Guide to the identification and collection of New Zealand rodents

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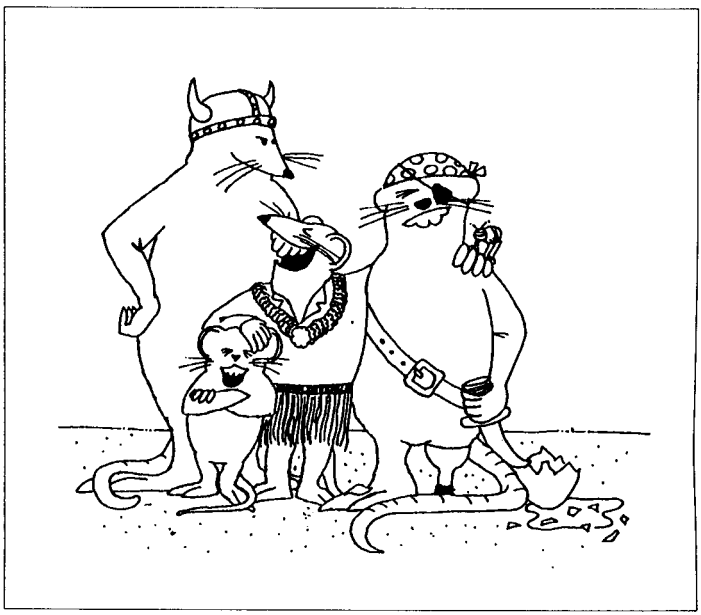

Preface to the Second Edition

Since the inception of the Department of Conservation in April 1987, knowledge of how to identify, monitor, control, and eradicate rodents has increased many-fold. During this time, demand for the First Edition of this guide, published by the New Zealand Wildlife Service in 1983, has also increased. The authors felt it timely to produce a Second Edition with up-to-date and new, relevant information which takes account of eradications, discoveries, and threats from potential new invaders. There is also new information on preservation of samples.

The Publications Section of Science and Research Division, Department of Conservation, agreed to re-publish the new edition in this format, to assist field staff of the department and local authorities with the practical and technical aspects of their work. When faced with rodents, we think this Second Edition will help people to answer the recurring question: "What have we got?"

D.M. Cunningham & P.J. Moors

Preface to the Third Edition

The second edition, published in May 1993, continued to satisfy an ever-present need for basic information to the extent that the print-run of 200 was used up in just over two and half years. With a clear need so abundantly demonstrated, this presented an opportunity to again expand the content of the guide. We have added identification features of rats' skulls, because this is a frequently-found and readily identifiable part of the animal which can tell us a lot about its owner. Mouse skulls have not been included as they are so obviously different from those of rats.

D.M. Cunningham & P.J. Moors

Introduction

This booklet was initially produced in response to requests from staff of the New Zealand Wildlife Service for a field guide to the identification and trapping of rats and mice. Our main aim, therefore, is to facilitate the collection of reliable information on the species, distribution, abundance, size and breeding condition of rodents. We also wish to strengthen general acceptance of standard names for the rodents in New Zealand, as well as a standardised system for trapping them.

The methods of identification, trapping and autopsy which we describe are an amalgam from the field experiences of ourselves and others in New Zealand. The techniques are straightforward, requiring care, common sense and no special skills. We hope this simplicity will stimulate people to make routine records of rodents wherever they are encountered. Additional information on specialised techniques, the ecology of rats and mice, and their history and effects in New Zealand can be gained from the selected references listed at the end of this booklet.

What is a rodent?

Rodents (the mammalian order Rodentia), are considered to be as numerous, both in numbers and species, as all other orders of mammals combined. They include such diverse animals as squirrels, guinea pigs, rats, porcupines, voles and lemmings. However in New Zealand they are represented only by four introduced species from the family Muridae (rats and mice).

A prominent feature of all rodents is the presence of a single pair of large gnawing teeth in each of the upper and lower jaws. These incisor teeth grow throughout the animal's life and have to be continually worn down by gnawing. A long gap separates the incisors from the cheek or molar teeth. Adult rats and mice have a maximum of three molars in each tooth row, compared with at least four molars in other rodents.

New Zealand rodents

The four species of rodent in New Zealand are:

KIORE *Rattus exulans* (Polynesian rat, Maori rat, Pack rat) Introduced by early Polynesian settlers. Widespread prior to European settlement, but confined now to Fiordland and many offshore islands (including Stewart, Kapiti, Great Barrier, Macaulay and Raoul, and Chatham Island).

NORWAY RAT *Rattus norvegicus* (brown rat, water rat, sewer rat) Introduced late 18th century, established and common throughout the country by the 1850s. Now common only in wet habitats, urban areas and on some offshore islands (including Campbell, Stewart, Kapiti, Raoul, Chatham, but not Great Barrier).

SHIP RAT *Rattus rattus* (black rat, blue rat, bush rat, roof rat) Introduced into the North Island about the 1860s and the South Island in the 1890s; spread rapidly. Found in most habitats, and is now the most abundant and widespread rat on mainland New Zealand; also found on some offshore islands (Stewart, Chatham, Great Barrier, but not New Zealand subantarctic islands, Kapiti, Kermadecs).

HOUSE MOUSE *Mus musculus* (field mouse) Established in the Bay of Islands about 1830, reached South Island after 1852. Now common throughout mainland New Zealand from shoreline to snowline; also on some offshore islands (including Antipodes, Auckland, Chatham, and Great Barrier. Not on Campbell, Stewart, Mana, Kapiti, Kermadecs).

These rodents (especially the rats) are known to prey on many native animals, often causing a population decline or even extinction. The corresponding impact on native plants is largely unknown, but may also be considerable. Therefore it is essential that rodents do not reach any places currently lacking them, and that additional species do not become established in areas already having one or two species. Visitors to rodent-free islands must be especially vigilant with precautions against the inadvertent introduction of rats or mice (see Moors et al. 1989).

Rodent identification

The four New Zealand rodent species mentioned above can be identified using an aggregate of the features listed in Table 1 (next page).

ADDITIONAL IDENTIFICATION NOTES

(The numbers below refer to Table 1)

1. If identification is in doubt, always keep and preserve at least the head for later detailed examination. Body measurements (see Autopsy Procedures) are also useful.
2. Juvenile rats are sometimes difficult to identify and distinguish from mice, but the species can usually be separated on the basis of tail length, fur colour, hind foot and ear characteristics. If in doubt, keep the whole specimen either frozen or in excess 75% alcohol with the gut cavity opened.
3. The normal maximum weight and head-body length are given for each species. However, larger kiore may occasionally be encountered (e.g., on Lady Alice Island, Hauraki Gulf), maximum values are about 190 g and 185 mm.
4. There are three colour forms or morphs (not subspecies) of *Rattus rattus:*

   (a) "rattus" uniformly black back (sometimes has a blueish look); uniformly grey belly.

   (b) "alexandrinus" brown back with long black guard hairs; uniformly grey belly.

   (c) "frugivorous" brown back with long black guard hairs; uniformly white or creamy-white belly.

TABLE 1 IDENTIFICATION OF NEW ZEALAND RODENTS.

	HOUSE MOUSE *Mus musculus*	KIORE *Rattus exulans*
Normal adult weight	Up to 28 g	Up to 187 g (see note 3)
Max. head-body length (HBL)	101 mm	185 mm (see note 3)
Tail length	Slightly shorter or longer than HBL. Uniformly grey-brown.	Slightly shorter or longer than HBL. Thin and uniformly dark.
Ears	12.0-15.0 mm	15.5-20.5 mm, cover eyes when pulled forward. Fine hairs do not extend beyond edge of ear.
Adult hind foot	15.0-21.0 mm, small and thin	24.5-31.0 mm
Colour of upper side of hind foot	Uniformly grey	outer edge dark near ankle, rest of foot and toes pale.
Fur on back	Dull grey-brown	Brown
Fur on belly	Uniformly grey	White-tipped grey giving irregular colour.
Length of droppings	3.9-7.6 mm	6.4-9.0 mm
Number of nipples	10-12	8
Habits	Mainly ground-dwelling though capable climber; nests in small holes.	Agile climber; digs small holes, nests on ground; or in trees, feeds on ground and in trees; infrequent swimmer.

TABLE 1 *CONTINUED*

SHIP RAT *Rattus Rattus*	NORWAY RAT *Rattus norvegicus*
Up to 215 g	Up to 450 g
230 mm	275 mm
Much longer than HBL. Uniformly coloured	Clearly shorter than HBL. Thick with pale underside
19.0-26.0 mm, cover eyes when pulled forward. Fine hairs do not extend beyond edge of ear.	14.0-22.0 mm, do not cover eyes when pulled forward. Obvious hairs extend beyond edge of ear.
28.0-38.0 mm	30.0-41.5 mm
Uniform colouring over whole foot, usually dark	Always completely pale
Brown or black (see note 4	Brown
Uniform monotone of grey, white or creamy-white. (see note 4)	White-tipped grey giving irregular colour.
6.8-13.8 mm	13.4-19.1 mm
10-12, usually 10	12
Very agile and frequent climber; rarely burrows; nests mainly in trees and shrubs; infrequent swimmer.	Burrows extensively; climbs much less frequently than other rats; strong swimmer; nests underground; very wary.

References to note numbers within this Table refer to Additional identification notes in the main text.

RAT SKULLS

Distinguishing the three primary features (refer to numbers on Figure 1).

1. **Posterior extension of the zygomatic arch**

   NORWAY RAT: mostly non-existent

   SHIP RAT: very pronounced ridge extending back from the end of the arch. Noticeable even in Age Class 1 juveniles*

   KIORE: slightly pronounced

2. **Temporal ridges** (on the top corners above eye sockets)

   NORWAY RAT: slightly raised although more pronounced in older rats but only above orbits

   SHIP RAT: very pronounced particularly in older rats, extending well rear of orbits

   KIORE: slightly raised in older rats

3. **Holes in the floor of the skulls** between the teeth and the auditory bulla

   NORWAY RAT: two obvious pairs of holes

   SHIP RAT: two obvious pairs of holes

   MORE: one pair of holes - anterior pair non-existent

Adult ship rat skulls are generally more rounded than those of Norway rats which tend to be rectangular. Use a pair of Vernier calipers across the widest part of the cranium just above where the zygomatic arch makes contact. Hold the calipers horizontally and approach from the back end of the skull. If the skull is wider than the ridges at the back of the skull, it is a ship rat. If you cannot actually contact the cranium sides because of the ridges surrounding the foramen magnum (the large hole at the base of the skull where the spinal cord comes out), it is a Norway rat.

* Approximate age can be estimated from the amount of molar wear (Karnoukhova 1971). Note the wear on the molars relative to the size of the skull, e.g. well-worn molars on a small skull will indicate kiore.

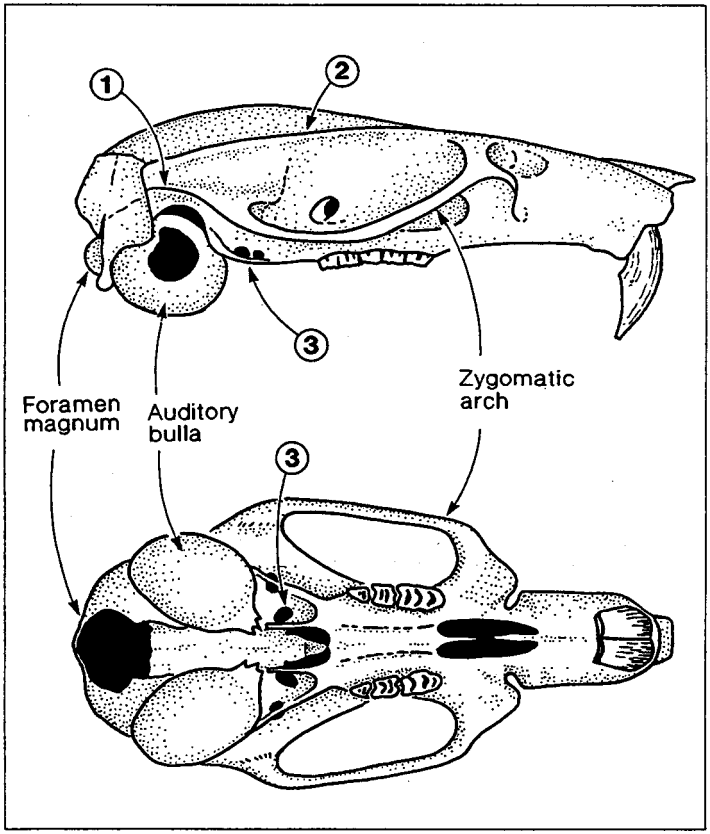

Figure 1 Diagram of a rat skull showing primary identification features (see text for explanation of the numbers).

Potential new invaders

Two more rodents which are not yet present in New Zealand are also real threats to our native biota. If you ever suspect that you have either one of these animals, freeze the specimen and send it **immediately** to D.M. Cunningham, Science & Research Division, Department of Conservation, PO Box 10-420, Wellington, for confirmation.

ASIAN SHIP RATS (*Rattus rattus flavipectus*)
Concern that a new subspecies of ship rat could find its way to New Zealand on board Asian and Russian ships prompted a brief survey in 1982-83 to determine if this was indeed the case.

The "Asian" ship rat differs principally from its "Oceanian" (present in New Zealand) counterpart in the chromosome count. Asian, 42 chromosomes, Oceanian, 38. Interbreeding is unknown and where the two subspecies occur sympatrically, there is clear habitat and prey separation. On islands where Asian ship rats arrive after the already established Oceanian rats, the new arrivals exploit prey species which have survived the impact of the earlier predators. This has very serious implications for native biota already under pressure from pressure from existing threats.

So far no Asian ship rats have been found in New Zealand except on infested vessels fumigated by the health authorities. All specimens recovered were distinguishable from the local subspecies by their distinct tawny colouring suffused throughout the background colours of "alexandrinus" and "frugivorous" colour morphs.

Any ship rats with a tawny appearance should be preserved, preferably by freezing, and sent for identification. Further trapping may be necessary to determine the extent of the invasion.

MUSK SHREW (*Suncus murinus*)

A single specimen of this species was recovered from a fumigated Asian fishing vessel in 1983. Musk shrews are now distributed from East Africa and Madagascar to Papua New Guinea from their original range of India to Indonesia.

Shrews are not rodents, but are insectivores. The establishment of this species, a successful coloniser elsewhere, would add to the two other types of insectivorous mammals in New Zealand: hedgehogs and bats. The threat to our invertebrate biota is obvious and potentially enormous.

Musk shrews are well known throughout their present range for their association with human habitation. They are regarded as a nuisance because of their characteristic smell, their noise, destructive habits, and spoiling of food.

Shrews have long heads with long, pointed noses, and small eyes. Unusual small mammals that do not look like mice, which are trapped or handed in by health authorities should be sent to D.M. Cunningham at Science and Research Division, Dept. of Conservation, PO Box 10-420, Wellington, **immediately**.)

Rodent trapping

SURVEY TRAPPING

This method is used to determine which species are present. It does not provide information on abundance.

INDEX TRAPPING

This technique was originally developed in New Zealand by Ecology Division, Department of Scientific and Industrial Research, as a standardised method for the regular sampling of rodents in the same habitat. In recent years it has been widely used in New Zealand to obtain information on both the identification and the abundance of rodents. The method allows for the calculation of an index of abundance, which can be used to compare populations in different regions, habitats, or seasons.

TRAPS

Ezeset "Supreme" rat and mouse traps are the types most frequently used for both survey and index trapping. They are readily available from most hardware shops. Always use the same brand of trap for all index trapping.

On new traps the end of the trigger arm often needs filing smooth for maximum sensitivity. Never leave traps out when not in use because the springs rust and weaken. Treat all metal parts with any proprietary brand of fish oil. Traps must be tied down so that injured rats and large mice do not drag them away, or are themselves dragged away by scavenging predators.

TRAP-SITES AND SETTING

Spacing between sites and the number of traps per site is not important in survey trapping. Place traps where there is plenty of natural cover and where rodents are likely to be active (e.g., alongside large rocks, around the bases of trees, under logs and overhanging vegetation). If rodent droppings, food remains or runways are visible, set traps nearby.

An index line should consist of at least 25 sites evenly spaced apart with two traps per site. The actual number of trap-sites will depend on the number of traps, the personnel available and the size of the habitat being sampled. Plan index trapping so as to give a minimum of 100 corrected trap-nights (see Results, below) in each habitat (e.g., 50 traps for three nights gives a maximum of 150 trap-nights). Index lines are usually run for three nights.

The spacing between sites should be as large as possible within the range of 25-50 m. Measure the intervals by pacing or preferably with a tape measure. Permanent index lines should be accurately measured. Place traps near or under natural cover whenever possible. Rodents are discouraged if traps move, so ensure that the traps are level and stable. This is especially important when Norway rats are present.

Set two traps at each site. This increases the potential capture rate (useful when rodent numbers are high) and also doubles the number of trap-nights for little extra effort. The traps may be placed up to a metre apart. When both rats and mice are present set a mouse and a rat trap at each site, and if only rats or only mice are present set two traps of the appropriate type.

COVERS

Covers for traps are not always necessary, but they should be used if non-target animals are likely to be caught, and to limit trap disturbance. Covers can be made of whatever material is suitable, available and reasonably portable (e.g., wire mesh, clear plastic sheet, or plastic drain pipe). Stones, sticks or wire pegs will hold covers in place. Bent wire hoops should be placed across the entrances to exclude non-target animals.

To maintain consistency during index trapping, covers should either be present at every site or be absent from every site, and, as there are indications that the type of cover may influence trapping success, should be left in place between trapping sessions to accustom the animals to their presence. If covers are used, both traps may be set back to back under the one cover. A wire hoop or forked stick placed in the space between them helps prevent one setting off the other. Ensure that the cover does not impede the action of the traps.

BAIT

During survey trapping use whatever bait catches rodents. If time is available, experiment with several baits because species and individuals vary in their preferences.

A stiff mixture of peanut butter and rolled oats is a recommended and reliable standard bait for index trapping, and it lasts well. Other suitable baits include cheese, nuts, chocolate, bacon, or leather soaked in fish oil. It is acceptable to use an alternative to peanut butter on one trap at each site, providing this is done at every site for consistency. Renew the bait whenever its attractiveness has been reduced (e.g., by rain, hot weather, mould, or partial consumption by ants or other insects).

RESULTS

Check the traps as early as possible each morning because tissue deteriorates quickly in warm weather, and carcasses become fly-blown. Make a brief note of the previous night's weather as this can affect the animals' behaviour and influence the trapping results. Record whether each trap is sprung or unsprung, and whether the bait has been removed, partly eaten, or left untouched. Also note any disturbance by other animals. For example, your field notes may read:

Line A 16/6/96 Pureora Forest.
Wet, windy, cold night.
Site 1. OK/Sp.B.OK (i.e., 1 trap unsprung, bait OK/1 trap sprung, bait OK)
2. 2 OK (i.e., both traps unsprung, bait OK)
3. RAT/OKB.G. (i.e., 1 capture/1 trap unsprung, baitgone)
4. RAT/Sp. B. ½G. (i.e., 1 capture/1 trap sprung, bait half gone).

The next step is to calculate the corrected number of trap-nights by making allowance for all those traps which had been set off. Subtract half a night for each of those traps (whether or not they had caught a rodent) on the assumption that each will have been sprung for an average of half the night. Do not make a correction for unsprung traps with the bait removed because they were still capable of catching a rodent and may actually do

so. The index of abundance is calculated at the end of the trapping session from the total number of rodents caught and the total number of corrected trap-nights, and is expressed as the number of captures per 100 trap-nights. In the following sample calculation, 7 rats have been caught and 13 traps sprung without catching anything:

50 traps set for 3 nights $= 50 \times 3$

$= 150$ Total trap-nights

Trap-nights lost $= {}^{1}/_{2}$ (captures + sprung, empty traps)

$= \frac{7 + 13}{2}$

$= 10$

Therefore the corrected number of trap-nights $=$ Total trap-nights — trap-nights lost

$= 150 - 10$

$= 140$

Index of abundance $= \frac{\text{Captures} \times 100}{\text{Corrected trap-nights}}$

$= \frac{7 \times 100}{140}$

$= 5.0$ captures/100 trap-nights.

Autopsy procedures

INSTRUMENTS

The following equipment is needed: a pair of vernier callipers and a ruler or tape measure; accurate spring balances (e.g., Pesola brand) able to weigh up to 500 g in 2 g steps; a pair of sharp scissors and a pair of forceps; small tie-on labels (available from stationery shops) and a pencil for labelling; and an adequate supply of preservative, usually 75% alcohol. Reproductive systems and stomachs should be preserved in 25 ml glass vials; individually labelled rat skulls can be preserved together in larger containers (e.g., 250 ml plastic water-tight jars).

WHAT INFORMATION TO RECORD

Measurements

- **Head-Body Length (HBL).** Place the animal flat on its back on a ruler or tape measure. Some careful flexing may be necessary with stiff, bent animals. Measure to the nearest millimetre from the tip of the nose to where a mounted needle (or other thin, pointed instrument) slid along the side of the tail is stopped by the pelvis.
- **Tail Length.** Measure to the nearest mm from the tip of the tail (excluding hairs) to the rear edge of the pelvis described above.
- **Right Hind Foot and Right Ear.** These are helpful measurements for separating species. Both must be taken with a vernier calliper to 0.1 mm. Flatten the toes between finger and thumb and measure the hind foot from the tip of the heel to the tip of the middle toe (excluding claw). The ear is measured from the lowest point of the basal notch to the furthest extremity, excluding hairs.
- **Weight.** Weigh with a spring balance to the nearest gram BEFORE dissecting, skinning or preserving the animal. Note if it is wholly or partly wet as dampness affects the weight.

Sex

Always record the sex of the animal. Juvenile males and females are sometimes difficult to separate as external sexual features can be similar. However, the distance between the anus and the urethral opening is greater in males than females. In addition, the vagina in very young females is completely covered with a translucent layer of skin. This appears as a small bald patch immediately to the rear of the urethral opening. See Figure 2.

Reproductive systems

If you are uncertain of the arrangement of rodent reproductive systems, consult a dissection manual (e.g., Rowett 1960) or refer to the diagrams of the female reproductive tract in Brown and Stoddart (1977).

- **Females**. Record if the vagina is open or closed (see Figure 1). In very young females the closed condition is usually quite distinct. In older animals it is usually obviously open or can easily be opened with a probe. This latter condition is still regarded as "open" or "perforate".

  Nipples occur only on females and should be carefully counted when visible; this is especially important with ship rats which often have an extra nipple or two. If nipples are large and there is very little hair around each one, check for lactation by attempting to express milk and by examining the development of mammary tissue underneath.

- **Males**. Note whether or not the testes are "scrotal" (in the scrotum). When captured, mature males sometimes retract the testes, but the presence of a dark bald patch on the scrotum is usually a good sign that the testes are normally scrotal.

General

Always note the colour morph of ship rats because the frequencies of the three morphs vary considerably throughout New Zealand. Recording the colour morph can help when there is doubt about identification. Make a brief note of fur condition and any injuries. If you have opened the gut cavity, note the amount of fat around the gut as "none, little, medium or heavy".

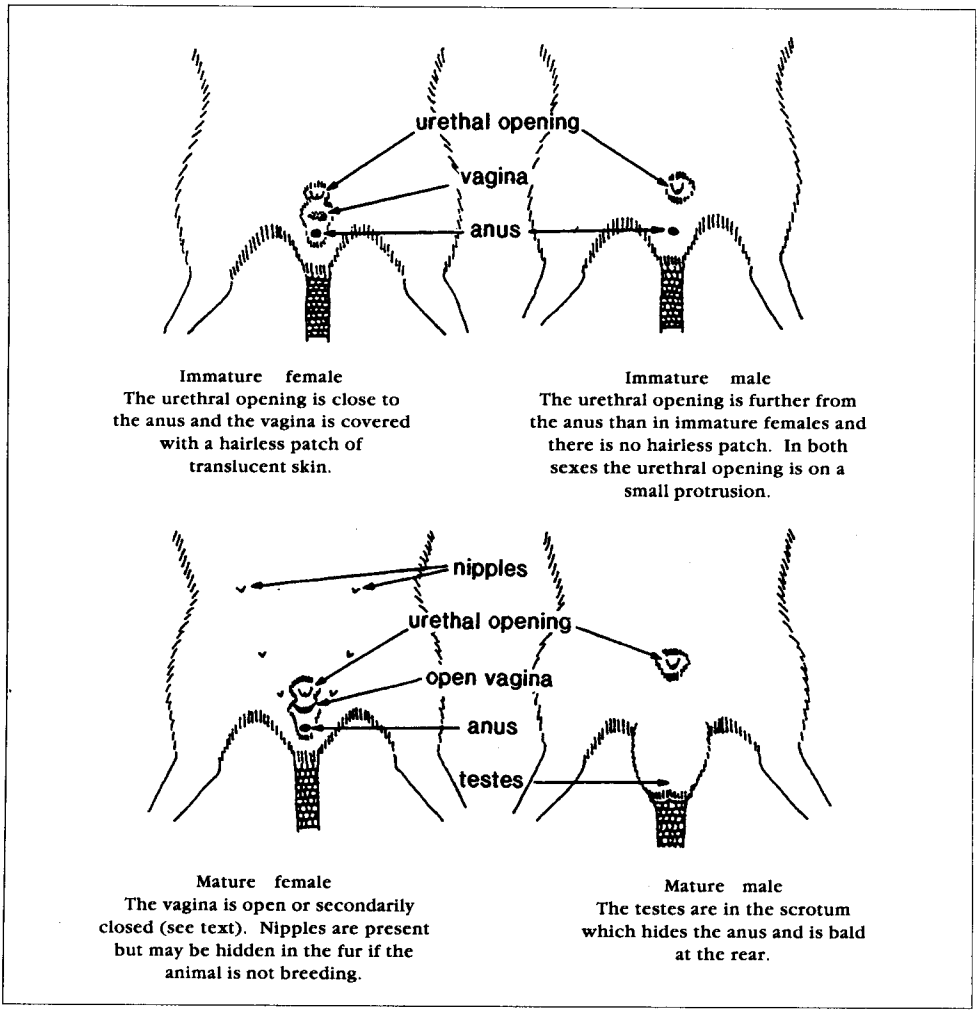

Figure 2 Comparison of external sexual features of immature and mature rats.

WHAT TO KEEP

If you are carrying out a detailed population study, preserve reproductive systems, stomachs and skulls. If, however, you are interested only in the abundance of a population and are certain of the identity of your animals, there is no need to keep any material. Unwanted samples have a habit of sitting around for years before being eventually dumped. If you are uncertain

of the identity or the specimens are from a rarely visited locality, then preserve at least the skulls (as skinned heads) because they are the single most useful piece of skeletal material for age-determination and species identification.

Keep any fleas and lice found in the fur in 75% alcohol, in a separate vial for each animal. Ectoparasites on whole preserved animals will fall off and collect on the bottom of the jar. This material should be kept.

Preservation of samples

Freezing is the best way of preserving samples for later examination. However, it is not suitable for long-term storage unless the samples have been completely sealed in polythene bags to prevent desiccation. There are two basic fixatives (i.e., preservatives) for general use.

75% ALCOHOL

Usually obtained commercially as 96% isopropyl alcohol. Dilute 4 parts alcohol with 1 part water. Always use in excess, that is, at least an equal volume of alcohol to material. Open the gut cavity of whole animals to ensure rapid and complete penetration by the fixative. Do not fill the container with samples and then fill the remaining space with fixative: there will be insufficient fixative to reach inner tissues, which will then decay. If you are short of containers and have to condense your samples, replace the alcohol at least twice within the first week. The cost of alcohol and containers is minimal compared with the cost of obtaining those samples.

10% FORMALIN

The commercial stock liquid is a 40% solution of formaldehyde. Dilute 1 part stock with 9 parts water. Formalin is not a convenient general fixative as it hardens the tissue and makes later examination of carcasses a most unpleasant task. It is generally used for specialised requirements.

Although ethyl alcohol is more pleasant and safer to handle, isopropyl alcohol is easier and cheaper to obtain. As ethyl alcohol is the alcohol of alcoholic drinks, its sale in useful quantities is very strictly controlled. With a permit, isopropyl alcohol is readily available from the petro-chemical manufacturers. However, isopropyl alcohol is toxic in smaller amounts and prolonged, careless handling can lead to skin irritation. NEVER consume isopropyl alcohol, not even diluted.

In an emergency you can use vinegar, methylated spirits or a very strong salt solution for a short time. This must be replaced by 75% alcohol as soon as possible. Whatever you use, it MUST be water soluble in order to penetrate body tissues.

LABELLING

Ensure that all labels are clearly written with pencil NOT ink on plain white paper or card. Inadequately identified samples are worthless. Label frozen specimens with tags which will not disintegrate with thawing. Vials and jars should contain a label plus a reference number on the lid. Simply writing on the side or lid of the container is asking for trouble, as any spilt alcohol will dissolve what you have written.

Finally, when you have finished, WASH YOUR HANDS thoroughly. Although most wild rats are not health hazards, some carry diseases such as leptospirosis.

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Source literature

*Barnett, A. and Dutton, J. 1995. Expedition field techniques: Small mammals (excluding bats). Expedition Advisory Centre, London.

Brown, J. Clevedon and Stoddart, D.M. 1977. Killing mammals and general post-mortem methods. Chapter 7 in: Techniques in mammalogy. *Mammal review* 7: 63-94.

*Corbett, G.R. and Southern, H.N. 1977. Handbook of British mammals. (2nd ed.). Blackwell, Oxford.

Cunningham, D.M., and Moors, P.J. 1983 A guide to the identification and collection of New Zealand rodents. *Occasional publication No 4.* N.Z. Wildlife Service, Dept. of Internal Affairs, Wellington.

Cunningham, D.M., and Moors, P.J. 1993. A guide to the identification and collection of New Zealand rodents. (2nd ed.) Dept. of Conservation, Wellington.

*Dingwall, P.R., Atkinson, I.A.E. and Hay, C. (Eds) 1978. The ecology and control of rodents in New Zealand nature reserves. *Department of Lands and Survey Information Series Vol. 4.*

*Flowerdew, J.R. 1976. Ecological methods. Chapter 4 in: Techniques in mammalogy. *Mammal review* 6:123-159.

*Innes, J., Warburton, B., Williams, D., Speed, H. and Bradfield, P. 1995. Large-scale poisoning of ship rats (*Rattus rattus*) in indigenous forests of the North Island, New Zealand. *N.Z. journal of ecology* 19(1): 5-17.

Karnoukhova, N.G. 1971. Age determination of brown and black rats. *Soviet journal of ecology* 2: 144-147.

*King, C.M. 1990. The handbook of New Zealand mammals. Oxford U.P., Auckland.

Moors, P.J., Atkinson, I.A.E., and Sherley, G.H. 1989. Prohibited immigrants: the rat threat to island conservation. World Wildlife Fund for Nature, Wellington.

Nelson, L. Jr. and Clark, F.W. 1973. Correction for sprung traps in catch/effort calculations of trapping results. *Journal of mammology* 54: 295-298.

Rowett, H.G.Q 1960. The rat as a small mammal. (2nd ed.). John Murray, London.

*Twigg, G.I. 1975. Catching mammals. Chapter 2 in: Techniques in mammalogy. *Mammal review* 5: 83-100.

* Items for further reading.