeds, but small pieces of leather with a few drops of linseed or canola oil (or similar) added are perhaps the best option, as the leather can be attached permanently. Live capture traps are most commonly baited with peanut butter and rolled oats or wheat. Trapping alone can be used as an index of abundance by using trap success or population estimates made via capture–recapture methods.

Trapping has an advantage over other monitoring techniques, because it also allows the breeding status of mice to be evaluated, thus indicating the potential for population growth (Caughley *et al.* 1998a). Signs of breeding are obviously pregnant females or those that have already started to breed (indicated by prominent mammary glands), as well as the size of mice caught (finding mice with head/body measurements smaller than 72 mm indicates that juveniles are already present) (Saunders 2000).

Trap success

Trap success is the most commonly used method to monitor mouse abundance, and may utilise any trap type (Caughley *et al.* 1998a). Trap success is measured by the number of mice caught divided by the number of trap-nights (trap-nights – the number of traps placed out multiplied by the number of nights of trapping). This figure is often expressed as captures per 100 trap-nights, and is corrected for sprung traps or traps that catch non-target animals by subtracting the number of these failed traps ('null' traps) from the total number of traps set (Aplin *et al.* 2003).

Because the relationship between population density and percentage trap success is curvilinear (see Figure 1), the frequency-density transformation of Caughley (1980) is used to account for traps becoming unavailable once an animal is captured (Ruscoe *et al.* 2001; Aplin *et al.* 2003):

$$v = \log_e(1-f)$$

where f is the frequency of capture and v is the transformed estimate. However, as trap success increases, the precision of this index declines, and during plagues (≥ 90% trap success) its use as an index is no longer practical (Caughley *et al.* 1998a). The simplest index uses a minimum of 20 snap-back traps set out in straight lines, with traps 10 m apart, in a variety of habitats, for two or three consecutive nights (Mutze 1991; Saunders 2000). Trap success is averaged over all habitats and nights to produce an index of abundance (Davis *et al.* 2003). The more traps and transects that are placed, the greater the precision of the index. However, trap success will distinguish only between major differences in abundance (Mutze 1991).

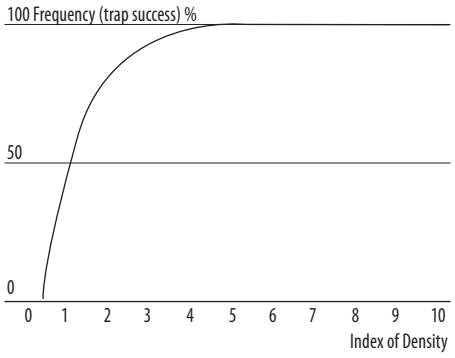


Figure 1. Relationship between frequency (trap success) and density of trapped population (Source: Caughley 1980).

Materials required

Traps – minimum of 20 snap-back traps or live-capture traps

Bait – leather with oil (e.g. canola) dropped on it (use a squeeze bottle or eye dropper), bacon rind or pumpkin seeds

Flagging tape or similar

Bucket – to carry traps in

Countsheet (see example in Table 1)

How to trap

- Select the monitoring sites: choose a variety of habitats/crops and fence lines.
- Ensure that traps are in good working order (it is wise to test-fire them before setting).
- Set out traps in a straight line at 10 m intervals in the late afternoon.
- Mark the location of traps with flagging tape or similar.
- Return early the following day and record the number of mouse captures.

Note: If you are using live-capture traps, check the traps early each morning (just after dawn).

- Reset the traps and repeat the procedure for two or more nights.
- Calculate the trap success (corrected for sprung traps and traps that catch non-target animals).

Trap success (TS) =
no. mice captured ÷ [total no. traps set – (sprung traps + nontarget captures)]

Transform using frequency-density transformation, or an approximation can be made from Table 1.

Adjusted trap success (ATS) = $ln(1 - TS) \times (-100)$

Repeat the count monthly.

Standards

Trap design – use the same type of trap.

Bait – use the same type of bait.

Site – use the same sites for each monitoring effort.

Animal welfare considerations

Impact on target animals – snap-back traps are designed to be lethal, so it is necessary to ensure that each trap's spring is in good working order.

Impact on non-target animals – small mammals such as antechinus and native rodents may be captured in live-capture traps. It is therefore necessary to provide some bedding material (e.g. cotton wool) inside the traps.

Mouse caught in a snap-back or mortality trap

To minimise stress on the animals, the traps should be checked in the early morning after sunrise, with the traps left closed throughout the day to ensure that no captures are possible during the heat of the day. Reset the traps in the late afternoon before sunset.

National Standard Operating Procedures for humane control and research

RES005 *measurement and sampling of pest animals used in research* (Sharp & Saunders 2005).

Health and safety considerations

The health and safety considerations here are important. Ensure that cuts are covered up, avoid mouse urine and faeces, wear protective clothing, and wash hands thoroughly after handling mice.

Training required

Identification of native small mammals and house mice (if using live-capture traps).

Worked example

Two sites were monitored for three consecutive nights at site A (recently-sown sunflower crop) and site B (storage sheds). Twenty traps were placed at each site, with the following results:

Night 1
Site A – 5 mice, 2 sprung traps,
Site B – 12 mice, 1 sprung trap

Night 2
Site A – 7 mice, 1 sprung trap,
Site B – 9 mice, 3 sprung traps

Night 3
Site A – 4 mice, 1 sprung trap,
Site B – 13 mice, 2 sprung traps

Total
Site A – 16 mice, 4 sprung traps,
Site B – 34 mice, 6 sprung traps

Trap success (Site A) $= 16 \div (60 - 4) = 0.286$

Trap success (Site B) $= 34 \div (60 - 6) = 0.630$

Adjusted trap success (site A)
$= ln(1 - 0.286) \times (-100) = 34\%$

Adjusted trap success (site B)
$= ln(1 - 0.630) \times (-100) = 99\%$

Using the transformation, it can be seen that at site B, the estimated density of mice is almost three times as high as at site A, rather than double, as would have been presumed from the untransformed frequencies (adapted from Caughley 1980).

Table 1. Mouse-trapping: example of a count sheet

MOUSE COUNT SHEET					
Date:		Trapper:			
Property name:			Owner's name:		
Nearest town:			RLPB:		
Weather conditions: Cold Cool Mild Warm Hot					
Last rain: > 2 weeks 2 weeks 1 week this week					
CROP			**REFUGE HABITAT**		
Traps set	Mice caught	Traps sprung	Traps set	Mice caught	Traps sprung
Traps interfered with	Cards set	Cards missing	Traps interfered with	Cards set	Cards missing
Cards interfered with			Cards interfered with		
Card percentages	1.	2.	Card percentages	1.	2.
3.	4.	5.	3.	4.	5.
6.	7.	8.	6.	7.	8.
9.	10.	11.	9.	10.	11.
12.	13.	14.	12.	13.	14.
15.	16.		15.	16.	

OFFICE USE

Traps %	
Cards %	

Traps %	
Cards %	

Capture–recapture

Capture–recapture methods are based on multiple sampling, and use repeated capture or sightings of marked or tagged individuals to estimate population size. Animals in the first sample are marked uniquely and then released back into the population. The second sample captures marked (recaptures) and unmarked animals, which are then marked and released, and so on, until the monitoring is finished. The resultant capture history is then used to produce an estimate of the population. Various capture–recapture methods are available for both closed and open populations, and have been reviewed in detail elsewhere (e.g. Seber 1982; Pollock *et al.* 1990; Schwarz & Seber 1999; Buckland *et al.* 2000). All these methods make assumptions that should be satisfied in order to produce unbiased estimates. Assumptions common to mark–recapture models are (from Southwood 1989; Krebs 1989):

1. all animals have equal catchability (marked animals at any given sampling time have the same chances of capture as unmarked animals)
2. marked animals are not affected by being marked (in behaviour or life expectancy)
3. marks are not lost or overlooked (i.e. all previously marked animals can be distinguished from unmarked animals).

Because mice are relatively trappable, capture–recapture studies can work well for them, and have been used in a variety of agricultural habitats (Brown *et al.* 1997; Mutze & Hubbard 2000; Moro 2001; Twigg *et al.* 2002; Jacob *et al.* 2003; Arthur *et al.* 2003). Trapping is carried out using either grids or lines of traps situated in crops or along fence lines. Trapped mice are marked by ear-tag, punch, and/or toe-clipping (less invasive methods of marking could also be investigated, such as DNA analysis, microchipping, tattooing and dye markers) and then released. In Europe, traps specifically designed for multiple capture (e.g. Ugglan traps) are used to enhance capture rates, but these have seldom been used in Australia, and their performance has not compared favourably with that of single-capture traps, such as Elliott or Longworth traps (Jacob *et al.* 2002a). Trapping usually takes place over two or three consecutive nights. The number of marked animals relative to the total number caught is used to estimate the number of mice within the sampled area.

Materials required

Live-capture traps – about 50 Elliott or Longworth traps per site

Bait – wheat, rolled oats and peanut butter

Flagging tape or similar

Ear tags/ear-punch/toe clipper (or other marking method)

Count sheet

How to do the count

- Select the monitoring sites: choose a variety of habitats/crops and fence lines.
- Ensure that traps are in good working order.
- Set out traps in the late afternoon, in a 5 × 5 (or greater) grid with 10 m intervals, placed 50 m from the edge of the crop, and a line of 20 traps at 10 m intervals along a fence line next to the trapping grid.

- Place a small amount of bait and bedding (e.g. cotton wool) inside each trap.
- Mark the locations of traps with flagging tape or similar.
- Return early the following morning and check the traps.
- Record details of captured mice (e.g. sex, weight, reproductive condition).
- Tag/punch the ear and/or clip a toe (record the details of marking).
- Release at point of capture.
- Leave the traps closed until the late afternoon.
- Reset the traps in the late afternoon and repeat the procedure for two or more nights
- Calculate population estimate using an appropriate estimator (e.g. Petersen's index):

$$N = (\text{no. mice caught and marked on 1st night} + 1) \times (\text{total no. mice caught on 2nd night} + 1) \div (\text{no. mice caught on 2nd night that are marked} - \text{recaptures}) - 1$$

$$N = (C_1 + 1) \times (C_2 + 1) \div (R + 1) - 1$$

Standards

Trap design – use the same type of trap.

Bait – use the same type of bait.

Site – use the same sites for each monitoring effort.

Duration – use the same number of trap nights.

Animal welfare considerations

Impact on target animals – to minimise mortality and stress, bedding (e.g. cotton wool) needs to be provided inside the traps, and they should be checked in the early morning after sunrise. Traps should be left closed throughout the day to ensure that no captures are possible during the heat of the day, and then reset in the late afternoon before sunset.

Impact on non-target animals – as for target animals

National Standard Operating Procedures for humane control and research

RES001 *live capture of pest animals used in research* (Sharp & Saunders 2005)

RES002 *restraint and handling of pest animals used in research* (Sharp & Saunders 2005)

RES004 *marking of pest animals used in research* (Sharp & Saunders 2005)

RES005 *measurement and sampling of pest animals used in research* (Sharp & Saunders 2005)

Health and safety considerations

The health and safety considerations here are important. Ensure that cuts are covered up, avoid mouse urine and faeces, wear protective clothing, and wash hands thoroughly after handling mice.

Mice as commensal rodents will often be found in large groups

Training required

- Identification of native small mammals and house mice.
- Correct setting of traps, opening of traps (also care with traps to ensure they function properly), handling, measuring and marking of wild mice.

Bait-take

The amount of bait (toxic or non-toxic) taken from bait stations can be used as an index of mouse abundance (Saunders 1983). This method uses measured quantities of grain placed in bait stations in transect or grid patterns. The weighed amount of bait taken each day can then be used as an index of abundance, and changes in bait take can be plotted over time. A problem with this method is bait-take by non-target species, such as birds and ants. These species, particularly ants, may take bait at a faster rate than mice, thereby confounding the results of bait-take sampling (Jacob *et al.* 2002a). Bait stations should therefore be constructed so that they are inaccessible to birds and animals that are larger than mice. There is no way to exclude ants and still allow access to mice. A simple example of a bait station is an ice-cream container that has holes cut in the side large enough to allow entry to mice and is covered with a weighted lid. A further problem associated with bait stations is the food preference of mice at the time of sampling. If food is plentiful or there is a more attractive food source available, the ability of bait stations to accurately detect change will be limited.

Materials required

Bait stations – containers that allow only access to mice

Bait

Countsheet

Weight scales

Flagging tape or similar – to mark bait station locations

How to do the count

- Select the monitoring sites: choose a variety of habitats and fence lines, and sites around buildings.
- Set out bait stations in a 5 × 5 (or greater) grid with 10 m intervals, or in a line of 20 or more at 10 m intervals.
- Place an equal amount of bait inside each bait station (e.g. 100 g of grain).
- Mark the locations of bait stations with flagging tape or similar.
- Return the following morning and check the bait stations.
- Weigh the amount of grain remaining and determine how much bait was consumed.
- Replace the bait, so that all bait stations have an equal amount of bait.
- Repeat the procedure for two or more nights.

- Calculate the average daily bait take per grid/transect to use as an index of abundance.
- Conduct the survey on a monthly basis.
- Plot the bait-take over time, to evaluate trends in mouse abundance.

Standards

Monitoring site – use the same sites for every survey.

Bait – use the same bait type and amount.

Duration – monitor for the same number of nights.

Animal welfare considerations

Impact on target animals – nil

Impact on non-target animals – nil

National Standard Operating Procedures for humane control and research

None

Health and safety considerations

None

Training required

None

Mouse index squares

During the 1970 mouse plague in south-eastern Australia, a monitoring technique using adding machine paper soaked in cooking oil was developed (Ryan & Jones 1972). This led to the development of 10 cm squares of paper, also soaked in cooking oil, to monitor mouse activity. These are now commonly referred to as census cards, which implies a complete survey of the population; we will use the term 'mouse index squares' here to prevent confusion. This method uses oil-soaked paper squares pegged to the ground and left overnight to attract mice. The proportion of the square that is chewed by the mice is then used as an index of abundance. Mouse index squares are a simple way of monitoring, and it has been shown that the proportion chewed is significantly correlated with the estimated mouse density and percentage trap success (Caughley *et al.* 1998b), although in some situations there is no correlation. The squares may provide a reliable index of population change when mouse population density is high, but their reliability when lower densities occur has been questioned (Caughley 1998; Mutze 1998), with the availability of alternative food influencing the success of this technique in detecting change. The optimum times of mouse index square use are autumn and winter, when crops are immature and food is limited (Mutze 1998; Walsh *et al.* 2000). Mouse index squares may be used to determine the timing of mouse control measures, with an average consumption of more than 20% of the squares on each card indicating that mouse density is already high and that active control measures are required to minimise crop damage (Saunders 2000). Consumption of 10% of the squares has been recommended as the threshold for which cost-effective baiting should be undertaken to prevent an explosion of mouse numbers (Caughley *et al.* 1998b; Caughley 1998).

Materials required

White A4 copy paper

Photocopier

Skewers or pegs

Flagging tape

Canola oil or linseed oil

Scissors

Container to hold oil while mouse index squares are being soaked

Bucket to carry items in

How to use mouse index squares

- Select the monitoring sites: choose a variety of habitats and fence lines.
- Cut enough mouse index squares for the count (10 mouse index squares per monitoring line) (see template in Figure 2).
- Mouse index squares are 10 cm × 10 cm (a piece of A4 paper gives six complete mouse index squares).
- Draw 1 cm grids on the mouse index squares or photocopy them (see attachment for template).
- Soak the mouse index squares in canola or linseed oil for approximately 1 hour.
- After soaking, drain the mouse index squares for approximately 10 minutes.
- Put mouse index squares out in the late afternoon, with 10 mouse index squares spaced at 10 m intervals, making up a monitoring line 100 m long.
- Record the location and type of vegetation and mark the line (e.g. with flagging tape).
- Secure the mouse index squares to the ground with skewers, thin pegs or wire spikes, so that they do not get blown away or carried off by mice or other animals, such as foxes.
- Collect the mouse index squares the following morning.
- Mouse presence and damage potential can be assessed by determining the percentage of each mouse index square eaten. Counting the number of 1 cm × 1 cm portions eaten will give a percentage for the whole mouse index square (each 1 cm × 1 cm portion is 1% of the mouse index square).
- Use the average of the 10 mouse index squares to determine the percentage eaten for each site.
- Repeat the count on the following night and average the score for each site.

Standards

Mouse index squares – use the same type of paper (white bond paper) and same size (10 cm x 10 cm).

Distance – place at 10 m intervals.

Oil – use the same type of oil (canola or linseed).

Site – Use the same monitoring lines each time for comparison.

Animal welfare considerations

Impact on target animals – nil

Impact on non-target animals – nil

National Standard Operating Procedures for humane control and research

None

Health and safety considerations

None

Training required

None

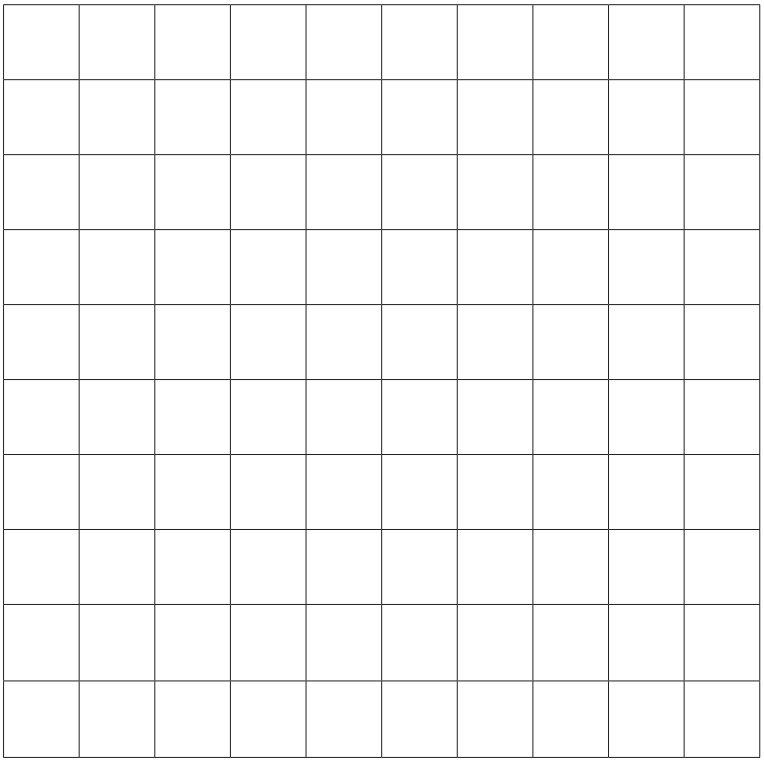
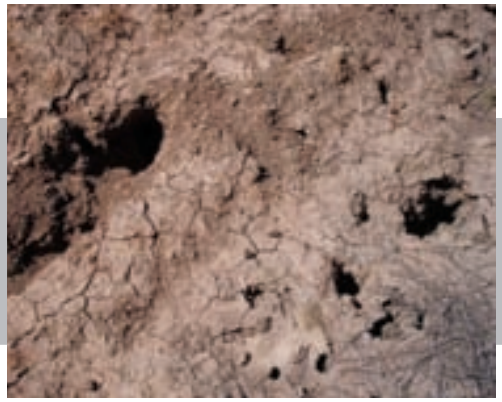


Figure 2. Template for mouse index squares



Mouse holes with an active hole (top left)

Counts of active holes and runways

The number of active mouse holes or runways (paths) within a given area or along transects can be used to monitor mouse activity. Active holes can be identified by lightly covering all holes encountered with soil or small pieces of paper and then counting the reopened holes the following day. Alternatively, husks of grain or freshly excavated soil at the entrance just after rain are signs of an active hole (Caughley *et al.* 1998a). Active holes counts are best conducted in areas where the soil type is conducive to hole digging. If the soil is self-mulching or cracking, this technique has limited application, because mice do not readily dig burrows. Mice tend to use the same runway when moving between refuge areas and feeding sources, and these can be clearly seen passing through undisturbed vegetation when high densities of mice are present (Brown & Singleton 2001).

A major limitation to active hole counts is that it is not possible to determine the number of mice that inhabit each burrow. Active hole counts will tend to underestimate reductions in mouse populations. After baiting, mice may redistribute themselves to utilise the available burrows. For example, if 20 mice inhabit a hole before baiting and one mouse survives control, no reduction in activity would be recorded because the hole would appear as active as if all 20 had survived (provided the survivor continues to use the same hole). The only way to correct for this would be to excavate a large proportion of holes at each site before and after control and count the number of mice per hole. This is beyond the scope of the majority of monitoring programs. As a result, active hole counts should be used only to monitor for increases in mouse abundance and activity.

Materials required

Countsheet

Pegs to mark transect or plot active holes

Active hole identifier – e.g. paper or soil; flour or talcum powder has also been suggested)

How to do the count

- Select sites to be monitored: choose a variety of soil types and crop growth.
- More sites means a more accurate count. (This can be said for any monitoring technique.)
- Repeat the counts monthly, or weekly if there is concern of mouse abundance increasing.

Transect count (active holes):

- Use pegs to mark out a straight line transect 100 m long.
- Lightly cover with soil all mouse holes that are within 1 m either side of the transect (200 m^2 strip).
- Return the next day and count the number of holes that have been reopened within 1 m either side of transect.
- Average the counts and multiply by 50 to get the number of active holes per hectare.

Transect count (runways):

- Use pegs to mark out a straight line transect 100 m long.
- Count the number of runways that are within 1 m either side of the transect (200 m^2 strip).
- Average the counts and multiply by 50 to get the number of active holes per hectare.

Plot count:

- Mark out a 10 m radius circle (314 m^2).
- Try to keep the hole covering method the same (e.g. cover with soil or paper).
- Cover with soil all mouse holes that are within the plot.
- Return the next day and count the number of holes that have been reopened within the plot.
- Average the counts and multiply by 31.85 to get the number of active holes per hectare.

Standards

Sampling sites – use the same sites for each count.

Animal welfare considerations

Impact on target animals – nil

Impact on non-target animals – nil

National Standard Operating Procedures for humane control and research

None

Health and safety considerations

None

Training required

Identification of mouse burrows (compared with lizard or scorpion burrows)

Mouse sign

Sightings of mice

Mice may be seen running across roads and tracks at night, and in homes and sheds, providing a general indicator that mouse densities are high. Factors other than density determine the movement of mice across roads, for example weather conditions: after rain, mice may come to the edge of the road to drink (Brown & Singleton 2001). Mice will be most noticeable in and around buildings in winter, when they are likely to move in to seek shelter from the cold and find food (Caughley et al. 1998a). The number of mice seen in a shed in a one minute period after switching on the light may provide an indicator of mouse abundance (Caughley 1998). However, if mice are not seen running across the road at night or in buildings, do not assume that they are not present.

Faecal pellets and faecal smears

Faecal pellets are useful for indicating the presence of mice in larger crops such as sunflower and corn, where they will be found at the junctions of the leaves and stem, or on the seed head (Saunders 2000). Mice will also leave characteristic faecal smears on surfaces that are visited often, such as metal railings inside sheds, which sometimes act as highways for mice (Brown & Singleton 2001).

Gnawing

Mice need to gnaw to keep their continuously growing incisor teeth sharp, and their tooth marks can be monitored on food, wood, electrical wiring and other building materials and farm equipment (Brown & Singleton 2001).

Nest sites

Mice do not only nest in holes: they will also use pipes, areas underneath planks and tin, and irrigated crops such as rice (in rice, their nests will often be visible as leaf and stem platforms just above the water level) (Caughley et al. 1998a; Saunders 2000). Likely or previous nest sites can be monitored on a monthly basis for mouse sign, and can also be used to detect when mice are breeding (Caughley et al. 1998a). Mice will also live in irrigation siphons, and the number of mice running out of the siphons when water is put into the crop may be counted (Brown & Singleton 2001).

MONITORING MOUSE IMPACT

Monitoring of the levels of damage caused by mice can be used to determine whether control should be conducted.

This section discusses the different methods that can be used to monitor the impact caused by mice. The summary tables at the end of this handbook summarise these methods and compare them with the methods of monitoring mouse abundance discussed in the previous section.

Monitoring economic costs

Costs of control

The cost and/or effort involved with annual mouse control can be used as a surrogate for estimating trends in mouse abundance. Aerial baiting or ground baiting costs can be evaluated, and either the total regional cost or cost for individual properties (ha^{-1}) recorded. Similarly, the quantity of bait dispensed at a regional scale or individual property scale can be monitored.

Other costs

It is difficult to estimate accurately the agricultural costs attributable to mice in Australia on a national, state or regional level (Bomford & Hart 2002). Conservative estimates have placed a monetary value of $35.6 million on the national annual cost impact of mice (McLeod 2004), with plagues causing 'spikes' of losses. For example, a mouse plague in South Australia and Victoria in 1993 cost an estimated $64.5 million, with almost 95% of this impact felt by grain growers (Caughley *et al.* 1994). However, the annual value is based on limited information that has been extrapolated from sources such as government agency estimates and landholder surveys, and it has been acknowledged that there are many gaps in the knowledge (Bomford and Hart 2002; McLeod 2004). Individual landholders can therefore play a significant role in filling these gaps by calculating and monitoring all the costs attributable to mice. These costs include control expenditure; infrastructure installation, inspection and maintenance (e.g. mouse-proofing); changes in livestock production (e.g. poultry); and changes in crop production output. These costs can be recorded as part of the economical management of a property, and hence there is little extra expense to the landholder. By monitoring these costs (see Table 2), the actual costs associated with mouse control can be determined at a local level. The inference that is made from cost monitoring is that a decline in costs is associated with a decline in mouse abundance.

Table 2. Example of a sheet used to monitor other costs

ACTIVITY	LABOUR ……h @ $ h^{-1}	MATERIAL	COST $
Poison baiting		Vehicle @ $ km^{-1} Poison bait	
Crop inspection			
Mouse-proofing maintenance			
Mouse-harbour management			
Crop damage assessment			
Re-sowing costs			

Monitoring crop damage and yield losses

Mice can have an impact on crops at any stage of their development. Damage is generally visible when mouse density is high (> 200 mice ha^{-1}), but is often overlooked or not perceived at lower densities (Caughley *et al.* 1998a; Brown & Singleton 2001). However, this depends on the type of crop: for example, cauliflower growers have a much lower tolerance than wheat growers. When assessing crop damage, it is necessary to distinguish between the damage attributable to mice and that caused by insects, birds or frost. Damage by mice includes the following.

- After sowing and before germination, mice may dig freshly sown seeds out of the ground, leaving a funnel-shaped hole, and may sometimes leave the husk of the seed nearby.
- Immature crops may have stems gnawed just above the nodes, causing the heads of the plants to die off and become visible as brown patches in a green crop.
- In more mature crops, damaged heads tend to stand higher because of the removal of the grain.
- Other signs include mounds of gnawed grain at points within the crop, particularly around burrow entrances (Saunders 2000).

Damage to grain in some crops, such as sunflower, can be differentiated from the damage caused by birds, as birds tend to remove the entire seed from the head, whereas mice gnaw at the seed while it is still attached. Other distinguishing features of mouse damage are chewed husks, mouse droppings, and other debris at the base of the plants (Caughley *et al.* 1998a).

Damage to emerging crops

The percentage of seed lost can be estimated by a simple quadrat sampling method (Aplin *et al.* 2003), by comparing the number of plants that germinate per unit area with the quantity of seed that was sown across the same area. This technique will overestimate the extent of mouse damage, because of the infrequency of 100% germination of sown seeds. A more accurate method of assessing damage to emerging crops is to use exclosures to compare protected and unprotected areas (Aplin *et al.* 2003). Take care to ensure that the exclosure is proof against the burrowing, gnawing and climbing abilities of mice. Problems with this technique include the effects of the fencing on the crop, due to changes in sunlight, humidity and wind flow. This can be partly overcome by sampling only from the centre of the exclosure. However, the fencing also provides perching sites for birds, and could thus potentially increase the rate of bird damage to the protected area.

Materials required

Four star posts or similar per exclosure (three or more exclosures per crop)

Plastic fencing (e.g. roofing material at least 700 mm wide)

Fencing wire

Shovel and post driver

Exclosure construction

- Construct exclosures immediately after sowing of the crop is finished.
- Place exclosures at intervals of 10, 20 and 50 m from the edge of the crop (if possible construct at least two exclosures at each distance, for replication).
- Each exclosure should be the same size (e.g. 2 × 2 m).
- Drive posts into the ground in a square formation, and put fencing up so that it is buried to at least 100 mm and at a height of at least 600 mm above the ground.
- Secure the plastic sheet fencing with wire between the posts.
- Ensure that the corners are mouse-proof.

How to do the count

- When the crop germinates, count the number of emerging plants in the centre of each exclosure using a 1 m^2 quadrat.
- Count the number of emerging plants in a corresponding number of unprotected areas using a 1 m^2 quadrat.
- The placement of these quadrats depends on the pattern of damage: if the damage seems to be randomly distributed, use random placement; if the damage seems to have some form of pattern, stratified random sampling (see below) is required.
- Determine the damage rate:

Damage rate (%) =
(1 – no. of plants in unprotected area × no. of plants in exclosure) × 100

Standards

Exclosures – ensure that exclosures are all the same size.

Quadrat – use the same-sized quadrat for all counts.

Animal welfare considerations

Impact on target animals – nil

Impact on non-target animals – nil

National Standard Operating Procedures for humane control and research

None

Health and safety considerations

None

Training required

None

Damage to maturing crops

The proportion of damaged tillers (plant stems that produce grain heads) to undamaged tillers in a sample of individual plants can be used as an estimate of the damage to a cereal crop. The numbers of tillers that are undamaged, recently damaged, previously damaged but regrowing, or previously cut but not regrowing are recorded for each plant. The sum of these will give the total number of tillers in the sample and the proportion of damaged tillers can be calculated (Aplin *et al.* 2003). As this technique involves an arduous process it is often completed only once, just before harvest, to give a minimum estimate of yield loss. Stratified random sampling has been suggested as the most appropriate method, as crop damage could vary with the distance from the crop edge (Singleton *et al.* 1991; Mutze 1993; Brown & Singleton 2001). Four (or more) transects are established, reaching to the centre of the field and separated by 20 m. On each transect, plants are assessed for damage at five positions separated equidistantly. The information is then converted to an estimate of proportional damage for the entire field. Yield losses can be calculated by converting the crop damage information, although this relationship is complicated by two factors: damage may have occurred throughout the growing period, resulting in a cumulative effect on yield at harvest; and there may have been growth compensation by plants following damage (Aplin *et al.* 2003).

Materials required

Damage assessment data sheets (example in Table 3)

How to do the count

- Establish a baseline along the long axis of the field.
- Set out four transects, each separated by 20 m, perpendicular to the baseline running from the edge to the centre of the crop.
- On each transect establish five equidistantly separated points (e.g. if the transect has a length of 100 m, there will be points at 20, 40, 60, 80 and 100 m from the baseline).
- At each point assess mouse damage on every fifth plant along a line perpendicular to the transect until 10 plants are counted.
- Record the information on a standard data sheet.
- Repeat the counts for the other three transects.
- Calculate the estimated proportion of mouse damage for the field:

$\hat{p}_{ST} = \Sigma N_h \hat{p}_h \div N$

Stratified average proportion damaged by mice =
Σ size of stratum *h* (in no. of sample units) × estimated proportional damage for stratum *h* ÷ total field size (in no. of sample units)

(see worked example in Table 4)

Standards

Number of transects – keep constant if comparing between crops or years.

Number of plants counted – keep constant

Strata – use the same strata for comparative counts.

Observer – use the same person(s) for each transect.

Animal welfare considerations

Impact on target animals – nil

Impact on non-target animals – nil

National Standard Operating Procedures for humane control and research

None

Health and safety considerations

None

Training required

Use of quadrats

Health and safety considerations

None

Training required

None

*Table 3. Example of a crop damage data sheet**

CROP TYPE												
District:			Site name:					Transect no.:				
Date:	Name of data recorder:	Entered by:				Verified by:				Page of		
DISTANCE	**NUMBER OF TILLERS ON PLANT**	**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											

* Adapted from Aplin *et al.* (2003).

Table 4. Worked example of crop damage calculations

The following example has been adapted from Aplin *et al.* (2003).

Calculation of damage caused by mice in a field that is 250 m × 100 m
Size of field = 25 000 m^2
Area of one set of transect samples (40 plants) in m^2 (size of stratum h (N_h)) = 0.5
Total area in units of samples (N) = 25 000 ÷ 0.5 = 50 000

STRATA	NUMBER OF SAMPLED TILLERS DAMAGED BY MICE								AVERAGE PROPORTION (damaged tillers ÷ total no. of tillers in stratum$_h$)	STRATUM SIZE (N_h) (N/no. of strata)
	Transect 1		Transect 2		Transect 3		Transect 4			
	Damaged	Total	Damaged	Total	Damaged	Total	Damaged	Total		
Edge of field	9	115	4	60	2	89	9	115	24 ÷ 379 = 0.0633	10 000
20% in	8	117	4	62	3	94	1	76	16 ÷ 349 = 0.0458	10 000
30% in	1	76	8	93	6	90	9	70	24 ÷ 329 = 0.0729	10 000
40% in	4	108	2	87	4	74	11	109	21 ÷ 378 = 0.0556	10 000
Centre of field	2	72	4	92	1	96	4	73	11 ÷ 333 = 0.0330	10 000

Estimated mean proportion damaged averaged over all strata ($\hat{p}_{ST}$) = sum of stratum size × average proportion
= [(0.0633 × 10 000) + (0.0458 + 10 000) + (0.0729 × 10 000) + (0.0556 × 10 000) + (0.0330 × 10 000)] ÷ 50 000 = 0.0541

Calculation of standard error of the stratified mean proportion, SE($\hat{p}_{ST}$): where

N_h = size of stratum h (in number of sample units)
n_h = sample size in stratum h
$\hat{p}_h$ = estimated proportion damaged for stratum h
$\hat{q}_h = 1 - \hat{p}_h$
N = total field size (in number of sample units)

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

$$SE(\hat{p}_{ST}) = \frac{1}{50\,000}\sqrt{\sum\left[\begin{array}{l}\dfrac{10\,000^2 \times (10\,000 - 379) \times (1 - 0.0633)}{(10\,000 - 1) \times (379 - 1)} + \\ \dfrac{10\,000^2 \times (10\,000 - 349) \times (1 - 0.0458)}{(10\,000 - 1) \times (379 - 1)} + \\ \dfrac{10\,000^2 \times (10\,000 - 329) \times (1 - 0.0729)}{(10\,000 - 1) \times (329 - 1)} + \\ \dfrac{10\,000^2 \times (10\,000 - 333) \times (1 - 0.0333)}{(10\,000 - 1) \times (333 - 1)}\end{array}\right]}$$

SE($\hat{p}_{ST}$) = 0.0113

A Microsoft EXCEL spreadsheet program (Stratified_Damage_Estimates.xls) that calculates the mean proportional damage and standard error (even if only one transect is completed) is available on request from rodent-inquiries@csiro.au.

SUMMARY OF MOUSE MONITORING TECHNIQUES

The various mouse abundance and impact monitoring techniques discussed in this manual, and their advantages and disadvantages, are listed in Table 5. Table 6 compares the different monitoring techniques.

Table 5. Advantages and disadvantages of the monitoring techniques discussed in this manual

MONITORING TECHNIQUE	ADVANTAGES	DISADVANTAGES
Trap success	• simple and inexpensive • breeding condition of mice can be assessed	• can detect only large changes in abundance
Capture–recapture	• accurate estimate of abundance • breeding condition of mice can be assessed	• expensive • labour intensive • time consuming
Bait-take	• simple	• no easy way to determine if bait-take is solely by mice • bait-take will be influenced by abundance and availability of other food sources
Mouse index squares	• simple and inexpensive • can indicate optimal timing of control	• cannot be used when crops are mature or succulent • unreliable when mouse population density is low • cannot be used in wet weather
Active hole/runway counts	• simple and inexpensive	• not useful in areas where mice do not create holes • number of active holes may not be strongly correlated with mouse density • unreliable indicator of control success • time consuming
Costs of control	• inexpensive – part of control program • can be incorporated into existing economical management	• unreliable if degree of effort or methodology changes • costs increase each year – need to account for inflation
Other cost monitoring	• inexpensive • can be incorporated into existing economical management	• assumed relationship with mouse abundance
Crop damage: exclosures	• gives indication of seed loss attributable to mice	• expensive • may increase bird damage to protected area of crop
Crop damage: proportion of damaged tillers	• gives indication of grain loss/damage attributable to mice	• labour-intensive • compensatory growth in tillers can disguise the extent of mouse damage occurring before harvest

Table 6. Mouse monitoring techniques ranking table

	LABOUR	START-UP COST	EXPERTISE AND TRAINING	SPECIALISED EQUIPMENT	HUMANENESS	OH&S RISK	COMMENTS
Trap success	Medium	Low	Low	Low	Moderate	Low	Lethal monitoring technique
Capture–recapture	High	Medium (need to purchase traps)	Medium (need to know how to set traps properly and collect proper data)	Medium (need proper traps and equipment for measuring mice)	Low	Low, but if manual handling is involved there may be a disease risk	Humaneness depends on marking and handling techniques
Bait-take	Low	Low	Low	Low	Moderate	Low	Lethal monitoring technique. Humaneness subject to type of poison used.
Mouse index squares	Medium	Low	Low	Low	High	Low	
Active hole/runway counts	Medium	Low	Low	Low	High	Low	
Crop damage: exclosure	High	Medium	Medium	Medium (need specialist equipment and good construction	High	Low	
Crop damage: proportion of damaged tillers	High	Medium	Medium	Low	High	Low	

GLOSSARY

Elliott trap

A small aluminium box with a treadle snare in the middle and a small flap that snaps shut. Used to catch small mammals. Generally single animal capture.

Exclosure

An area that is fenced off to rest it from grazing or other use by animals.

Index of abundance

A relative measure of the abundance of a species (for example, catch per unit effort).

Longworth trap

A live-catch aluminium trap for small mammals. It has a tunnel leading to a large nest box. The trigger sensitivity is adjustable. Generally single animal capture.

Petersen estimate

A method of estimating population abundance on the basis of the ratio of marked to unmarked individuals within a population. It assumes that the population is closed to immigration and emigration, and assumes that population size is related to the number of marked and released animals in the same way that the total caught at a subsequent time is related to the number recaptured.

Quadrat

An ecological sampling unit that consists of a square frame of known area. The quadrat is used for quantifying the number or percentage cover of a given species within a given area.

Runway

Pathway used by a mouse to access nesting sites etc.

Stratified random sampling

(Also called *proportional* or *quota* random sampling) sampling technique in which the population is divided into homogeneous subgroups and then a simple random sample is taken from each subgroup.

Transect

A straight line placed on the ground, along which ecological measurements are taken.

Trap night

The number of traps placed out multiplied by the number of nights of trapping.

Ugglan trap

A wire mesh trap equipped with an aluminium roof for sun and rain protection and a plastic bottom plate to prevent frost injuries and make cleaning easy. They are constructed with a tramp plate, a tip function and a catch cage to hold bait and bedding. The trap mechanism is not spring loaded, but gravity controlled, and can be used for capturing more than one animal.

REFERENCES

Aplin, K.P., Brown, P.R., Jacob, J., Krebs, C.J. and Singleton, G.R. (2003) *Field Methods for Rodent Studies in Asia and the Indo-Pacific.* Australian Centre for International Agricultural Research, Monograph No. 100, Canberra.

Arthur, A.D., Pech, R.P., Drew, A., Gifford, E., Henry, S. and McKeown, A. (2003) The effect of increased ground-level habitat complexity on mouse population dynamics. *Wildlife Research* **30**: 565–572.

Bomford, M. and Hart, Q. (2002) Non-indigenous vertebrates in Australia. In *Biological Invasions: Economic and Environmental Costs of Alien Plant, Animal, and Microbe Species.* Pimentel, D. (ed). CRC Press, Boca Raton: pp. 25–44.

Braysher, M. (1993) *Managing Vertebrate Pests: Principles and Strategies.* Bureau of Resource Sciences, Canberra.

Brown, P. and Singleton, G. (2001) Mouser v. 1.0. CD-ROM, CSIRO and the University of Queensland.

Brown, P.R. and Singleton, G.R. (2001) Relationship between mouse abundance and damage to crops. In *12th Australasian Vertebrate Pest Conference.* Department of Natural Resources and Environment, Melbourne: pp. 273–277.

Brown, P.R., Singleton, G.R., Kearns, B. and Griffiths, J. (1997) Evaluation and cost-effectiveness of strychnine for control of populations of wild house mice (*Mus domesticus*) in Victoria. *Wildlife Research* **24**: 159–172.

Buckland, S.T., Goudie, I.B.J. and Borcher, D.L. (2000) Wildlife population assessment: past developments and future directions. *Biometrics* **56**: 1–12.

Caughley, G. (1980) *Analysis of Vertebrate Populations*. John Wiley and Sons, Chichester. Reprinted with corrections.

Caughley, J., Bomford, M., Parker, B., Sinclair, R., Griffiths, J. and Kelly, D. (1998a) *Managing Vertebrate Pests: Rodents*. Bureau of Resource Sciences and Grains Research and Development Corporation, Canberra.

Caughley, J., Donkin, C. and Strong, K. (1998b) Managing mouse plagues in rural Australia. In *Proceedings of the 18th Vertebrate Pest Conference*. Baker, R.O. and Crabb, A.C. (eds). University of California, Davis: pp. 160–165.

Caughley, J., Monamy, V. and Heiden, K. (1994) *Impact of the 1993 Mouse Plague*. Grains Research and Development Corporation and Bureau of Resource Sciences, Canberra.

Caughley, J.A. (1998) *House Mouse (Mus domesticus) in Queensland*. Natural Resources and Mines, Queensland.

Chambers, L.K., Singleton, G.R. and Krebs, C.J. (2000) Movements and social organization of wild house mice (*Mus domesticus*) in the wheat lands of Northwestern Victoria, Australia. *Journal of Mammalogy* **81**: 59–69.

Davis, S.A., Akison, L.K., Farroway, L.N., Singleton, G.R. and Leslie, K.E. (2003) Abundance estimators and truth: accounting for individual heterogeneity in wild house mice. *Journal of Wildlife Management* **67**: 634–645.

Edwards, G.P., Pople, A.R., Saalfield, K. and Caley, P. (2004) Introduced mammals in Australian rangelands: Future threats and the role of monitoring programmes in management strategies. *Austral Ecology* **29**: 40–50.

Elzinga, C.L., Salzer, D.W., Willoughby, J.W. and Gobbs, J.P. (2001) *Monitoring Plant and Animal Populations*. Blackwell Science, Malden, Massachusetts.

Engeman, R.M. and Witmer, G.W. (2000) IPM strategies: indexing difficult to monitor populations of pest species. In *Proceedings 19th Vertebrate Pest Conference*. Salmon, T.P. and Crabb, A.C. (eds). University of California, Davis: p. 183–189.

Goldsmith, F.B. (1991) *Monitoring for Conservation and Ecology*. Chapman and Hall, New York.

Hone, J. (1994) *Analysis of Vertebrate Pest Control*. Cambridge University Press, Cambridge.

Jacob, J., Ylönen, H. and Hodkinson, C.G. (2002a) Trapping efficiency of Ugglan traps and Longworth traps for house mice in south-eastern Australia. *Wildlife Research* **29**: 101–103.

Jacob, J., Ylönen, H., Perry, J.A. and Singleton, G.R. (2002b) Who eats first? uptake of pellet bait by target and non-target species. *International Biodeterioration and Biodegradation* **49**: 121–124.

Jacob, J., Ylönen, H., Runcie, M.J., Jones, D.A. and Singleton, G.R. (2003) What affects bait uptake by house mice in Australian grain fields? *Journal of Wildlife Management* **67**: 341–351.

Krebs, C.J. (1989) *Ecological Methodology*. Harper Collins Publishers, New York.

Krebs, C.J., Kenney, A.J. and Singleton, G.R. (1995) Movements of feral house mice in agricultural landscapes. *Australian Journal of Zoology* **43**: 293–302.

McLeod, R. (2004) *Counting the Cost: Impact of Invasive Animals in Australia*, 2004. Cooperative Research Centre for Pest Animal Control, Canberra.

Moro, D. (2001) Evaluation and cost-benefits of controlling house mice (*Mus domesticus*) on islands: an example from Thevenard Island, Western Australia. *Biological Conservation* **99**: 355–364.

Mutze, G.J. (1991) Mouse plagues in South Australian cereal-growing areas. III. Changes in mouse abundance during plague and non-plague years, and the role of refugia. *Wildlife Research* **18**: 593–604.

Mutze, G.J. (1993) Cost-effectiveness of poison bait trails for control of house mice in mallee cereal crops. *Wildlife Research* **20**: 445–456.

Mutze, G.J. (1998) The 1993 strychnine baiting program for mouse control in South Australian grain crops. I. Efficacy. *Wildlife Research* **25**: 533–546.

Mutze, G.J. and Hubbard, D.J. (2000) Effects of molluscicide baits on field populations of house mice. *Agriculture, Ecosystems and Environment* **80**: 205–211.

Olsen, P. (1998) Australia's Pest Animals: New Solutions to Old Problems. Bureau of Rural Sciences, Canberra.

Pollock, K., Nichols, J., Brownie, C. and Hines, J. (1990) Statistical inference for capture-recapture experiments. *Wildlife Monographs* **107**: 1–97.

Possingham, H. (2001) *The Business of Biodiversity*. Australian Conservation Foundation, Melbourne.

Redhead, T.D., Enright, N. and Newsome, A.E. (1985) Causes and predictions of outbreaks of *Mus musculus* in irrigated and non-irrigated cereal farms. *Acta Zoologica Fennica* **173**: 123–127.

Ruscoe, W.A., Goldsmith, R. and Choquenot, D. (2001) A comparison of population estimates and abundance indices for house mice inhabiting beech forests in New Zealand. *Wildlife Research* **28**: 173–178.

Ryan, G.E. and Jones, E.L. (1972) A report on the mouse plague in the Murrumbidgee and Colleambally Irrigation Areas, 1970. NSW Department of Agriculture

Saunders, G. (2000) *Early Detection of Mouse Plagues*. NSW Agriculture, Orange.

Saunders, G.R. (1983) Evaluation of mouse-plague control techniques in irrigated sunflower crops. *Crop Protection* **2**: 437–445.

Saunders, G.R. and Giles, J.R. (1977) A relationship between plagues of the house mouse, *Mus musculus* (Rodenti: Muridae) and prolonged periods of dry weather in south-eastern Australia. *Australian Wildlife Research* **4**: 241–247.

Schwarz, C.J. and Seber, G.A.F. (1999) Estimating animal abundance: review III. *Statistical Science* **14**: 427–456.

Seber, G.A.F. (1982) The Estimation of Animal Abundance and Related Parameters. Charles Griffin, London.

Sharp, T. and Saunders, G. (2005) *Humane pest animal control – codes of practice and standard operating procedures*. NSW Department of Primary Industries. Document available in print or electronically from www.dpi.nsw.gov.au/pubs/humane-research

Singleton, G.R. (1989) Population dynamics of an outbreak of house mice (*Mus domesticus*) in the mallee wheat lands of Australia—hypothesis of plague formation. *Journal of Zoology, London* **219**: 495–515.

Singleton, G.R. and Brown, P.R. (1999) Management of mouse plagues in Australia: integration of population ecology, bio-control and best farm practice. In *Advances in Vertebrate Pest Management*. Cowan, D.P. and Feare, C.J. (eds). Filander Verlag, Fürth: pp. 189–203.

Singleton, G.R. and Redhead, T.D. (1989) House mouse plagues. In *Mediterranean Landscapes in Australia. Mallee Ecosystems and their Management*. Noble, J.C. and Bradstock, R.A. (eds). CSIRO, Melbourne: pp. 418–433.

Singleton, G.R., Twigg, L.E., Weaver, K.E. and Kay, B.J. (1991) Evaluation of bromadiolone against house mouse (*Mus domesticus*) populations in irrigated soy beans crops. II. Economics. *Wildlife Research* **18**: 275–283.

Southwood, T.R.E. (1989) *Ecological Methods, 2nd Edition.* Chapman and Hall, London.

Thomas, L. (1996) Monitoring long-term population change: Why are there so many analysis methods? *Ecology* **77**: 49.

Twigg, L.E., Martin, G.R. and Stevens, T.S. (2002) Effect of lengthy storage on the palatability and efficacy of zinc phosphide wheat bait used for controlling house mice. *Wildlife Research* **29**: 141–149.

Walsh, A., Oliver, S. and Croft, D. (2000) Mouse Plague Survival Guide for Rural Lands Protection Board Staff. NSW Agriculture, Orange.