Inventory Methods for Small Mammals: Shrews, Voles, Mice & Rats

Standards for Components of British Columbia's Biodiversity, No. 31

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Preface

This manual presents standard methods for inventory of shrews, mice, voles and rats in British Columbia at three levels of inventory intensity: presence/not detected (possible), relative abundance, and absolute abundance. The manual was compiled by the Elements Working Group of the Terrestrial Ecosystems Task Force, under the auspices of the Resources Inventory Committee (RIC). The objectives of the working group are to develop inventory methods that will lead to the collection of comparable, defensible, and useful inventory and monitoring data for the species component of biodiversity.

This manual is one of the Standards for Components of British Columbia's Biodiversity (CBCB) series which present standard protocols designed specifically for group of species with similar inventory requirements. The series includes an introductory manual (Species Inventory Fundamentals No. 1) which describes the history and objectives of RIC, and outlines the general process of conducting a wildlife inventory according to RIC standards, including selection of inventory intensity, sampling design, sampling techniques, and statistical analysis. The Species Inventory Fundamentals manual provides important background information and should be thoroughly reviewed before commencing with a RIC wildlife inventory. RIC standards are also available for vertebrate taxonomy (No. 2), animal capture and handling (No. 3), and radio-telemetry (No. 5). Field personnel should be thoroughly familiar with these standards before engaging in inventories which involve either of these activities.

Standard data forms are required for all RIC wildlife inventory. Survey-specific data forms accompany most manuals while general wildlife inventory forms are available in the Species Inventory Fundamentals No. 1 [Forms] (previously referred to as the Dataform Appendix). This is important to ensure compatibility with provincial data systems, as all information must eventually be included in the Species Inventory Datasystem (SPI). For more information about SPI and data forms, visit the Species Inventory Homepage at: http://www.env.gov.bc.ca/wld/spi/ric_manuals/

It is recognized that development of standard methods is necessarily an ongoing process. The CBCB manuals are expected to evolve and improve very quickly over their initial years of use. Field testing is a vital component of this process and feedback is essential. Comments and suggestions can be forwarded to the Elements Working Group by contacting:

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The Resources Inventory Committee consists of representatives from various ministries and agencies of the Canadian and the British Columbia governments as well as from First Nations peoples. RIC objectives are to develop a common set of standards and procedures for the provincial resources inventories, as recommended by the Forest Resources Commission in its report "The Future of our Forests".

For further information about the Resources Inventory Committee and its various Task Forces, please contact:

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All decisions regarding protocols and standards are the responsibility of the Resources Inventory Committee. The current version of this manual was the result of the hard work and expertise of Markus Merkens (PAW Research Services) with valuable input from Dr. Charles Krebs (UBC), John Boulanger (Integrated Ecological Research), Todd and Linda Zimmerling (Applied Ecosystem Management), Dr. Tom Sullivan (Applied Mammal Research Institute) and Mark Fraker (TerraMar Environmental Research). Specialized methods for water shrews were provided by Lisa Hartman (Ministry of Environment, Lands and Parks).

Some of the background information and protocols presented in this document are based on Version 1.1 of this manual and the unpublished government report, *Inventory Techniques for Small Mammals*, prepared for the Resources Inventory Committee by Laura Darling (Ministry of Environment, Lands and Parks) with assistance from Ruth van den Driessche and Tom Ethier.

The Standards for Components of British Columbia's Biodiversity series is currently edited by James Quayle with data form development by Leah Westereng.

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1. INTRODUCTION

The small mammals covered in this manual are taxonomically very diverse. They include: mice, voles and shrews whose habits and habitats can be quite dissimilar. Despite the differences in their natural history, populations of small mammals in this inventory group are censused using similar sampling techniques. However, their behaviours can complicate efforts to estimate their numbers. Variations in the general census technique are necessary to account for differences such as activity periods or habitat conditions.

This manual provides descriptions of each species in the small mammal inventory group (Section 2). Consideration for survey design and recommendations for presence/not detected, relative abundance and absolute abundance are summarized (Section 3) along with details of office and field procedures and equipment for each inventory objective. The specifics of sampling design in a study of a particular species will have to take into account the natural history and status of that species. Studies of the diversity of small mammal species in a community will have to be designed to address and acknowledge species differences in status, biology and trappability.

2. INVENTORY GROUP

There are 32 species within 2 Orders and 4 Families (one of which has 3 subfamilies) in the small mammal inventory group covered here (Table 1). They consist of herbivores, insectivores, granivores or omnivores with habitats that range from dry grasslands to old-growth forests to moist riparian areas. To escape predators, they have developed a host of defensive, elusive or protective behaviours. Some of these mammals are diurnal while others are crepuscular or nocturnal. A few are ubiquitous throughout British Columbia, while others have ranges restricted to small portions of the province or very specialized habitats. Some species can be captured throughout the year whereas others have distinct active seasons outside of which they are difficult or impossible to census. Ecologically speaking, mice, voles and shrews are a major food source for animals higher on the food chain, including avian, mammalian and reptilian predators. Generally, all of the small rodents and insectivores included in the group produce relatively large numbers of young over a short life time.

Table 1. Species covered in the small mammal inventory group.

Order **INSECTIVORA** / Family **SORICIDAE**: **Shrews**

Common name	**Scientific name**	**Species code**	**Status in B.C.**[1]
Black-backed Shrew	*Sorex arcticus*	SOAR	Yellow
Pacific Water Shrew	*Sorex bendirii*	SOBE	Red
Common Shrew	*Sorex cinereus*	SOCI	Yellow
Pygmy Shrew	*Sorex hoyi*	SOHO	Yellow
Dusky Shrew	*Sorex monticolus*	SOMO	Yellow
Water Shrew	*Sorex palustrus*	SOPA	Red
Trowbridge's Shrew	*Sorex trowbridgii*	SOTR	Blue
Tundra Shrew	*Sorex tundrensis*	SOTU	Red
Vagrant Shrew	*Sorex vagrans*	SOVA	Yellow

[1] All species formerly ‘not listed’ are now on the Yellow list.

Order **RODENTIA** / Family **HETEROMYIDAE**: **Heteromyids**

Common name	**Scientific name**	**Species code**	**Status in B.C.**
Great Basin Pocket Mouse	*Perognathus parvus*	PEPA	Blue

Order **RODENTIA** / Family **MURIDAE /** Subfamily **ARVICOLINAE: Voles and Lemmings**

Common name	**Scientific name**	**Species Code**	**Status in B.C.**
Southern Red-backed Vole	*Clethrionomys gapperi*	CLGA	Blue[2], Red[2]
Northern Red-backed Vole	*Clethrionomys rutilus*	CLRU	Yellow
Brown Lemming	*Lemmus sibiricus*	LESI	Yellow
Long-tailed Vole	*Microtus longicaudus*	MILO	Yellow
Montane Vole	*Microtus montanus*	MIMO	Yellow
Tundra Vole	*Microtus oeconomus*	MIOE	Yellow
Creeping Vole	*Microtus oregoni*	MIOR	Yellow
Meadow Vole	*Microtus pennsylvanicus*	MIPE	Yellow
Water Vole	*Microtus richardsoni*	MIRI	Yellow
Townsend's Vole	*Microtus townsendii*	MITO	Red[2]
Heather Vole	*Phenacomys intermedius*	PHIN	Yellow
Northern Bog Lemming	*Synaptomys borealis*	SYBO	Red[2]

Order **RODENTIA** / Family **MURIDAE**/ Subfamily **MURINAE: Old world rats and mice**

Common name	**Scientific name**	**Species code**	**Status in B.C.**
House Mouse	*Mus musculus*	MUMU	Yellow
Norway Rat	*Rattus norvegicus*	RANO	Yellow
Black Rat	*Rattus rattus*	RARA	Yellow

[2] Indicate designation as either Red- or Blue-listed at subspecies level only (see individual species descriptions in Section 2.0 for details).

Order **RODENTIA** / Family **MURIDAE**/ Subfamily **SIGMODONTINAE: New world rats and mice**

Common name	**Scientific name**	**Species code**	**Status in B.C.**
Deer Mouse	*Peromyscus maniculatus*	PEMA	Yellow
Columbian Mouse[3]	*Peromyscus oreas*	PEOR	Yellow
Sitka Mouse[3]	*Peromyscus sitkensis*	PESI	Yellow
Western Harvest Mouse	*Reithrodontomys megalotis*	REME	Blue

Order **RODENTIA** / Family **DIPODIDAE**: **Jumping Mice and Jerboas**

Common name	**Scientific name**	**Species code**	**Status in B.C.**
Meadow Jumping Mouse	*Zapus hudsonius*	ZAHU	Blue[2]
Western Jumping Mouse	*Zapus princeps*	ZAPR	Yellow
Pacific Jumping Mouse	*Zapus trinotatus*	ZATR	Yellow

The following species descriptions are intended to assist in the design of an inventory project. Although a good starting point, additional references should be consulted. The following references have been used to develop the species descriptions for species covered in this manual and are cited by their number in this list. Some additional references are listed under individual species. An asterisk (*****) indicates a primary reference for species biology, habitat use, activity and status; a greater-than sign (>) indicates a reference not reviewed but cited in Stevens and Lofts (1988).

1. Nagorsen 1990
2. Cowan and Guiget 1975
3. Burt and Grossenheider 1964
4. van Zyll de Jong 1983
5. Stevens and Lofts 1988
6. Sadoway 1986
7. Eisenberg 1964
8. Whitaker and Maser 1976
9. Belk *et al.* 1990
10. Terry 1978
11. Hawes 1977
12. Brown 1967
13. Banfield 1974
14. 1996 Provincial Red and Blue Lists (Ministry of Environment, Lands & Parks)

[3] The Columbian Mouse *(Peromyscus oreas)* and the Sitka Mouse *(Peromyscus sitkensis)* may be referred to as one species, Keen's Mouse (*Peromyscus keeni)*, as found in Hogan *et. al* (1993).

15. Stevens draft/1992
16. Maser and Storm 1970
17. Forsyth 1985

Information on species is presented under the following headings:

- Synonyms
- Other common names
- Distribution
- Status
- Habitat requirements
- Daily activity and movement patterns
- Seasonal activities and movement patterns
- Reproductive parameters
- Food habits
- Other

2.1 Insectivora / Soricidae: Shrews

2.1.1 Black-backed Shrew *(Sorex arcticus)*

Synonyms

Sorex richardsonii [1]

Other common names

Arctic Shrew, Saddle-backed Shrew [1]

Distribution

Found throughout the boreal forest of Canada, and restricted to the north-east corner of B.C. in the Peace River area [1,2,3,4]. Biogeoclimatic zone: Boreal White and Black Spruce. Ecoregion: Alberta Plateau, Fort Nelson Lowland.

Status

Not in jeopardy, not protected [14].

Habitat requirements

In B.C., in tamarack and spruce swamps [3], in marshy areas and along the edge of muskegs [2], on the edges of sphagnum bogs and marshes, often on the edge of an alder or willow shrub zone [13], in grass-sedge marshes, meadows, willow-alder copses, and to a lesser extent, in forests, and black spruce and larch bogs [4]. Favours a slightly drier habitat than other *Sorex* species [13, 4].

Daily activities and movement patterns:

Active through day and night [4,13], with main activity during the night [4]. May be territorial, home range estimated at 0.10 acre [13] and 0.59 ha [4].

Seasonal activities and movement patterns

No information found.

Reproductive parameters

Breeding period probably from May to August [2,4]. Number of embryos range from 5-10, mean 6.0-6.3 [3,4,13], and 2 [13] or 3 [4] litters per year may be produced. Depending on resources, may breed at 4-5 months old [4] although this may be rare [13]. Mortality rate of 80% in first 3-4 months [4]; life expectancy of 15 [4] to 18 [13] months. Peak numbers are present in August and September [4]. Population density may fluctuate annually from <1 to 10 animals/ha [4].

Food habits

Almost exclusively insectivorous. Very little known [2,13]. Insects and other invertebrates particularly sawflies [3,4].

Other

No evidence of positive or negative interactions with *Microtus pennsylvanicus* [4] however *S. arcticus* will utilize relatively drier microhabitats when sympatric with *S. cinereus* [17].

Other references

Clough 1963; Soper 1964

2.1.2 Pacific Water Shrew (*Sorex bendirii*)

Synonyms:

Atophyrax bendirii, *Neosorex bendirii* [1]

Other common names

Marsh Shrew, Bendire Shrew [1]

Distribution

Coastal forest of the Pacific Northwest, restricted to the Fraser River delta in B.C. [1,2,4,13]. Biogeoclimatic zone: Coastal Western Hemlock. Ecoregion: Lower Mainland.

Status

Red-listed because of restricted range in highly populated area.

Habitat requirements

Dense, moist coniferous forests, on beaches, and in marshes [13], in heavily wooded, wet areas, on the banks of sluggish streams, in beach debris [2,4], and during winter rainy season may be found well away from water [4]. Near estuaries, wetlands, lakes, streams, and in agricultural areas and riparian forests [15]. In Oregon, restricted to Skunk Cabbage Marsh and Riparian Alder/Small Stream habitats throughout most of the year [8].

Daily activity and movement patterns

Not described. Active 24 hours [8].

Seasonal activities and movement patterns

Not described.

Reproductive parameters

Little known. Breeding begins in January or February [4].

Food habits

Insectivorous. Foods include soft-bodied arthropods and terrestrial and aquatic invertebrates [4]; insect larvae, slugs and snails, Ephemeroptera naiads, earthworms and unidentified invertebrates, primarily aquatic [8].

Other

Enters water readily [2,4,13], and swims and dives well [2,4].

2.1.3 Common Shrew (*Sorex cinereus*)

Synonyms

Sorex personatus [1]

Other common names

Cinereus Shrew, Masked Shrew [1]

Distribution

Predominately northern half of North America; in B.C., entire mainland and some small, adjacent coastal islands [1,2,3,4,13]. Biogeoclimatic zones: all. Ecoregions: all except Queen Charlotte Lowland, Queen Charlotte Ranges, Western Vancouver Island, Eastern Vancouver Island, and Strait of Georgia.

Status

Not in jeopardy, not protected [5].

Habitat requirements

Wide variety of habitats - coniferous and deciduous forests [2,4], alpine meadows [13], stable talus slopes [5] or subalpine rock slides [12], alder or willow brush [3,4], grassland [4,13], and along margins of marshes, bogs and streams [4,12]. Moist habitats may be preferred [3,4,12,13] but also use dry [4,12] habitats. Nest of grass or dry leaves in stumps or under logs or piles of brush [3]. Forages in runways along the ground, among the leaves and debris of the forest floor and may also forage in trees [13]. Requires escape cover such as dense herbaceous layer, decayed logs, or forest litter [5,12]. Abundance of invertebrate prey will determine shrew abundance in a habitat [12].

Daily activity and movement patterns

Active 24 hours a day [3,13]. Peak activity reported just after midnight [4] and at dawn and dusk [13]. Prefers darkness of cloudy or rainy nights [4]. Home range 0.1 acre [13] and 0.4 ha [5]. Will defend nest and surrounding territory [13].

Seasonal activities and movement patterns

Active all year, will travel in tunnels under the snow during Winter [13].

Reproductive parameters

Breeding season varies with latitude [4], but may last from March to October [3], or from late April to October [4,13]. In B.C. breeding begins in mid-May [3]. Litter size varies with latitude [4]; reports of 1 or occasionally 2 litters per season [3,13] and up to 3 litters per season [2]. May breed in winter [3]. Litter size varies from 2 to 10 [2,3,4], averaging 4.4 [13], 6 [2], or 7 [4]. Some females may be sexually mature at 4-5 months old [3,13]. Gestation of 17-18 days [2,13]. Young are weaned at about 20 days [4]. Life expectancy 14-16 months [2,3,13]. Population eruptions not uncommon [3], and populations fluctuate considerably from year to year [4].

Food habits

Primarily insectivorous but eats all kinds of other invertebrates including earthworms, isopods, snails, millipedes, centipedes, molluscs, sowbugs, and spiders [2,3,4,13]. Salamanders, young mice and bird nestlings also reported [13]. Vegetable matter may also be consumed [4,13]. May need to consume >3 times its own body weight per day [4,13].

Other

Probably unable to dig its own tunnels in anything but loose soils [4,13], but uses tunnels of larger species [13]. Senses of hearing, sight and smell are well developed, may use echolocation [4]. Predators are snakes, hawks, owls, mammalian carnivores, and larger shrews [4,13]. Can be taken in snap traps or pitfalls, but the latter is better [12].

Other references

Ingles 1965

2.1.4 Pygmy Shrew (*Sorex hoyi*)

Synonyms:

Microsorex hoyi [1]

Other common names

None [1]

Distribution

Throughout Alaska, Canada and the northern U.S., except in tundra, prairies and west coast. Occupies northern, central and eastern parts of B.C. [1,2,3,4,13]. Biogeoclimatic zones: all. Ecoregions: all but those in Coast and Mountain and Georgia Depression Ecoprovinces, and except Chilcotin Ranges and Okanagan Range.

Status

Few records from B.C. [1,2]. Relatively rare /scarce; < one tenth abundance of *S. cinereus* in most habitats [4].

Habitat requirements

Preference for mesic forest habitats [4]: grassy areas within the boreal forest [13]; wet or dry wooded or open areas [3]; near water in willow thickets, open spruce forest and aspen parkland [2]; shrubby borders of wet meadows, bogs, swamps, marshes [4,13]; occasionally in drier meadows, dry clearings, sand dunes and savannah parkland [4,13].

Daily activity and movement patterns

Active 24 hours [3,4,13] with peak activity at night [4].

Seasonal activities and movement patterns

Not described.

Reproductive parameters

Litter size unknown but about 6 embryos on average [4]. Pregnant and lactating females reported from July and August. Number of litters unknown [4,13], but only 1 per year suspected [13]. Life expectancy not described. Population stability not described.

Food habits

Insectivorous - Lepidoptera, Coleoptera, and Diptera, various adult and larval insects [4,13]. Will also eat flesh of other small mammals [3,13].

Other

Able to climb and jump up to 4.5 inches, and swim passably well [13]. Snap traps may not capture this species even where abundant; but they are susceptible to pitfall traps [12].

2.1.5 Dusky Shrew (*Sorex monticolus*)

Synonyms:

Sorex dobsoni, *S. longicauda* (and 2 subspecies), *S. monticolus mixtus, S. obscurus* (and 7 subspecies), *S. setosus, S. vagrans obscurus, S. vagrans similis*. Nomenclature and taxonomy of this species is controversial, particularly its relationship to *S. vagrans* [1,4]

Other common names

Montane Shrew [1]

Distribution

Throughout western North America. In B.C. it occurs throughout the entire mainland and many coastal islands, including the Queen Charlotte Islands and Vancouver Island [1]. Biogeoclimatic zones: all. Ecoregions: all.

Status

Not in jeopardy, not protected. Abundance in B.C. unknown [5].

Habitat requirements

A variety of different habitats under a wide range of climatic conditions [4]. Montane, boreal or coastal coniferous forests, alpine or subalpine wet meadows and alpine tundra [5]. Marshes, heather, dry hillsides, rain-forest thickets [3,13]; streamside *Equisetum* stands, sphagnum bogs [13]. Wetlands and moist habitats in the arid portion of south-central B.C. [5]. Does better in areas with acidic soils than *S. vagrans* which does better on richer soils [4,11]. Unlike *S. vagrans*, it is rare or absent in open grassy areas and is usually associated with the forest floor litter [4]. Nests often constructed in decaying logs, stumps, forest litter, or a hole in the ground [3,5]. Favours habitats with dense ground cover [9]; duff, litter and herbaceous and woody vegetation serve as escape cover [4,5]. Habitat use varies with temporal changes in herbaceous growth and death [9].

Daily activity and movement patterns

Active 24 hours [3] with a 3-4 hour activity cycle [5]. Solitary [5]. Home range in coastal B.C. varied between breeding status, sex and season from 0.003 to 0.07 ha (mean 0.04 ha) [11]. Non-breeders maintain territories to ensure survival through winter, but breeding adults are not territorial [11].

Seasonal activities and movement patterns

Active all year [5].

Reproductive parameters

Little described for *S. monticolus*, though can extrapolate from *S. obscurus* and *S. vagrans* of earlier literature. Reproductive activity begins in mid-winter [11] (mating is reported to occur as early as March [11,13] or April [4]) and continues through August [4]. Average litter size is about 5, ranging up to 7 [4,3], although *S. vagrans* in earlier descriptions are described as having from 2 to 9 young, averaging 6.4 [13]. Young are weaned within 3 weeks, and females may produce up to 3 litters per season [11]. Life expectancy is 14 to 16 months [11].

Food habits

Insectivorous. Predominantly insects and their larvae, spiders, snails, earthworms and other invertebrates [4,5,8] but may take small salamanders [2]. The more acid soils of *S. monticolus'* habitats provide smaller invertebrates, and their teeth are more robust and slower wearing to cope with relatively higher chitin content of their prey [11].

Other

Because of recent changes in nomenclature, many details of habitat use, activities and feeding must be extrapolated from early descriptions of *S. vagrans* [2,13] and *S. obscurus* [3]. Males may shift or abandon their original home ranges when sexually mature, whereas females held the same range through their entire lives [11].

Other references

Dalquest 1948; Ingles 1960; Ingles 1965; Hawes 1975

2.1.6 Water Shrew (*Sorex palustris*)

Synonyms

Neosorex navigator [1]

Other common names

American Water Shrew, Navigator Shrew [1], Northern Water Shrew [3]

Distribution

From southern Alaska, through Canada, to the Coastal, Rocky and Appalachian mountains of the U.S. Throughout B.C. except Queen Charlotte Islands [1,2,3,4,13]. Coniferous forest region [4,13].

Status

Subspecies *S. palustris brooksi*, an insular race restricted to Vancouver Island [1], is red-listed [14]. Otherwise, not in jeopardy, not protected [5].

Habitat requirements

Near water - lakes, ponds, swift and sluggish small streams, cold fast mountain streams, stagnant water of bogs and marshes [4]. Streambanks, lakeshores [2,4,13]. Adequate cover is required - overhanging banks, logs, boulders, tree roots [3,4,5,13] or dense vegetation [5]. Usually dense climax coniferous forest streamside habitats [13]. May adapt to drier habitats or habitats with ephemeral water [4]. Nests of sticks and leaves [3,13]; globular [4].

Daily activity and movement patterns

Active throughout the day, but peaks for one hour before dawn and 3-4 hours after sunset [4,13]. No evidence of territoriality [4,13]. Little known of home range size [4], but expected to be small and associated with stream or wet area [5].

Seasonal activities and movement patterns

Active all year [4,13].

Reproductive parameters

All males and most females become sexually active in midwinter following their birth [4,13]; some females may breed during their first summer [3,4]. Ovulation is possibly induced by copulation [4]. Mean litter size is 6 young (range 4-8), and 2 or 3 litters are produced per season [2,3,4,13]. Gestation and duration of lactation are unknown [4,13]. Thought to die after their second summer, before 18 months [4,13].

Food habits

Insectivorous. Small aquatic organisms [3], insects, invertebrates, small fish [2,4,13], fish eggs [13] and larval amphibians [4]. Especially aquatic nymphs of caddisflies, stoneflies and mayflies [4,13]. Find food by smell and using tactile vibrissae on snout [13]. Excess food may be hoarded [4,13].

Other

Adapted for swimming, very buoyant, readily takes to water [3,4,13]; may be seen running across water surface [4,13]. May swim under ice in winter [13]. No social hierarchy or territoriality [4,13]. Echolocation suggested but not confirmed [4]. Solitary and aggressive [4]. Able to dig short tunnels in loose soil [4].

Other references

Conaway 1952; Sorenson 1962; Ingles 1965; Kritzman 1977

2.1.7 Trowbridge's Shrew (*Sorex trowbridgii*)

Synonyms

None [1]

Other common names

None [1]

Distribution

The Pacific coast, from California to the lower Fraser River valley, in extreme south-western B.C. [1]. Biogeoclimatic zones: Coastal Western Hemlock (CWH) and Coastal Douglas-fir (CDF)? [15] Ecoregions: Fraser Lowland (FRL) and Northwestern Cascade Ranges (NWC) [15].

Status

Blue-listed [14].

Habitat requirements

Dense coniferous forests and adjacent wooded areas generally at low elevations but extending up almost to timberline [2,13]. Adaptable, occurring in all forest habitats [8]. Riparian, wet and mesic forests, well-drained coniferous forest with low vegetation and ground litter [4,15]. Found in moist litter of forest floor [13], among cover of logs, stumps and decaying vegetation [13]. Does not use brushy clear-cut habitats [15]; populations much reduced in logged sites [13]. Rare in wet meadows [4]. Burrows in the organic surface layer of the soil [4].

Daily activity and movement patterns

Active throughout the day [4]. Breeding adults more than twice as active as non-breeders and immatures [4].

Seasonal activities and movement patterns

Active all year [4,13].

Reproductive parameters

Most litters produced in April through early June [3,4,13]. Two litters per season are possible [2,4,13]. Five young per litter, range 3-6 [2,3,4,13]. Lifespan 12-18 months [3,4,13].

Food habits

Insectivorous. Consumes more food types than other shrew species, reflecting its adaptability to different forest microhabitats [8]. Centipedes primarily [8], and also insects, spiders, isopods, small invertebrates, worms [2,3,4,13], ants, termites, millipedes [10], beetles, flies, insect larvae, slugs and snails [8]. Will eat vegetable matter such as Douglas-fir seeds, other herbaceous plant seeds and some mushrooms, particularly in winter [3,4,13] and in captivity [10]. Hoarding has been observed [10].

Other

Usually excluded from moister sites inhabited by *S. vagrans* with which it competes if it is present [4]. Will share habitat with *S. monticolus* because the latter is more active in the litter of the forest floor rather than the organic surface layer of the soil [4].

Other references

Whitaker and Maser 1976; Terry 1978

2.1.8 Tundra Shrew (*Sorex tundrensis*)

No descriptions of the biology of this species could be found. It is listed only in Nagorsen (1990). It is not listed in van Zyll de Jong (1983), Burt and Grossenheider (1964) or Cowan and Guiget (1975). Banfield's distribution map for the old subspecies designation (*S. arcticus tundrensis*) includes northern Yukon only.

Synonyms

Sorex arcticus tundrensis [1]

Other common names

None [1]

Distribution

Alaska, Yukon Territory and north-western Northwest Territories, and in the Haines Triangle in extreme north-western B.C. [1]. Ecoregion: Tahltan Highland (TAH) [15] and Tatshenshini Basin (TAB).

Status

Red-listed [14]. Known from 5 specimens [1].

Habitat requirements

Well drained hillsides in alpine scrubland [15].

2.1.9 Vagrant Shrew (*Sorex vagrans*)

Synonyms:

Sorex vancouverensis, S. vagrans vancouverensis [1]

Other common names

Wandering Shrew [1]

Distribution

Western North America. In Canada, restricted to southern B.C., including the southern half of Vancouver Island and some of the islands in the Strait of Georgia [1,4]. Ecoregions: Western and Eastern Vancouver Island, Lower Mainland, Strait of Georgia, Pacific and Cascade Ranges, Fraser Plateau, Columbia Mountains and Highlands, and all regions of the Southern Interior Ecoprovince.

Status

Not in jeopardy, not protected. [5]

Habitat requirements

Much the same as *S. monticolis*. Strong orientation to grass and meadow habitats [8]. Wooded and open grassy habitats on well drained to moist sites [4], deciduous riparian forests, wetland habitats [5], habitats with rich soils of low acidity [4,11]. Nests of dry grass in decayed logs, stumps, burrows, under woody debris [5]. Forages in runways under logs, leaf litter, and fallen grass; usually close to water, near marshes, streams, riparian inclusions, thickets, roadsides [5]. Seeks cover in runways, dense herbaceous and/or shrub layer, and forest litter [5].

Daily activity and movement patterns

Active all day, but predominantly at night [4]. Solitary [5]. Home range varies from 24 to 834 m^2 [5] and increases with onset of reproduction [11]: for example, non-breeding females, 1039 m^2, breeding females, 3258 m^2 (there is no difference between sexes) [4]. Territories are maintained by the non-breeding population to preserve resources for over-wintering; breeding adults do not defend territories [11] but defend the nest [5].

Seasonal activities and movement patterns

Active all year with an increase in diurnal activity in spring [4].

Reproductive parameters

Breeding from March to September, but most in March to May [4]. Often 2, sometimes 3 litters per season [11]. Litter size ranges from 2 to 9, averaging 5.2 [4]. Gestation approximately 20 days [4] and weaning occurs at about 21 days [11]. Where food is abundant in lowland grassy habitats, *S. vagrans* produce more litters per year from an earlier age [4]. Generation length is about 12 months, but some *S. vagrans* may live to 16 [11] or 24-25 months [4].

Food habits

Insectivorous. Preys on insects and insect larvae, earthworms, and other small invertebrates [4], including slugs and snails [8]. Small salamanders may be taken [4]. The subterranean fungus, Endogone, is also a major food [8]. Captives will eat various herb, shrub and tree species, and carrion and mushrooms [10]. May consume 1.68 times their weight in food per day, and may store food for future use [5].

Other

Confirmed use of echolocation to orient itself [4]. Will successfully coexist with *S. cinereus* in unequal numbers. In its preferred habitats, *S. vagrans* always outnumbers *S. cinereus* [4]. Uses the runways of other species, especially *Microtus spp*. [5]. The sex ratio is nearly equal over the year, but varies seasonally - more females in summer, more males in autumn [4].

Other references

Ingles 1960; Ingles 1961; Ingles 1965; Hooven *et al* 1975; Maser *et al*. 1981

2.2 Rodentia / Heteromyidae: Heteromyids

2.2.1 Great Basin Pocket Mouse (*Perognathus parvus*)

Synonyms

Abromys lordi, Perognathus laingi, P. lordi

Other common names

None

Distribution

Northern periphery of its range, dry southern interior Thompson River Valley (Ashcroft to Kamloops), Okanagan Valley (Vernon, OK landing, Anarchist Mountain).

Status

Blue-listed [14].

Habitat requirements

Inhabits arid, sandy short-grass steppes, and brushland covered with sagebrush, bitterbrush or rabbit brush.

Daily activity and movement patterns

Crepuscular and nocturnal spending most of the daylight hours in underground tunnels.

Seasonal activities and movement patterns

Hibernate during the winter.

Reproductive parameters

Breed upon emergence in spring (March or April in BC). May be as many as two litters per year with 2-8 young per litter [13].

Food habits

Omnivorous in diet. Specific food items include vegetation, seeds, and insects.

Other references

Ingles 1965

2.3 Rodentia / Muridae / Arvicolinae: Voles and lemmings

2.3.1 Southern Red-backed Vole (*Clethrionomys gapperi)*

Synonyms

Clethrionomys occidentalis, C. occidentalis caurinus, C. occidentalis occidentalis, Evotomys caurinus, Evotomys galei, Evotomys gapperi, Evotomys gapperi athabascae, Evotomys gapperi saturatus, Evotomys occidentalis, Evotomys phaeus

Other common names

Gapper’s Red-backed Vole, Boreal Red-backed Vole

Distribution

Extremely large range across the boreal regions of Canada and the United States. In B.C. it occupies the northeastern, central and southern mainland and some coastal islands [1].

Status

C. gapperi occidentalis is red-listed [14]; it is known from only one specimen from Point Grey [1]. *C. gapperi galei*, found in extreme south-eastern B.C. (Akamina Pass) [1], is blue-listed [14].

Habitat requirements

Primarily a forest species. Found in both coniferous and mixed-wood forests. Associated with the litter of decaying tree trunks and moss covered forest floor. [2, 13, 17].

Daily activity and movement patterns

Although this species can be active at all hours of the day, it is most active from dusk to dawn. Homerange sizes as large as 1.44 ha during summer, can be 1/10 this size in winter [13].

Seasonal activity patterns

Active year round. Somewhat solitary during summer months, except for the female and her litter. In winter they will congregate in family groups [13].

Reproductive parameters

1-8 young/litter [13] born May to October [13]; probably 3 or 4 litters/year [13].

Food Habits

Omnivorous in diet. Seeds, mushroom, grasses, berries [2], forbs, shrubs, leaves [13].

Other references

>Butsch 1954; *Maser and Storm 1970; *Mihok 1979; Martell 1981; >Merritt 1981, 1982; Bondrup-Nielsen 1987; Hayes and Cross 1987; Nordyke and Buskirk 1991.

2.3.2 Northern Red-backed Vole (*Clethrionomys rutilus*)

Synonyms

C. dawsoni, C. dawsoni dawsoni, Evotomys dawsoni

Other common names

Tundra Red-backed Vole

Distribution

Northern B.C., extreme north-west (Haines Triangle) to Summit Pass on the Alaska Highway [1], north-west and north-central B.C.

Status

Not in jeopardy, not protected.

Habitat requirements

Biology not known to differ from *C. gapperi* [2]. Tundra and damp forest floors, alpine conditions [3].

Daily activity and movement patterns

Mainly nocturnal or crepuscular however, will remain active during prolonged arctic daylight hours [13].

Seasonal activities and movement patterns

Active year round. In winter will frequently invade human dwellings [13].

Reproductive parameters

4-9 young/litter [3] born May to September [3]; probably 2 litters/year [3].

Food habits

Primarily herbivorous. Will eat leaves, buds, twigs and fruit of a variety of shrubs and forbs [13]

Other references

West 1982 (see lit cited in Bondrup-Nielsen 1987)

2.3.3 Brown Lemming (*Lemmus sibericus*)

Synonyms

Arvicola helvolus, Lemmus helvolus, L. trimucronatus helvolus [1].

Other common names

Siberian Lemming [1]

Distribution

Northern and central regions [1]; widely distributed over northern half of B.C. but not in north-east lowlands [2]; all of northern and central B.C., including north-east B.C. [3].

Status

Not in jeopardy, not protected.

Habitat requirements

Alpine and subalpine habitats [1]; alpine meadows, occasionally into subalpine timber where grass and weed grown meadows occur [2]; tundra [3].

Daily activity and movement patterns

Makes well beaten runways [2]. Active day and night [3]. Summer nests underground [2,3], winter nests at surface [2,3].

Seasonal activities and movement patterns

Fall dispersal very evident where numbers high [2].

Reproductive parameters

2-6 young/litter [2,3], at least 2 litters/summer [2,3]. Breeding in June to August [3]

Food habits

Entirely herbivorous [2].

Other

Periodic fluctuation in abundance [2,3], with peaks every 3-4 years [3].

2.3.4 Long-tailed Vole (*Microtus longicaudus*)

Synonyms

Arvicola longicaudus, A. mordax, Microtus cautus, M. macurus, M. mordax, M. vellerosus [1].

Other common names

None [1]

Distribution

Entire B.C. mainland and some coastal islands [1]. Along coast, some coastal islands, Okanagan, Rocky Mountains, sea level to 4,000 feet [5].

Status

Not in jeopardy, not protected.

Habitat requirements

Extremely variable [2,5]; grassy forest openings [5] and forest edges from sea level to 4,000 feet [2]; sometimes sedge and grass meadows [2]. Streambanks and mountain meadows, marshes and marshy areas along water courses; banks of cold swift rivers and streams [16,5], in logs, shrubs, talus slopes near water [16], occasional dry situations [16], shrubby areas in winter [3]. Prefers lowland meadows, subalpine meadows and wetlands for reproduction [5].

Daily activity and movement patterns

Seldom makes runways, trails or tunnels [2] except in some meadows [2,5], but will make tunnels in snow [5]. Seldom in large colonies except during peak abundance [2]. Nests above ground in winter, burrows in summer [3,16,5]. Generally diurnal [5], in some areas, may be more nocturnal than diurnal [16]. Home range is 0.1-0.4 (mean is 0.2) hectares [5].

Seasonal activities and movements

Active year round.

Reproductive parameters

From 2 or 3 to 7 (mean is 4 or 5) young/litter [2,16]. Breeding April to September [2](or May to November [16]) shorter in interior [2]. Nests underground, under logs or in rotten logs [5].

Food habits

As for other *Microtus* sp. [2]. Grasses, bulbs, twigs, bark [3], forbs, shrubs, leaves [5]. May eat hypogeous fungi [4].

Other

Density up to 30-40 per hectare in recently logged areas [5]. Cyclic population with 3-4 year cycle [5].

Other references

>Ingles 1965; *****Maser and Storm 1970; >Maser *et al.* 1981; Spencer 1984 (see lit cited in Nagorsen 1987).

2.3.5 Montane Vole (*Microtus montanus*)

Synonyms

Microtus cunicaudus, M. nanus canescens

Other common names

Mountain Vole

Distribution

In B.C. at northern periphery of its range [1], restricted to dry interior of southern B.C., Okanagan valley to Williams Lake [1], (Okanagan Valley to Kamloops, eastward to the Kettle Valley [2]; Okanagan, Kettle, and Thompson Valleys, north to the Cariboo [5]).

Status

Not in jeopardy, not protected.

Habitat requirements

Arid grasslands of sagebrush community and higher grassland community [2]; sagebrush steppe and southern bunchgrass steppe [5], usually at higher elevations than other voles [4], forage in dense cover of grasses, reeds or sedges near water [4]. Varied habitats in US range: sagebrush-juniper woodlands to sagebrush flats to cultivated fields, into subalpine and alpine meadows and near water, ditches, streams and marshes [16].

Daily activity and movement patterns

Little known about its biology [2,3]. Rest in shallow burrows or in nests under log or rock cover, requiring herbaceous layer as escape cover [5]. Dependent on extensive runway systems in grasslands [16], summer above ground nests, winters below ground [16]. Home range in the range of 0.1 hectare.

Seasonal activities and movement patterns

Active all day, all year [5].

Reproductive parameters

1-9 (mean is 1-4) young/litter [5]; probably breeds throughout the year.

Food habits

Leaves and stems of forbs and grasses [5], rushes [5], and mullein (*Verbascum thapsus*) under snow.

Other

3-4 year population cycle [5]. Often associated with Harvest Mice, Vagrant Shrews, and Muskrats [5]. Good swimmers [16]. Runways often lined with faeces, food, refuse and clipped vegetation.

Other references

Goertz 1964; >Ingles 1965; ***Maser and Storm 1970.

2.3.6 Tundra Vole (*Microtus oeconomus*)

Synonyms

Microtus operarius, M. yakutatensis

Other common names

Root Vole

Distribution

Extreme north-western B.C. [1] at Stonehouse Creek [2].

Status

Not in jeopardy, not protected.

Habitat requirements

Sub-tundra vegetation of cotton rush sedge, grasses, heaths and flowering plants [2], moist to wet tundra and other wet locations [3].

Daily activity and movement patterns

Makes runways through tundra vegetation [3]. May store some food [3]. Nests in shallow burrows or under debris [3].

Seasonal activity and movement patterns

Active year round.

Reproductive parameters

3-11 embryos reported [3].

Food habits

In summer diet consists of clipped sedges and grasses. Will store grass seeds and forb roots in their burrows during autumn.

2.3.7 Creeping Vole (*Microtus oregoni*)

Synonyms

Microtus serpens

Other common names

Oregon Vole

Distribution

Coastal lowlands and coastal mountain ranges of south-western B.C. [1], Fraser River delta, Vancouver to Hope, north to Agassis, east to Allison Pass, south to border; not north of Burrard Inlet [2].

Status

Not in jeopardy, not protected.

Habitat requirements

Deciduous forest of Puget Sound Lowlands Biotic Area [2], Burrows in loose soil among woody debris [2].

Daily activity and movement patterns

Surface runways in dense grass [2].

Seasonal activities and movements

Active year round, however, very few live more than 1 year [13]. Fall populations consist primarily of young born in that year.

Reproductive parameters

1-5 (mean is 3) young/litter [2], up to 5 litters/season [2]. Breeding March to September.

Food habits

Vegetation - stems, roots, bulbs, tubers, plant crops, fallen fruit [2].

Other references

Goertz 1964; ***Maser and Storm 1970; Doyle 1987.

2.3.8 Meadow Vole (*Microtus pennsylvanicus*)

Synonyms

Arvicola drummondii, A. modesti, Microtus pennsylcanicus funebris, M. pennsylvanicus rubidus, M. stonei [1].

Other common names

None

Distribution

East of coastal mountain ranges [1].

Status

Abundant, not in jeopardy, not protected [5].

Habitat requirements

Variety - with ground cover of sedges, grass, weeds; usually near water [2]; cultivated fields, wild meadows, wet alpine meadows, forest glades, sedge meadows, tundra pond edges [2]; low moist areas or high grasslands with rank growths of vegetation; near streams, swamps, occasionally in forests with little ground cover; orchards with grass undergrowth [3]; wetlands and lowland wet vegetation, streambanks, swampy areas, ponds, sedge meadows, lake shores with adequate cover, roadside ditches with grass and weeds, grassy forest openings [5]; most common in marshy areas near streams, lakes and potholes [16].

Daily activity and movement patterns

Complicated network of runways on surface [3], underground [2,5] and under snow [5]. Active day and night [3,5], may be more nocturnal in summer, and diurnal in winter and cloudy days [16 citing Jackson 1961]. Nests above or below ground [3]; burrows along surface runways [3], nest in ball of grass in abandoned burrows, grassy tussocks, rocks, or under logs or boards. Home range 1/10 - 1 acre [3]

Seasonal activities and movement patterns

Active all year [5].

Reproductive parameters

1-9 [3] or even 11 [2] (commonly 3-7 [3,16]) young/litter, 4 or more litters/year, mature at 30 or 40 days, breeds throughout the year [3,5], 21 day gestation [3], main season March to November [5].

Food habits

Various, vegetation [2], grasses sedges, seeds, grain, bark, some insects [3], winter - seeds, roots, bulbs, bark [4], stores seeds and tubers for winter use.

Other

Excellent swimmer [2,3,16], frequently enters water [2,16] population fluctuates every three to four years [3]. Lives for 3 years [3]. Good fighter, probably territorial [3].

Other references

>Getz 1961; *Maser and Storm 1970; >Kritzman 1977; Getz 1982; Klatt and Getz 1987; Hall *et al.* 1991; Whittaker *et al.* 1991.

2.3.9 Water Vole (*Microtus richardsoni*)

Synonyms

Arvicola richardsonii, Aulacomys arcivoloides, Microtus principalis [1]

Other common names

Richardson's Vole [1]

Distribution

Alpine and subalpine areas of 2 disjunct ranges: Coast Mountain ranges of southwestern B.C. (Cascades and S. Coast Mountains as far north as Lillooet) and the interior mountains in south central B.C. (Selkirks to Mt. Revelstoke [2], Purcell, Monashee and Rocky Mountains to latitude of Mt. Robson [2], excluding Columbia River valley below Golden [2]).

Status

Not in jeopardy, not protected.

Habitat requirements

Alpine, subalpine [1]; mountain stream banks near timberline in mixed stands of low willows and dense herbage [2], creek banks and marshes; mountains above timberline [3].

Daily activity and movement patterns

Shallow burrows [2]; extensive tunneling when abundant [2]. Surface runways broad and distinct, almost always lead to water [2] some entrance to burrows below water (!!!)[3]. Nests beneath roots, old stumps, logs; winter nests on ground below snow [2].

Seasonal activities and movements

Active year round.

Reproductive parameters

2-8 (mean is 5) young/litter [2]; Born June to late September [2]. Breeding season not known [3].

Food habits

Herbivorous [2]. Little grass or sedge, prefers herbs [2].

Other

Swims and dives well [2,3]. Colonial in behaviour [13].

Other references

***Maser and Storm 1970; Doyle 1987; Ludwig 1988.

2.3.10 Townsend's Vole (*Microtus townsendii*)

Synonyms

Arvicola occidentalis, A. tetramerus, A. townsendii [1].

Other common names

None [1]

Distribution

Lower mainland (Puget Sound lowlands biotic area) [2], west of Cascades, north to Burrard Inlet, east to Chilliwack [1], Vancouver Island and some adjacent islands (Triangle, Scott, Bowen, Texada, Saltspring and Pender Islands) [1]

Status

Subspecies *M. townsendii cowani*, an insular race restricted to Triangle Island [1], is red-listed [14].

Habitat requirements

Varied; from sea level to 6,000 feet [16], from beach debris [16] and moist fields and sedge meadows of alluvial areas of Fraser River delta up to timberline, to alpine/subalpine on Vancouver Island [2], dense lush vegetation on seabird colony islands [2], sedges, tules, meadows, from tidewater to alpine meadows [3], usually near water [3] (occasionally far from permanent water [16]), marshes and marshy areas, wet meadows with grass or fern cover [16], logged areas with suitable habitat [16].

Daily activity and movement patterns

Active day and night [16]. Surface runways and extensive underground tunnels in loose soil [2], runways lead to water, scattered with food and faeces, latrines at intersections up to 6” x 4” pile [16].

Seasonal activities and movement patterns

Nests are underground in summer [3], above ground in winter [16]. May be crowded during winter flooding, but disperse with drying conditions.

Reproductive parameters

1-9 (mean is 4) young/litter [2] (or 2-10 (averaging 5-8) young/litter [16]. Breeding season varies from March to September [2, 3] or from May to November or even January in some areas [16].

Food habits

Stem bases and roots of grass and sedge, other succulent herb plant stems (2). Heath at higher altitudes on Vancouver Island [3], cut or stored [16]. There is evidence of Townsend's Voles eating bark and needles of conifers [16].

Other

Good swimmer, dives readily [16], vicious fighters [16].

Other references

Goertz 1964; ***Maser and Storm 1970; Taitt *et al.* 1981; Taitt and Krebs 1981; Beacham 1982; Hilborn 1982

2.3.11 Heather Vole (*Phenacomys intermedius*)

Synonyms

Microtus pumilis, Phenacomys constablei, P. olympicus, P. oramontis, P. ungava mackenzii [1].

Other common names

None [1], Mountain Phenacomys, Heather Phenacomys [3]

Distribution

Disjunct distribution [2], lower mainland (Mount Seymour, Mount Whistler, Garibaldi Park), south Rocky Mountains extreme south-eastern area, mid-coastal to interior (Kimsquit to Cariboo) [5], north of Peace barely into northwestern B.C. [16]. All B.C. except Vancouver Island, North coast and north of the Kimsquit [3].

Status

Not in jeopardy, not protected.

Habitat requirements

Varied [2,3], typically under stumps, trunks, bearberry in dry places [2] near surface water, grassy or brushy covered areas, grassy areas near mountain tops [3], Douglas fir/lodgepole pine forests and Engelmann spruce and subalpine fir forests [5], pine and spruce forests, coniferous, open, seral or mature forest in dry sites, rocky slopes, tundra, dry areas and near water [3]. Winter nests are made of twigs and lichen and are made above ground under shrubs or rocks [3,5]. In summer, nests are below ground under rocks, stump debris [3,16,5]. A wide range of dry habitats: lowland, subalpine and alpine meadows [5], but a preference for areas with abundant coarse woody depris (CWD) near water and forage [5].

Daily activity and movement patterns

No runways except around nest, but will use those of other species [2], or make short ones from nest only [16], most active at twilight and night [3,16] and during day [5].

Seasonal activities and movement patterns

In winter, nests are above ground under shrubs or rocks [3,5]; in summer, nests are below ground under rocks and stump debris.

Reproductive parameters

1-8 (usually 4 or 5) young/litter [2], (2-8 young/litter [3], 2-8 young/litter (usually 4-6) [16]). Born June to September, gestation 21 days [3], 2 or more litters/year. Sexually mature at 4-6 weeks, breed first year [3].

Food habits

Various vegetarian [2], *Arctostaphylos uva-ursi* = 85-90% of diet - leaves and berries [2], bark of dwarf birch and willow, seeds, lichens, berries, green vegetation [3], graminoids, forbs, shrub foliage [5]. In winter they eat bark, twigs and lichen [5]. Heather voles sometimes hoard food [2,3,5].

Other

Solitary in summer; family groups in winter.

Other references

>Shaw 1924; >Foster 1961; *****Maser and Storm 1970; Johnson 1973 (see lit cited in Nagorsen 1987); >Kritzman 1977; >Innes and Millar 1981; Corn and Bury 1988.

2.3.12 Northern Bog Lemming (*Synaptomys borealis*)

Synonyms

Arvicola borealis, Synaptomys andersoni, S. ballatus, S. borelais wrangeli, S. chapmani, S. dalli, S. truei, S. wrangeli.

Other common names

None

Distribution

Found in most areas of B.C. [2].

Status

S. borealis artemisiae, restricted in B.C. to the Similkameen and Okanagan valleys [1], is red-listed [14]. *S. borealis borealis*, found south of the Peace River in north-eastern B.C. [1,2], is not listed [14].

Habitat requirements

Although this species primarily inhabits bogs, it may also be present in other wet habitats such as: deep mossy spruce woods, wet subalpine meadows and alpine tundra. The *S.b. artimisiae* subspecies occupies much drier habitat and has only been found in the dry sagebrush slopes of the southern Okanagan valley.

Daily activity and movement patterns

Active at all hours of the day.

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Breeding season extends form May to August [13], 1-3 litters per year [17], 2-8 young/litter [2,13,17]

Food habits

Herbivorous. Subsisting on grasses and sedges and other plant types. Will pile clippings in runways.

Other

Very distinct latrines along runways. Some may have several cups of droppings in them [2].

Other references

*****Maser and Storm 1970

2.4 Rodentia / Muridae / Murinae: Old world rats and mice

2.4.1 House Mouse (*Mus musculus*)

Synonyms

Mus domesticus

Other common names

None

Distribution

Introduced species found throughout B.C. in areas of human habitation.

Status

Not in jeopardy, not protected.

Habitat requirements

Originally adapted to dry arid steppes within its natural range. Closely association with human habitation and cultivated fields. Is not found in forested habitats.

Daily activity and movement patterns

Primarily nocturnal.

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Breeds throughout the year under ideal conditions. Several litters per year are possible. Litter sizes can range from 1-12 at the extreme. Sexual maturity is reached at 35 days.

Food habits

Omnivorous. Food consists of grains, fruit and vegetables, stored food and refuse.

Other

Colonial species

Other references

Sheppe 1967; >Maser *et al.* 1981

2.4.2 Norway Rat (*Rattus norvegicus*)

Synonyms

Mus norvegicus

Other common names

None

Distribution

Introduced species associated with urban and agricultural areas in the southern mainland in BC.

Status

Not in jeopardy, not protected.

Habitat requirements

Strongly associated with human habitation. Will live in buildings, landfill sites and open fields.

Daily activity and movement patterns

Active at all hours of the day. Very small home range sizes have been recorded (as little as 25 m in longest length of travel at times).

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Extremely prolific. Average number of litters per year is 5 [13] with as many as 22 young per litter.

Food habits

Omnivorous.

Other references

Chitty and Shorten 1946

2.4.3 Black Rat (*Rattus rattus*)

Synonyms

Mus rattus

Other common names

Roof Rat

Distribution

Introduced species associated with urban and agricultural areas in the southwest coast, Vancouver Island and many islands in the Queen Charlotte Islands.

Status

Not in jeopardy, not protected.

Habitat requirements

Strongly associated with human habitation. Will live in buildings, landfill sites and open fields. Have been known to inhabit islands containing sea bird colonies. Also lives along forest edges.

Daily activity and movement patterns

Active at all hours of the day. Very small home range sizes have been recorded (as little as 25 m in longest length of travel at times).

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Extremely prolific. Similar reproductive potential to *R. norvegicus*.

Food habits

Omnivorous.

Other references

Chitty and Shorten 1946

2.5 Rodentia / Muridae/ Sigmodontinae: New world rats and mice

2.5.1 Deer Mouse (*Peromyscus maniculatus*)

Synonyms

Hesperomys austerus, Permomyscus maniculatus angustus, P.m. balaclavae, P.m. georgiensis, P.m. saturatus, Peromyscus texanus, Sitomys americanus, Sitomys americanus artimisiae, Sitomys keenii, Sitomys macrorhinus.

Other common names

White-footed mouse

Distribution

Found throughout BC

Status

Not in jeopardy, not protected.

Habitat requirements

No specific habitat requirements other than sufficient food, water and cover. Broad tolerance for a variety of habitats. Found from marine shorelines to old growth coniferous forests.

Daily activity and movement patterns

Primarily nocturnal, however can be active during day. Home range sizes are generally in the neighborhood of 1 ha, however greater distances can be covered by individuals [13, Vanessa Craig, pers.com.].

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Breeding season ranges from April to October. 2-4 or occasionally more litters can be produced per year. Litter sizes typically range from 2-8 young.

Food habits

Omnivorous. Primarily a seed eater, however diet can include carrion, other plant materials, mushrooms, insects and other invertebrates. Will store seeds in caches.

Other references

Hooven 1958; >Sadleir 1974; Petticrew and Sadleir 1974 (see lit cited in Bondrup-Nielsen 1987); *Mihok 1979; Martell and Macaulay 1981; >Maser *et al.* 1981; Van Horne 1981 (see lit cited in Bondrup-Nielsen 1987); Hayes and Cross 1987.

2.5.2 Columbian Mouse (*Peromyscus oreas*)

Synonyms

Peromyscus maniculatus beresfordi, P.m. carli, P.m. doylei, P.m. interdictus, P.m. oreas, P.m. sartinensis, P.m. saxamans, Peromyscus sitkensis doylei, P.s. isolatus, P.s. saxamans

Other common names

Cascade Deer Mouse

Distribution

Occupies the Coastal Mountains from Washington to Rivers Inlet on the British Columbia Mainland [1]

Status

Not in jeopardy, not protected.

Habitat requirements

Similar to *P. maniculatus*. More common at higher elevations [13].

Daily activity and movement patterns

Similar to *P. maniculatus*.

Seasonal activities and movement patterns

Similar to *P. maniculatus*.

Reproductive parameters

Breeding period more restricted than for *P. maniculatus* occurring from April to the end of July. Litters may be a little larger than for *P. maniculatus*.

Food habits

Similar to *P. maniculatus*.

Other references

Sheppe 1961; Merkens 1994

2.5.3 Sitka Mouse (*Peromyscus sitkensis*)

Synonyms

P. maniculatus sitkensis, P. prevostensis [1], *P. maniculatus precostensis* [2], *P. sitkensis prevostensis* [2]

Other common names

None [1]

Distribution

Isolated outer islands of the Queen Charlotte Islands

Status

Not in jeopardy, not protected.

Habitat requirements

Similar to *P. maniculatus*.

Daily activity and movement patterns

Similar to *P. maniculatus*.

Seasonal activities and movement patterns

Similar to *P. maniculatus*.

Reproductive parameters

Similar to *P. maniculatus*.

Food habits

Similar to *P. maniculatus*.

2.5.4 Western Harvest Mouse (*Reithrodontomys megalotis*)

Synonyms

Reithrodon megalotis

Other common names

None

Distribution

Restricted to southern B.C. and southeastern Alberta in Canada. At its northern periphery of its geographic range here. More specifically, within B.C. it is confined to the floor of the Okanagan Valley [2].

Status

Blue-listed [14].

Habitat requirements

Primarily a grassland species that can be found within the dense vegetation along the margins of lakes, streams and salt marshes, in rose thickets along the margins of cultivated areas, as well as wooded ravines.

Daily activity and movement patterns

Nocturnal with peak activity occurring between 7 and 9:30 p.m. [13]. May reduce activity during bright, moonlit nights.

Seasonal activities and movement patterns

Active year round.

Reproductive parameters

Breeds in late spring and summer. Several litters are born each year with litter sizes ranging from 1-9.

Food habits

Subsists largely on seeds and vegetation. During fall and winter, seeds are predominate in their diets.

Other references

Pearson 1960

2.6 Rodentia / Dipodidae: Jumping mice and jerboas

2.6.1 Meadow Jumping Mouse (*Zapus hudsonius)*

Synonyms

Dipus hudsonius, D. labradorius, Gerbillus canadensis, G. labradorius, Jaculus labradorius, Meriones labradorius, Zapus tenellus.

Other common names

None

Distribution

Found throughout the northern half of B.C. east of the coast range and the central plateau of B.C. ranging from the Skeena Valley in the north to the Okanagan Valley to the South.

Status

Z. hudsonius alascensis, of the Haines Triangle area in extreme north-western B.C. [1], is blue-listed [14].

Habitat requirements

Prefer moist grassland [13]. Can be found in association with brushy margins of streams, clearings and other openings [2].

Daily activity and movement patterns

Mostly nocturnal but will appear on damp, dark afternoons.

Seasonal activities and movement patterns

Enters hibernation in late September to early October. Males may emerge from hibernation early in May with females emerging in late May. They can be quite nomadic with movements in the neighborhood of 1 km [13].

Reproductive parameters

Breeding season commences upon emergence from hibernation and continues to the end of August. 2-3 litters can be produced with an average of about 5 young/litter [13].

Food habits

Primarily granivores. During summer they will consume fruits. They also consume both adult insects and larvae.

Other references

Quimby 1951; Whitaker 1972; Muchlinski 1988

2.6.2 Western Jumping Mouse (*Zapus princeps*)

Synonyms

Zapus saltator [1]

Other common names

None

Distribution

Throughout mainland B.C. except extreme southwestern [1] in the Fraser River lowlands [2]: disjunct through south-central B.C., and excluding the northern Omineca-Peace region (Ft. Nelson, north).

Status

Common in appropriate habitat [5], not in jeopardy, not protected [5]

Habitat requirements

Brushy stream margins, clearings, openings [2], lush grass and herb growth [3]moist lowland meadows, alpine and subalpine meadows, fresh emergent wetland [5], grassy meadows, thickets, forest openings, brushy margins of streams and lakes [5], requires dense security cover - herb, shrub, logs [5].

Daily activity and movement patterns

Chiefly nocturnal [3], nests on surface under grass or herbs [3]. Grass nests in shallow depression among grass clumps, rocks or logs [5]. Home range is about 1/4 hectare for both sexes, range 100-400 m along grassy banks and wet areas [5].

Seasonal activities and movement patterns

Hibernates from mid-September until new growth of ground vegetation is well underway [2]: Hibernation from September/October to April/May [3]. Winter nests 0.3-1 m underground [5].

Reproductive parameters

3-5 or 2-7 young/litter [2], and 1 litter/year [2] born in June and July [3]

Food habits

Herbivorous diet including seeds [2] or primarily composed of seeds [3] also containing other plants and berries [5].

Other

Good swimmer [3], can jump 4-6 feet [3].

Other references

Brown 1967; Brown 1970; Kritzman 1977

2.6.3 Pacific Jumping Mouse (*Zapus trinotatus*)

Synonyms

Z. imperator, *Z. princeps trinotatus* [1], *Z. trinotatus trinotatus*

Other common names

Northwestern Jumping Mouse [2]

Distribution

Extreme southwestern B.C., as far north as Garibaldi Provincial Park, east to Allison Pass in Manning Provincial Park [1], Puget Sound Lowlands of Fraser River delta east to Allison Pass [2].

Status

Not in jeopardy, not protected.

Habitat requirements

Moist meadow land and edges of riparian thickets [2], wet, marshy areas, open meadows, woods to timberline [3].

Daily activity and movement patterns

Little described, probably as in *Z. hudsonius* and *Z. princeps*; nocturnal.

Seasonal activities and movement patterns

As for *Z. princeps*; probably hibernates September through May.

Reproductive parameters

As for *Z. princeps*

Food habits

As for *Z. princeps*, seed eating

Other

Swims strongly and takes to water readily [2].

3. Protocols

The secretive nature and physical size of the species outlined in this manual make them difficult to inventory by direct counts or quantification of sign left by them. For the most part, determining the presence or abundance (relative or absolute) of any of these species requires physical capture of individuals to provide data that can be interpreted correctly. For this reason, trapping is the most efficient means of inventorying individual small mammal species as well as entire small mammal communities.

Several methods of animal capture are available and the choice of the appropriate survey method as well as sampling design will be dependent on the objectives of the inventory or study, the species that are being targeted and the length of time available to complete the surveys. Small mammal trapping can be accomplished by three means: live-trapping, pitfall-trapping and, in some restricted cases, snap-trapping. Live-capture trapping (accomplished by both live- and pitfall-trapping) is the most appropriate method for any studies of, or in areas where there may be, threatened or endangered species and is necessary for any estimation of absolute abundance. Live-capture trapping provides the most reliable and most informative data about population numbers and demography, and is the best method to determine differences between habitat types and for monitoring population changes following disturbance (Ritchie and Sullivan 1989) or over time. Snap-trapping (or kill trapping) itself will have an impact on how the resident population changes with time (both over the short term and long term) and may bias data. Snap-trapping may be useful for gathering information about diet (stomach contents) or reproduction (placental scars), but is very limited as an inventory method.

3.1 General considerations for inventory

3.1.1 Inventory objectives

The most important step in the inventory process is to define the objectives clearly and precisely to determine the staff, level of effort, budget required, sampling design and appropriate analysis methods. While a multitude of survey objectives are possible, for the purposes of inventory they may be generalized as objectives to estimate population distribution (presence/not detected), relative abundance and absolute abundance. Additional objectives may be met by the methods described here. Some of these are discussed in the section describing each survey intensity level.

3.1.2 Selecting areas to survey

Once survey objectives have been established, the next step in inventory is to define the area to be surveyed. The bounds of a project area for abundance and composition surveys are often arbitrary. They can be as large as management units, regions or ecoprovinces, or as small as some specified portion of a stand of trees. The extent of the overall project area is frequently tied directly to the objectives of the survey.

Once the overall project area is defined, then study areas within the bounds of the project area can be determined. To accomplish this, the overall project area should be stratified by habitat type and survey work should be concentrated in those areas most likely to provide the data necessary to meet the objectives. Delimiting ideal study areas requires knowledge of the target species natural history as well as physical limiting factors such as access.

Information within Section 2 will assist in determining study areas however, additional sources of information should be consulted prior to initiation of field work. Consult the Conservation Data Centre for any potential species records in the study area or nearby. Review the results of any prior research or inventory within the area. Talk to the people who conducted the studies. Local naturalists and biologists may also be able to provide useful information. Provincial Ministry of Forestry and Ministry of Environment, Lands and Parks offices may have local vegetation or habitat maps available, and information on populations.

Ultimately, study areas should reflect objectives, animal habitat associations, and available budget.

3.1.3 Personnel

An experienced professional biologist should undertake the design, logistic planning, and data analysis for inventories. When necessary, specialists or persons with additional experience, should be consulted to save time, money, and frustration. Prior to initiating major inventory surveys, a short workshop with inventory experts present, may be useful to update inexperienced personnel on survey procedures and requirements.

A qualified biologist and a field crew with experience and competency in identification of species in the hand are mandatory. Juveniles of some species are particularly difficult to identify. A library of identification manuals and field books can be useful when in the field: Burt and Grossenheider (1964), Maser and Storm (1970), Banfield (1974), Cowan and Guiget (1975), van Zyll de Jong (1983) and Sullivan (1997). Up-to-date taxonomic classification for B.C. species is presented in the RIC manual No. 2 entitled, *TheVertebrates of British Columbia: Scientific and English Name*s. The mammal taxonomy section of this manual is largely based on Nagorsen (1990). An identification key based on external features (Nagorsen in prep.) would be handy in the field. Ideally, a customized summary description from all of these sources would be helpful for species expected in the study area.

All personnel should thoroughly review manual No.3, *Live Animal Capture and Handling Guidelines for Wild Mammals, Bird, Amphibians, and Reptiles*, and manual No.4 *Voucher Specimen Preparation* before commencing with a RIC wildlife inventory survey that requires capture and/or handling.

3.1.4 Equipment

A significant amount of specialized equipment is required to conduct surveys of small mammals. The most important kind of field equipment you will use is trapping and animal handling equipment. Ensure that all field personnel are familiar and comfortable with using all the equipment before heading into the field. Below are detailed descriptions of the equipment necessary to conduct field work.

1. Traps

Live traps

The two most common live traps for the smaller species are the Sherman trap (H.B. Sherman Traps, Tallahassee, Florida) and the Longworth trap (Longworth Scientific Instrument Co., Ltd.). The Sherman is available in 2 sizes (13 x 13 x 38 cm and approx. 7.5 x 9 x 22 cm) and comes in collapsible and fixed-sided models. A third trap, the Bolton trap (B.N. Bolton, Vernon, B.C.), is identical to the Longworth but is a Canadian product and can be ordered in stainless steel or aluminum models.

Longworth and Bolton traps are particularly recommended for voles and lemmings. Shermans, Longworths or Boltons are acceptable for mice, jumping mice and rats. The best choice depends on the habitat, the objectives of the study, funds available for purchasing traps, and access constraints to the sampling site. Because the Sherman comes in a collapsible model, it is easier to carry long distances. Longworth and Bolton traps have the advantage of a mechanism to lock the trap door open.

To leave open Shermans in the field for pre-baiting they must be locked open by applying a 1” spring clamp to the door mechanism or by bending the treadle-trigger. In the latter case the treadle trigger must be bent back to reset the trap. The sensitivity of the trigger is undoubtedly altered by bending it back and forth and continued fiddling will lead to metal fatigue; therefore the spring clamp is recommended.

Pitfall traps

Pitfall traps are not available commercially; but are constructed from readily available materials. Pitfall traps come in a variety of shapes and sizes, but the general design is a hole in the ground into which an unwary animal falls. The trap is usually a can or bucket dug into the ground so the rim is flush with the surface. The depth and diameter of the pit are selected to be large enough so that the species of concern cannot crawl or jump out. Woody debris or a shingle is supported above the hole to protect the victims from exposure and predation, and to attract animals to the safe haven under the debris. Drainage holes and floatable debris such as squares of Styrofoam are provided for the captured animals.

Pitfall equipment includes: tin cans, plastic buckets, duct tape, metal hole punch, canvas, roof sheathing, or garden lawn border. Depending on the soil substrate, shovels, grub-hoes, and/or heavy crowbars are required to prepare holes. Variations in trapping success with different pitfall configurations and dimensions have been described in the literature for several small mammal communities (Bury and Corn 1987, Szaro *et al.* 1988, Friend *et al.* 1989).

Use of drift fences increases the number of individuals and species captured (Bury and Corn 1987, Friend *et al.* 1989); the length of fencing rather than the orientation of the fencing is believed to be the determining factor (Williams and Braun 1983, Bury and Corn 1987, Friend *et al.* 1989). Drift fences can be made of 40 cm high plastic sheeting or tarp material and act as a guide to direct animals into the pitfall traps.

Snap traps

Snap traps are available in 3 sizes:

- the Museum Special Mouse Trap (Woodstream Corp., Lititz, PA) and the Victor Mouse Trap (Four Ways Animal Trap Co., Lititz, PA) which are suitable for mice, voles and smaller chipmunks and squirrels;
- the Victor Rat Trap (Four Ways Animal Trap Co., Lititz, PA) and the McGill Rat Trap (McGill Metal Products Co., Marengo, Il; Carly and Knowlton 1971 cited in Johnson and Keller 1983) which are suitable for *Rattus* and small mammals in the 100-200g range, but which also capture mice and voles;
- the generic, household mouse trap available at hardware stores, suitable for mice, voles and shrews.

Comparison of snap trap designs and newer models are available (Johnson and Keller 1983, West 1985, Mengak and Guynn 1987). Not surprisingly, larger and stronger traps take larger animals more efficiently, but generally, the Museum Special is recommended as the best snap trap option (Johnson and Keller 1983, Weiner and Smith 1972 cited in ibid, Mengak and Guynn 1987).

2. Bait for traps

Whole oats or peanut butter mixed with rolled oats are recommended as baits for rodents. For rodent trapping carrot slices provide moisture to prevent dehydration and stress. Green vegetation may provide sustenance to captured voles. Shrews, being primarily insectivorous, may not be as attracted to peanut butter or oats as other species nor will this type of bait provide the appropriate sustenance for shrews. Walnuts, bits of earthworm or insect larvae (meal worms) may prove more attractive and provide shrews with better nourishment. If live-trapping is being conducted for shrews, food should be supplied within the trap, be it pitfall or live-trap.

3. Bedding and Floats for traps

Coarse brown cotton (available at Western Fibres, Vancouver) provides insulation for captured animals. *Peromyscus* make a good bed of cotton but *Microtus* are more likely to use the bedding if it is first pulled apart and fluffed up.

Squares of Styrofoam provide floatation for the trapped animals should the pits fill with water.

4. Weigh scale

Portable spring scales are ideal for weighing captured animals in the field. A variety of scales (50, 100, 300 and 500-g models) are available from Pesola (distributed by Le Naturaliste, Quebec) or Avinet (distributed by Avinet Inc., Dryden, N.Y.). Choice of scale is dependent on species to be weighed. Always use the smallest possible scale that can accommodate the weight of the captured animal to ensure best accuracy. Remember to pad the clamp of the scale with adhesive tape before attaching it to an animal to prevent it from inflicting injury.

5. Equipment for marking animals - Ear tags and pliers, marking pens or hair dye

Ear tags

Size 1 (National Band and Tag Co., Newport, KY) or Clasping Fish Fingerling size (Salt Lake Stamp Co., Salt Lake City, UT). National Band and Tag tags appear to work best. The Clasping Fish Fingerling tags may no longer be available; however some inventory of these still exists within government offices in BC.

Pliers, necessary to attach tags, are available from the tag manufactures and can be relatively expensive. These can break down or be left behind in the field, therefor a back-up pair is suggested. Cheaper home-made pliers, if even slightly off-design can also work well.

Black permanent ink pen

Permanent ink pens can be used for marking captured animals on their ventral surface for future recognition in the capture session. This need only last the length of the capture (trapping) session and is not intended to endure between sessions. Test pens prior to field use to ensure that marks will not be lost.

Hair dye

Hair dye can be used to mark animals. Miss Clairol has worked (V. Craig per. comm.). Use various colours (black, red, blonde etc.) and apply to different parts of the body and in different combinations to give the individual a unique identifier.

6. Scissors

Scissors should be of a size that can be easily wielded and kept sharp; include a backup pair. These are important if the animal becomes tangled in the trap cotton or for any other complications. For instance, one researcher described how he once accidentally attached a mouse to the glove used to handle the animal while applying an ear tag (M. Merkens, pers. comm.). The glove had to be trimmed to release the mouse.

7. Animal handling container

These are used to contain captured animals after they are emptied from the trap. Heavy weight plastic ore bags (available from Industrial Plastics, various locations in BC) or tall plastic buckets can be used for this. The bag must be large enough to easily go over the end of the trap and roomy enough for the handler to put a gloved hand in to grasp the animal. Five gallon plastic buckets can be obtained from food service industries and are excellent for containing animals. These buckets should be of sufficient height to prevent animals from jumping out over the rim.

8. Plastic bags and Waterproof specimen labels

For accidental live trap and pitfall trap mortalities as well as voucher specimens.

9. Gloves

Soft cotton work gloves are sufficient to handle most small mammals captured. You may still be bitten through the gloves, but bites usually do not draw blood. If you are concerned about personal injuries, leather gloves can be used; however, dexterity and sensitivity is reduced and you may end up stressing the animal more than necessary due to increased handling times. Given the size of rats, leather gloves are recommended when working with these species. Leather gloves may also be useful for incidental captures of species not covered by this manual, particularly if the researcher wishes to record and measure the captive. Soft gloves are adequate for chipmunks, but if you want to handle tree squirrels or small mustelids, animal handling cones are recommended. Light weight canvas and net bags are adequate for these and can be easily sewn into a shape that will restrain captured individuals.

10. Flagging tape

Flagging tape is used to mark capture (trap) stations over the duration of the study. Make sure that your choice of colour does not conflict with other activities in the area. For instance, you do not want to use the same colour that the local forest company uses to lay out roads or cutblock boundaries.

11. A small vial of 10% sugar in water

This is used to revitalize stressed, hypothermic or heat-stressed captures. A clean eye drop bottle works well to administer the sugar solution. Always carry the bottle and remember to change the solution often.

12. A waterproof trap record notebook

Data can be recorded in small notebooks provided that they are set up properly. Standard RIC data sheets can also be copied onto Write in the Rain Paper (available from Neville Crosby, Vancouver) and carried in the field on clip boards.

13. A short, simple identification key

Keys containing expected species can be easily made using field guides. Similar keys have been published (Sullivan 1997). More complicated keys that exist in literature can be scaled down by deleting those species that are not going to be encountered in the area. Time spent using keys will likely diminish over prolonged periods of trapping as individual surveyors become familiar with local species.

14. Magnifying headband

A binocular magnifying headband is useful in examining anaesthetized shrews for morphological features crucial in the determination of species.

15. Light weight boards

For protective covers over pitfalls and live traps left in the field for extensive periods. Recommend a minimum 30 cm x 30 cm plywood cuttings for use with live-traps. A regular 4ft x 8ft sheet of 1/4 inch plywood will provide 32 boards.

16. A large pack

To transport traps and trapping gear to the trap site.

3.1.5 Sample units

Cluster sampling can be defined by a process in which a population is surveyed using quadrats, index lines, trapping grids, or any other standardized survey method. The sample unit in the case of small mammals is an index line (transect line) or trapping grid as opposed to the individual animal in the population. Likewise, although individual traps are used to capture individual animals, sampling units are composed of a set number of traps set in a particular configuration defined by the objectives of the survey. Trap spacing within these sample units should be derived by considering the homerange of the target species especially when estimating absolute abundance. It has been suggested that four capture stations (trap stations) per home range be sufficient to allow equal access relative to the homerange size. Given this and assuming a square homerange with 4 capture stations per home range, then station spacing should be less than or equal to the square root of the area of the home range divided by 2. As a general guideline a spacing of approximately 15 m between capture stations will provide for adequate coverage.

3.1.6 Survey timing

Small mammal populations fluctuate annually with low populations occurring late in winter and high populations occurring after the breeding season. Along with this annual fluctuation are general population movements. As populations increase, subordinate individuals may be displaced from optimal habitat in species that have strong social structures. This displacement results in individuals potentially occupying habitat that can not be considered optimal or preferred for the species. The actual timing of surveys should be carefully considered to determine how it will affect the overall study objectives.

For presence/not detected surveys involved in establishing range of a species, sampling in late summer or early fall after the breeding season when populations are generally at their yearly maximum will likely result in the greatest probability of capturing any species. If the presence/not detected survey is being conducted to document habitat associations, then sampling should probably occur earlier in the year prior to juvenile dispersal when individuals are more likely to occur in their preferred habitats.

When surveying for relative abundance, capture sessions (trapping sessions) should occur at least twice during the active season (May to October for most species). Sessions should occur during the spring breeding period and during the fall, after breeding is completed. The data from spring sampling and fall sampling should be considered independently in analyses. Sampling should be conducted at the same time each year in multi-year monitoring studies.

When sampling for absolute abundance, more intensive trapping is required. Depending on the objectives of the study and choice of analysis models, trapping times and intensities will vary. See the absolute abundance survey section for more detail.

It is important to note that certain small mammal populations follow multi-year cycles, and this may influence survey results. Some species (particularly vole species) experience population irruptions at 3-4 year intervals. It would appear that these irruptions are, for the most part, synchronous over very large areas. During these irruptions, massive dispersions of animals may occur, flooding the landscape.

3.1.7 Survey forms and data recording

Standardized forms are necessary to ensure that data are not omitted. These data forms do not have to be taken into the field if small data books are set up to avoid data omission.

Ultimately, when data are entered into provincial data systems, it will be important that all fields have been collected using appropriate codes.

3.1.8 Safety

Some small mammal species are carriers of pathogens that can affect human health. The most serious of these is the Hanta virus which has received much notoriety in recent years. With precautions and appropriate field gear (gloves, respirator, coveralls) the exposure to these pathogens can be minimized. Consult the Worker's Compensation Board for the latest regulations regarding this issue.

3.1.9 Permits

Any project involving small mammal trapping will require a permit. These permits can be obtained from the Regional Office of the Ministry of Environment, Lands and Parks responsible for the project area.

3.2 Sampling Standards

3.2.1 Standards for accuracy and precision and survey bias

Accuracy refers to how close the parameter estimate is to the true population parameter, and it can be improved by designing a robust study which accounts for biases such as variable trappability. Precision is the closeness of repeated measurements to the mean population estimate. Precision is quantified by the sampling variance, and can be improved by replicating surveys, increasing the number of sample units, stratifying samples into groups where variation is expected to be lower, and by optimal allocation of sampling effort.

Accuracy and precision are both important for good survey estimates. Without those measures it is difficult, if not impossible, to compare studies over time or between areas. Bias can be classified into two types: 1) sampling bias; and 2) model bias. Model bias is the most serious of the two, since increasing sample size usually does not reduce the magnitude of the bias. All sample-based estimates are based on statistical models which depend on one or more assumptions. If all of the assumptions of the statistical model are not met, model bias results. More about the choice of models and associated bias is covered later, particularly in relation to the absolute abundance survey method.

Precision is commonly indicated by associating confidence intervals with the estimate. A confidence interval gives the known probability $(1 - \alpha)$ that the actual value of a parameter will be included within the interval. It is recommended that $\alpha = 0.10$ so that confidence intervals will provide a 90% probability that the actual value of a parameter will be included within an interval.

In addition to establishing *Type 1* errors (α levels), *Type 2* errors (β levels) should also be specified when using statistical tests. Gerrodette (1987) provides a good discussion of *Type 1* and *2* errors as they relate to population estimation. Briefly, the test of a null hypothesis results in either accepting or rejecting the hypothesis, based on some estimated risk of being wrong. The probability of rejecting the hypothesis when it is true is referred to as a *Type 1* error. The largest recommended acceptable risk of committing a *Type 1* error is $\alpha = 0.10$. A *Type 2* error (β) is the probability of concluding that the null hypothesis is true when in fact it is false. The largest recommended acceptable risk of committing a *Type 2* error is $\beta = 0.20$.

This will provide for statistical power (probability of rejecting the null hypothesis when null hypothesis is false) of 0.80.

3.2.2 Capture Probabilities

There are many confounding problems when sampling small mammal populations. Factors that influence survey bias and biases associated with model selection will all influence the accuracy of surveys and include:

- the area covered by the index trapping line (transect) or grid;
- the number of traps per capture station and in total;
- capture probabilities, of species or sex and age classes;
- Temporal and behavioural variation in capture probabilities;
- the number of trap-nights and capture sessions;
- the total population size; and
- how well survey design accommodates estimation model assumptions.

A carefully designed study can minimize the effects of confounding factors including variation in capture probabilities.

Simply stated, capture probability is the probability of an animal being caught in any trap. There can be great variation in capture probabilities between individuals within a population as well as over time. White *et al.* (1982) discuss practical and theoretical issues regarding variation in capture probability in great detail and identify three possible sources of variation in capture probabilities (heterogeneity, behaviour and time variation). To summarize from White *et al.* (1982), capture probabilities can:

1. vary over time due to weather effects or the amount of effort used to capture animals on any occasion (time variation);
2. vary amongst individuals due to innate factors such as age, sex, social status or the number of traps in its home range (heterogeneity);
3. vary depending on whether the individual had been previously captured (behaviour). If the capture probability of an individual decreases after the first capture than it is said to have become “trap shy”. On the other hand, if it increases it has become “trap happy”.

These sources of variation can be additive, resulting in very complex estimation models. They must be considered and tested for in any attempt to analyze data, especially absolute abundance data. Once again, careful design and implementation of studies can minimize capture probability variation in some part.

When laying out a trapping grid for absolute abundance estimates, the number of captures is further complicated by the fact that territory size and shape does not coincide with the shape of the area covered by the traps. A number of fractional territories overlap the trapping area and can introduce a bias factor. Problems associated with this can be minimized through proper study design. For instance, use of large grid areas and short sampling duration can minimize biases associated with edge (of the sampling grid) effects.

3.2.3 Standards for sex and age classification

The sex of small mammals can usually be determined through examination of genitalia and mammaries. Consult field guides to determine criteria for sex determination and become familiar with the relative positions and sizes of the genital papilae. Sex is particularly difficult to determine when examining young animals. Due to their size, shrews are also difficult to sex, however during breeding season, pregnant or lactating females are easily identified.

The age of live trapped individuals can generally be assessed by size, weight, pelage condition and colour. Sexual maturity and the duration of the breeding season can be determined according to the palpation of testes (scrotal or abdominal) for males and the conditions of the vaginal opening (perforate or not) and mammaries (large or small), and whether or not obviously pregnant for females.

3.2.4 Habitat Data Standards

A minimum amount of habitat data must be collected for each survey type. The type and amount of data collected will depend on the scale of the survey, the nature of the focal species, and the objectives of the inventory. As most, provincially-funded wildlife inventory projects deal with terrestrial-based wildlife, the terrestrial Ecosystem Field Form developed jointly by MOF and MELP (1995) should be used. However, under certain circumstances, this may be inappropriate and other RIC-approved standards for ecosystem description may be used. For a generic but useful description of approaches to habitat data collection in association with wildlife inventory, consult the introductory manual, No. 1, Species Inventory Fundamentals. When establishing habitat associations for individual species, habitat may have to be assessed at a much finer scale depending on the typical homerange size and movement patterns the species in question.

Because water shrews are riparian specialists, living at the interface of land and water, habitat variables from both the aquatic and terrestrial environment should be routinely described. Features of the aquatic environment that should be recorded include: channel width, wetted width, substrate particle size, flow rates, depth, gradient and channel cover. Availability of invertebrate prey and the presence and relative abundance of fish may also be recorded. Terrestrial features that should be described include riparian vegetation and coarse woody debris levels.

3.3 Inventory Surveys

Table 2 outlines the type of surveys that are used for inventorying small mammals for the various survey intensities. These survey methods have been recommended by professional biologists and approved by the Resources Inventory Committee. Detailed protocols for each survey type are outlined in the following sections.

Table 2. Types of inventory surveys, level of intensity and the data forms needed for a small mammal survey

Survey Type	**Data Forms Needed**	***Intensity**
Trapping (along index trap lines, no animal marking)	• Wildlife Inventory Project Description Form • Wildlife Inventory Survey Description Form - General • Capture Form - Small Mammal *Transect Layout* • Animal Observation Form - Small Mammal Capture • Ecosystem Form	• PN
Trapping (along index trap lines with animal marking)	• Wildlife Inventory Project Description Form • Wildlife Inventory Survey Description Form - General • Capture Form - Small Mammal -*Transect Layout* • Animal Observation Form - Small Mammal Capture • Ecosystem Form	• RA
Mark-recapture (on trapping grids)	• Wildlife Inventory Project Description Form • Wildlife Inventory Survey Description Form - General • Capture Form - Small Mammal *Grid Layout* • Animal Observation Form - Small Mammal Capture • Ecosystem Form	• AA
Any Survey Type	• Marked Animal Identification Form - use whenever an animal is marked. • Wildlife Inventory Survey Collection Label - is use whenever a voucher specimen is collected.	• PN • RA • AA

* PN = presence/not detected (possible); RA = relative abundance; AA = absolute abundance

Overall, the choice of trap type, configuration and necessity of marking captured individuals is dependent on the species being targeted and the level of inventory required to meet objectives of the inventory. Table 3 outlines guidelines for the selection of some of these criteria for various levels of inventory and species groups. Selection of appropriate trap spacing within configurations is discussed elsewhere in this document.

Table 3. Recommended trap type, configuration, and marking requirements for various levels of inventory for shrews, voles, mice and rats.

Species Group	Inventory Level	Preferred Trap Type	Configuration	Marking Required
Shrews	PN	Pitfall	Index Lines[4]	no
	RA	Pitfall	Index Lines	yes
	AA	Pitfall	Grids	yes
Water Shrews	PN	Pitfall	Riparian Transects	no
	RA	Pitfall	Riparian Transects	yes
Voles and Lemmings	PN	Longworth	Index Lines	no
	RA	Longworth	Index Lines	yes
	AA	Longworth	Grids	yes
Mice	PN	Longworth or Sherman	Index Lines	no
	RA	Longworth or Sherman	Index Lines	yes
	AA	Longworth or Sherman	Grids	yes
Rats	PN	Tomahawk	Index Lines	no
	RA	Tomahawk	Index Lines	yes
	AA	Tomahawk	Grids	yes
All Species	PN	Museum Special Snap	Index Lines	no

[4] Also referred to as transects or transect lines.

3.4 Survey Standards

3.4.1 Weather Standards

Weather can influence the behaviour and thus trappability of species. When possible, trapping should be temporarily (within the bounds of your study design) postponed during particularly wet or unseasonably cold weather. It may not be possible to avoid rain and cold weather during sampling and inventories can still be carried out under these less than ideal situations if appropriate study designs and robust models of estimation are used in the survey. Trapping can, for that matter, occur during the winter months if determining winter activity patterns or population trends are objectives. Sullivan (1997) provides details on how traps can be set up for winter use.

3.4.2 Time of day standards and frequency of checking traps

Small mammal traps are first set in the afternoon of the first day of trapping. The frequency of checking the traps will be governed by the species being surveyed and the objectives of the study. If conducting presence/not detected surveys, overnight trapping is sufficient given that trap mortalities will not affect analysis methods or model assumptions. If surveys are assessing abundance, traps will need to be checked frequently enough to minimize violations of the assumptions of some of the models used in surveys. More specifically, trap mortality should be minimized. Overnight trapping is sufficient for rodent species but more frequent checking (every 1.5-2 hrs) is required to ensure minimal mortality in shrews. Traps should be checked at a regular interval for the entire capture session (trapping session). During extremely hot weather, traps should be locked open or left closed during the late morning and early afternoon to minimize mortality due to heat stress. Between capture sessions traps can be locked open and left in place if studies require multiple sampling periods in the same year.

3.4.3 Survey Design Hierarchy

Surveys of small mammals follow a survey design hierarchy which is structured similarly for all RIC standards for species inventory. Figure 1 clarifies certain terminology used within this manual (also found in the glossary), and illustrates the appropriate conceptual framework for a small mammal trap survey. Any survey, including others outlined in this manual, which follows this design will lend itself well to standard methods and RIC data forms.

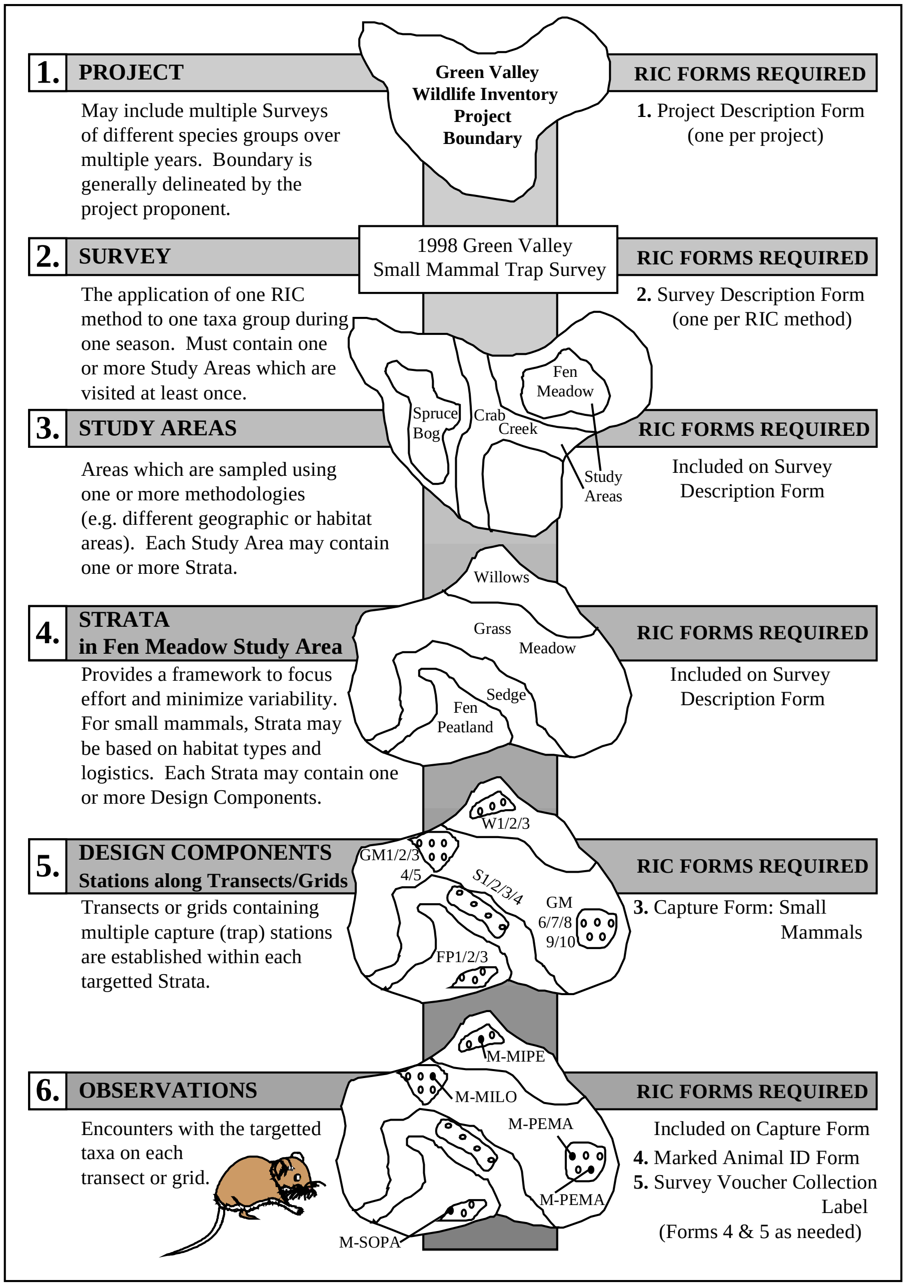

Figure 1. RIC survey design hierarchy for small mammals.

3.5 Presence/not detected (possible)

3.5.1 Recommended Method: Trapping

Single and Multiple species inventory

To determine species of small mammals in a specific area, the recommended procedures are a combination of live trapping and pitfall trapping (using the trap that best captures the target species, Table 3) with systematic sampling along index trap lines (transects) that are randomly placed within stratified sites.

In some cases, where the objective is to determine species presence or absence at a geographic scale (i.e. determining species range), and if it has been determined that no threatened or endangered species are in the project area, then snap traps are an alternative to live and pitfall trapping. This method of inventory is recommended with reservations as it does not capture all small mammal species or age classes equally (V. Craig pers. comm.) and has the potential to disrupt social structure, age structure, behaviour, reproductive parameters and immigration rates of a population. Furthermore, by removing a significant proportion of the biotic community through snap trapping, rare species will no longer be in a natural environment and their behaviour may not reflect their actual habitat preferences or association. Therefore snap trapping should probably not be used for determining habitat associations for rare species. Snap traps may be used for only one sampling session (capture session) per area and for presence/not detected (possible) estimates only.

3.5.2 Office Procedures

- Review the introductory manual No. 1 *Species Inventory Fundamentals.*
- Determine final objectives of the study (see Table 4 for examples)
- Determine species to be studied
- Determine other species expected in the project area
- Obtain maps for project and study area(s) (*e.g.*, 1:50 000 air photo maps, 1:20 000 forest cover maps, 1:20 000 TRIM maps, 1:50 000 NTS topographic maps).
- Outline the project area on a map and determine Biogeoclimatic zones and subzones, Ecoregion, Ecosection, and Broad Ecosystem Units for the project area from maps.
- Determine approximate location of study area(s) within this project area. Study areas should be representative of the project area if conclusions are to be made about the project area. For example, this means if a system of stratification is used in the Sampling Design then strata within the study areas should represent relevant strata in the larger project area.
- Stratify study areas based on habitats. This will allow you to distribute your effort in manner to maximize your ability to detect all possible species in the study area.
- Determine sampling area dimensions, trap spacing, trapping intervals
- Obtain permits
- Order and organize gear

3.5.3 Sampling Design

Every study involved with determining the presence of a species can be conducted by establishing randomly located transects of traps (index lines) within the study areas. The exact dimensions of these lines as well as the type and number of traps along each of these transects will be dependent on the target species, population levels and the overall objectives of the study. Live traps and pitfall traps provide a means of live-capturing individuals in an area whereas snap traps result in the permanent removal of captured individuals.

Live Traps

For those species best sampled with live traps (Table 3), the following index trap line sampling design is recommended to determine their presence: a straight (preferred) or meandering transect line with 20 capture (trap) stations per line at a minimum 15-m spacing with a minimum of 2 traps/station. If a continuous transect of 285 m is not possible, the transect may be split into shorter non-overlapping transects that sum to 20 stations. Care must be taken to ensure that line segments remain independent from one another when setting out fragmented lines. This can be achieved by spacing them relative to the home range size of the target species.

Capture (trapping) sessions can last from 2 to 6 nights depending on trapping success. Trappability can be improved after a period of pre-baiting. To prebait, traps containing bait and left locked open should be placed in the field for a minimum of two weeks prior to actual trapping. This allows resident animals to familiarize themselves with the traps.

Pitfall Traps

For those species best sampled with pitfall traps (Table 3), the following index trap line sampling design is recommended to determine their presence: a straight (preferred) or meandering transect line with 5 capture (trap) stations per line at a minimum 60-m spacing. An array of one central and three radial pitfalls within 3 m should be set up at each capture station. If a continuous transect of sufficient length to accommodate the 5 capture stations is not possible, the transect may be split into shorter non-overlapping transects that sum up to 5 stations. Care must be taken to ensure that trapping arrays remain independent from one another when setting out fragmented lines.

Capture (trapping) sessions can last from 2 to 6 nights depending on trapping success. Trappability can be improved after a period of inactive trap placement. During this period, pitfall trap arrays can be set up in the field with tight fitting lids on the traps or sufficient debris left in the traps to allow animals to climb out of them. Two weeks should be sufficient for this period. This allows resident animals to familiarize themselves with the traps.

Drift fences may be erected between pitfall traps within arrays to direct animals toward the traps and thereby increase the capture rate/unit effort. Corn and Bury (1990) describe pitfall layout and construction for terrestrial amphibian studies; their design is also suitable for small mammal studies (Aubry *et al.* 1991, Carey and Spies 1991, Corn and Bury 1991). Details of the setup of these pitfall arrays can be found within the RIC Inventory Manual for Plethodontid Salamanders (No. 36). Small piles of woody debris can be placed near the pits to attract animals to them. Make the debris pile inviting to small mammals seeking shelter.

Live Traps and Pitfall Traps

For multiple species inventory a combination of the two trapping methods (live and pitfall) is recommended: a 285-m straight (preferred) or meandering transect line with 20 capture stations per line at 15-m spacing with a minimum of two live traps/station and five arrays of four pitfalls regularly spaced along the trapping transect (e.g., at stations 1, 6, 11, 16, 20).

Snap traps

The ground layout for snap trapping is much like that of live trapping. The following index trap line sampling design is recommended: a straight (preferred) or meandering transect line with 20 capture stations per line at a minimum 15-m spacing with a minimum of 2 traps/station, where one trapping survey lasts at least two nights.

3.5.4 Sampling Effort

The general objective of presence/not detected surveys is to determine whether a species is present to document species geographic ranges or species richness in an area. Determining presence of a species is far more easy than establishing absence. Some species may occur at very low densities, be difficult to detect (have low trappability) and/or show a great degree of spatial and temporal variability in distribution. The documentation of the absence of any individual species can only occur after survey efforts are replicated sufficiently both spatially and temporally. Studies of abundant animals that are easily captured require less trapping effort than species that are rare or difficult to capture.

Sample size will affect the accuracy of any determination of presence and should be directed by the overall objectives of the study. If the objective of the study is to detect most of the species in a particular habitat type, the optimal sample size can be determined by plotting the cumulative number of species detected versus the number of surveys (comprised of both sampling units (index lines/transects) and number of sampling nights). Theoretically, this curve will plateau when the most detectable species have been found. This curve will change for different habitat types and different seasons and therefore must be redrafted when survey parameters change. The results of this type of determination are not necessarily statistical, but they give the biologist a rough estimate of the number of surveys needed.

If the objective of the study is to determine the presence of one particular species, especially rare species, then sample size should be determined experimentally. Data from previous studies in similar habitat or pilot studies can be used as a starting point for this. In general the species covered in this manual tend to have clumped populations and clumped populations are more difficult to detect than randomly distributed populations. Clumped populations do not fit normal distributions but rather conform to negative binomial distributions. Methods of determining optimal sample sizes for the detection of species conforming to the negative binomial distribution are covered extensively in Krebs (1989) and fine details of this process will not be covered here. It is important to adequately replicate sampling efforts so that some statistical estimate of a species absence can be made. The replicates should be both spatially and/or temporally independent from each other.

Another issue related to sampling effort is the number of traps placed at each capture sampling station on an index line (transect). If the population densities of local species are sufficiently high, then trap saturation may occur. That is, if all traps are filled with animals before they are checked and there are still animals at large and potentially trappable, then insufficient numbers of traps have been set out. Sufficient traps have been set out to enumerate a population if >20% of the traps remain empty following a night of trapping (Southern 1973, Gurnell 1976).

3.5.5 Assumptions of methods

The question of whether sampling effort is sufficient can only be answered (using the methods described above) if the following assumptions are met:

1. The distribution and density of animals, the environmental factors, and the size of the area surveyed to determine density and dispersion parameters for estimation of sample sizes are similar to those in the area to be surveyed for presence/not detected.
2. The distribution of animals in the study area is best approximated by the negative binomial distribution for determination of optimal sampling effort to detect individuals.
3. Surveys in both presence/not detected and abundance surveys are random samples.

3.5.6 Equipment

- Traps: Live traps; Pitfall traps; Snap traps and associated supplies
- Bait for traps
- Bedding and Floats for traps
- Weigh scale
- Plastic bags or Bucket
- Gloves
- Flagging tape
- Plastic sample bags (e.g., Ziploc sandwich size) and waterproof specimen labels
- A small vial of 10% sugar in water
- A waterproof Trap Record notebook and/or RIC dataforms on clipboard
- A short, simple identification key
- Light weight boards
- A large pack

3.5.7 Field Procedures

Based on an example of single and multiple species inventory: live traps, index traplines (transects)

1. Select a site that is homogeneous in habitat.
2. Establish a fixed tie point (reference point which is a known distance and bearing from the first capture (trap) station on the index trapline). Prepare detailed notes on access and the index trapline for other field crews. These should include UTM coordinates.
3. Establish a straight-line (preferred) or meandering transect through the study site. All points along the transect must be away from habitat edges, and sites of disturbance. Allow for at least 300 m of transect.
4. Use flagging tape to mark capture stations at intervals outlined above along the transect. Use trees or sturdy shrubs for the flags, or strong, well-placed sticks, re-bar or snow flags

in open or grassland habitats. Mark the transect number and capture station number on the flagging tape at each station.

5. For live traps, place traps within 2 m of the capture station. The central pitfall trap in the array should be as close as physically possible to the capture station. Live traps should be placed at microsites which might be attractive to mice or voles. Appropriate sites include positions along or under woody debris or rocks, under bushes, along worn travel trails.
6. Each trap should be baited. Slices of carrots will help avoid dehydration of some animals. Sufficient cotton bedding material should be provided. In Sherman traps it is essential that no grains, seeds, vegetable matter or bedding is allowed to hinder the function of the trap door trigger. A cover of debris, vegetation or lightweight board over the trap will decrease chances of hypothermic or hyperthermic captures. Attach a piece of coloured flagging tape to the trap or place one in branches above it for easy relocation.
7. A pre-baiting period of 2 weeks should precede sampling. Baited traps should be left at the sampling site with the doors locked open to allow the animals to familiarize themselves with the novel object in their environment. Traps should be left open and baited at the site between sampling capture sessions if possible.
8. Set the traps in the evening when the pre-baiting period is complete.
9. Check the traps the following morning as early as possible to minimize mortalities and trap stress. Under hot, cold or wet weather conditions, the trapline should be checked once in the late afternoon as well. Shrews, because of their high metabolic rates, do not survive long in live traps, if mortality is to be minimized more frequent trap checks are necessary. If endangered or threatened species are the focus of the study or are expected to be captured in the area, this afternoon check is mandatory to prevent unnecessary mortality. More frequent checks are recommended if mortality of shrews is of concern.
10. Captured individuals should be identified to species, and age, sex class and reproductive status determined. Standard morphometric measurements can also be taken if necessary. These include: total length, tail length, hind foot length, ear length, and weight. Not all measurements are necessary for all animals; however, weight should be taken for all. Records of each capture should include capture station number along with the biological data.
11. Trap mortalities should be described as above, and placed in individual plastic bags with a detailed data label. Mortalities should be frozen as soon as possible and prepared as voucher specimens. Check with the Royal BC Museum whether these vouchers should be added to their collection.
12. Index traplines should be active at least 2 nights; Ritchie and Sullivan (1989) recommend 4-6 nights to allow residents sufficient time to encounter the trap line. Longer periods may be required for rare species with long latency of detection periods.
13. When the capture session is complete, lock open the live traps and/or deactivate pitfall traps and leave them at the site (‘pre-baited’) if more sessions are to be conducted. Traps can be left, locked open, over winter for the duration of multi-year studies if necessary. If traps must be removed from the site between sessions (for example, because of limited trap numbers available), leave a similar item, such as a tin can with a little bait at each capture station to simulate the ‘pre-bait’trap.
14. On completion of the study, remove all flagging tape, markers, ‘pre-bait’ tin cans, or other debris from the site. Researchers wishing to repeat your study should be able to find your trapline from tie point and site description notes.

Single or multiple species inventory: snap traps, index traplines (transects)

1. Snap-trapping is a one-time event per area and should occur during the fall after breeding is completed, as populations are generally at their peak.
2. Select a site that is homogeneous in habitat.
3. Establish a fixed tie point (reference point which is a known distance and bearing from the first capture (trap) station on the index trapline). Prepare detailed notes on access and the index trapline for other field crews. These should include UTM coordinates.
4. Establish a straight-line (preferred) or meandering transect through the study site. All points along the transect must be away from habitat edges, sites of disturbance, and different habitats. Allow for at least 300 m of transect.
5. Use flagging tape to mark 20 capture stations at 15-m intervals along the transect. Use trees or sturdy shrubs for the flags, or strong, well-placed sticks or re-bar in open or grassland habitats. Mark the transect number and capture station number on the flagging tape at each station.
6. Place 2 traps within 2 m of the capture station (3 traps may be required for sites where animal densities are >50 per ha or where <20% of the traps remain empty after a night of trapping). Traps should be placed at microsites which might be attractive to mice or voles. Sites include along or under woody debris or rocks, under bushes, along worn travel trails.
7. Each trap should be baited with a mixture of peanut butter and oats (whole or rolled are suitable). Attach a piece of coloured flagging tape to the trap or place one in branches above it for easy relocation.
8. A pre-baiting period of 2 weeks should precede sampling. Baited traps should be left at the sampling site with the snap mechanism sprung (i.e. not set) to allow animals to familiarize themselves with the novel object in their environment. After pre-baiting, 2 trap-nights is the minimum recommended time for a capture (trap) session. If pre-baiting cannot be done, a longer capture (trapping) session is necessary. Ritchie and Sullivan (1989) recommend 4-6 nights to allow residents sufficient time to encounter the trap line, in the absence of pre-baiting.
9. Set the traps in the evening when the pre-baiting period is complete.
10. Remove captured animals and reset traps the following morning. To maximize capture success, clear and reset traps in the morning and evening throughout the capture session.
11. Records of each capture should include capture station number, along with the biological data. Standard morphometric measurements include: total length, tail length, hind foot length, ear length, weight; these measurements may be necessary for intensive population studies.
12. A representative sample of trap captures should be collected and preserved as voucher specimens.
13. On completion of the study, remove all flagging tape and markers.

3.5.8 Special case concerning inventory of water shrews

Water shrews have a reputation for being somewhat uncommon, or occurring at low densities. This interpretation is usually based on the results of a generalized small mammal sampling program rather than one which is modified to target this species according to its unique habits and niche. The water shrew is an extreme habitat specialist, spending the majority of its time in or alongside water. Thus, small mammal sampling that is not focused at the land/water interface is biased against detection of the species.

Recent work by L. Hartman (pers. com.) indicated that setting traps even a short distance from standing water significantly reduces capture success. Preliminary results of the Vancouver Island water shrew inventory showed that traps placed within centimeters of the water's edge may be more effective than traps placed 1 m from water's edge.

Special equipment requirements

- Pitfalls $\geq$20 cm in depth are adequate to trap water shrews and are reasonable to install in the rocky ground found alongside many streams. They have the added advantage of permitting the escape of deer mice, obviating the need for intensive Hanta virus precautions. If the intention is to conduct a multi-species inventory, it is easier to add snap or box traps to the line than to increase the depth of the pitfall in stream microhabitats. Twenty pitfall traps per sampling unit is recommended. Strong plastic, stackable containers are convenient and hold up to rigorous use in the field (commercially available leech pots are effective).
- Vapour barrier plastic cut into 1 foot high x 5 foot long sheets for drift fences
- Metal highway flags, with flags cut off (gives metal stakes about 18" long, about the thickness of coat hangers but straight) - used to erect drift fence as it can be difficult to use wood in stream channel because of rocks
- Flow meter or other device for estimating flow
- 1.5m pole demarcated into centimeters for stream depth and cover estimation
- Small ruler to measure substrate particle size
- Sponge, to clean and dry pitfall once set
- If live-trapping, you will need to add bait and bedding material to the pitfall traps. Bait - some studies use mealworms, they are clean and do not attract many other species; captive studies use small "bricks" of frozen meat, a mixture in equal parts of beef liver, beef brain and chopped beef suet. As a rough guide to quantity, shrews can eat their weight in food each day.

Field procedures

Trap checking frequency

Water shrews, like other shrew species, are highly vulnerable to trap death, due to a combination of exposure and starvation effects. It is not uncommon to observe several partially cannibalized shrews in a pitfall containing multiple captures. To minimize or avoid shrew mortality, traps must be checked every 1.5 to 2 hours, a constraint that clearly limits the number of sites that can be surveyed in a given field season. For this reason, many small mammal inventories make no attempt to ensure live-capture of shrews. However, the decision to live- or kill-trap must be based on more than logistic considerations. Instead, it must take into account the conservation status of the species, the potential for local population recovery, the goals of the study, and the funding available.

Two of the three taxa of water shrew occurring in British Columbia are red-listed. *S. bendirii* is the Pacific Water Shrew, confined to the Lower Mainland, and *S. palustris brooksi* is the Vancouver Island subspecies of the Water Shrew. Its mainland counterpart is the yellow-listed *S. p. navigator*, which occurs throughout the province except in the Lower Mainland. The conservation status of the former two taxa warrants special consideration in the development of methods; however it must also be recognized that this red-listed status potentially stems, in part, from an absence of appropriately focused survey effort. Thus, survey methods must be chosen which balance the need for new management information against the potential risks to the species, subspecies or population being studied.

If broad inventory is the objective, and tissues are of interest for genetic analysis, then kill-trapping to determine presence may be considered, BUT trapping at a given site should only continue until initial detection. Any work proposed for red-listed *S. bendirii* should employ live-trapping techniques, because of its presumed rarity, and its restriction to a highly fragmented and reduced range within the urbanized landscape of Greater Vancouver.

Pitfall traps require special modification to result in live-capture. In addition to being checked every 1.5 to 2 hours, traps should have food and bedding material placed inside.

Index line configuration

The current protocol for water shrew surveys is to set an index line (transect) of 20 pitfalls at 15-m intervals, along the edge of the waterway being sampled. Trap effort should be expended within 5-10cm of the water. To set pitfalls this close to water, workers must dig directly into the water table, hold the pitfall in place while backfilling bed material around it, and then weight it down with a rock on the rim. If bed material is quite rocky, a nearby source of finer substrate should be found when possible, to smooth out the area around the pitfall. In good weather, pitfalls can be placed deep enough that only 1 cm of the rim lies above the water level before backfilling. If it is rainy and streams are likely to rise, pitfalls should be set correspondingly higher. Once set, a sponge is useful to clean and dry the interior of the pitfall.

A drift fence of at least 1.3m should be put in place perpendicular to the stream, beginning about 30 cm into the water (unless flows preclude this), running across the mouth of the pitfall and extending up the bank. Vapour barrier plastic works well as it can be cut to accommodate large rocks or logs as needed. Wire stakes are recommended for supporting the drift fence, because they can be worked into the typically rocky substrates found near streams. Upon completion of the first sampling capture session, drift fences can be removed, stakes intact, and rolled up to form a compact bundle for future use. If live-trapping is proposed, a cover larger than the mouth of the pitfall trap should be placed about 5 cm above the pitfall to prevent exposure to rain.

Based on an example of water shrew survey

1. Select a site appropriate to survey objectives.
2. Establish a fixed tie point (reference point which is a known distance and bearing from the first capture (trap) station on the trapline). Prepare detailed notes on access and the trapline for other field crews. These should include UTM coordinates.
3. Establish a transect that follows the edge of the water. Allow for at least 300 m of transect.
4. Use flagging tape to mark capture stations at intervals outlined above along the transect. Use trees or sturdy shrubs for the flags, or strong, well-placed sticks or re-bar or snow flags in open or grassland habitats. Mark the transect number and capture station number on the flagging tape at each station.
5. Place pitfalls as described above.
6. Supply food and bedding to the traps if live-trapping is desired.
7. Periods of pre-baiting have not been tested but might improve trapping efficiency.

8. Set the traps in the evening of the first day of trapping.
9. Check the traps as frequently as necessary to meet objectives and conservation concerns. Daily checks are acceptable if traps are removed immediately following detection of the target species if they are species of concern (i.e. red- or blue-listed).
10. Captured individuals should be identified to species, and age, sex class and reproductive status determined if appropriate to the study. Records of each capture should include capture station number along with the biological data.
11. Trap mortalities should be described as above, and placed in individual plastic bags with a detailed data label. Mortalities should be frozen as soon as possible and prepared as voucher specimens. Check with the Royal BC Museum whether these vouchers should be added to their collection.
12. Index traplines (transects) should be active up to a maximum of 6 days to allow resident individuals sufficient time to encounter the trap line. Longer periods may be required for rare species with long latency of detection periods; however these latency of detection parameters have not yet been determined for water shrews.
13. When the capture session is complete, deactivate pitfall traps and leave them at the site if more sessions are to be conducted.
14. On completion of the study, remove all flagging tape, markers, or other debris from the study site. Researchers wishing to repeat your study should be able to find your index trapline from tie point and site description notes.

3.5.9 Analysis Methods

The actual analysis with presence/not detected data depends on the objectives of the overall inventory effort. Table 4 highlights suggested analysis methods for inventory objectives.

Table 4. Inventory objectives and analysis methods for presence/not detected data.

Objective	Analysis Methods
Document species range	analysis to ensure adequate sampling effort. Negative binomial test
Determine habitat associations	logistic regression

Quantifying probability of detection - The main purpose of these methods is to document species geographic ranges. From a statistical point of view it is important to attempt to quantify the detection probability (as a function of population density, population spatial distribution, species trappability, sampling effort and other covariates) for a species to allow a general estimate of the optimal effort needed for surveys.

Determining habitat associations - If describing habitat associations is a key objective, then it will be important to document habitat types at the scale of species home ranges.

3.6 Relative Abundance

3.6.1 Recommended Method: Trapping with Animal Marking

Single and Multiple species inventory

The main approach in determining relative abundance between two or more areas is completing some replicated census or count using equal effort in all areas to be compared. Since small mammals are easiest to "count" using trapping, methods of inventory at this level are similar to those at the presence/not detected level. Use the best trap for the target species (Table 3). To determine relative abundance between two or more habitat types or areas for multiple species, the use of live traps with other trap types has proven successful (Aubry *et al.* 1991, Corn and Bury 1991). A combination of live trapping and pitfall trapping along an index trapline is recommended.

Interspecific density comparisons are not recommended given that capture and detection probabilities (species trappability) are not constant between species. For example, a comparison of the relative abundance of the same species in two different habitat types may be achieved using a 300-m transect, but it may not be possible to conclude that one species is twice as abundant as another in a single trap line, unless you can correct for differential trappability.

Captured animals should be marked in some manner to prevent double counting and over-estimating abundance. Marking need not necessarily identify individuals but rather identify those animals previously captured during that trapping survey.

3.6.2 Office Procedures

- Review the introductory manual No. 1 *Species Inventory Fundamentals.*
- Determine final objectives of the study (see Table 5 for examples)
- Determine species to be studied
- Determine other species expected in the project area
- Obtain maps for project and study area(s) (*e.g.,* 1:50 000 air photo maps, 1:20 000 forest cover maps, 1:20 000 TRIM maps, 1:50 000 NTS topographic maps).
- Outline the project area on a map and determine Biogeoclimatic zones and subzones, Ecoregion, Ecosection, and Broad Ecosystem Units for the project area from maps.
- Determine approximate location of study area(s) within this project area. Study areas should be representative of the project area if conclusions are to be made about the project area. For example, this means if a system of stratification is used in the Sampling Design then strata within the study areas should represent relevant strata in the larger project area.
- Stratify study areas based on habitats. This will allow you to distribute your effort in manner to maximize your ability to detect all possible species in the study area.
- Determine sampling area dimensions, trap spacing, trapping intervals
- Obtain permits
- Order and organize gear
-

3.6.3 Sampling Design

Every study involved with determining the relative abundance of a species can be conducted by putting out replicated transects of traps (index lines) within the study area. It is essential that trapping effort between areas to be compared remains constant and that sampling in all areas is concurrent. The exact dimensions of these lines as well as the type and number of traps along each of these transects will be dependent on the target species, population levels and the overall objectives of the study.

Criteria for the selection and spacing of traps along index lines is covered in the preceding presence/not detected section. Use of snap traps is not recommended for determination of relative abundance because removal of individuals from a sampling site may bias results when trapping occurs over several days and the level of bias may not be constant between sampling sites.

3.6.4 Sampling Effort

The general objective of relative abundance surveys is to determine whether or not the abundance of an individual species is different between two or more habitat types, geographically isolated areas and/or time periods. To achieve a statistical estimate of significant differences in relative abundance, sufficient replicates of sampling units need to be established. Small sample sizes will result in low statistical power resulting in a higher probability of making a *Type 2* error in your estimates.

Sampling effort can be experimentally determined through analysis of data from previous studies in similar habitat or pilot studies. Before attempting to determine sample size, the distributions of the count data derived from these preliminary studies have to be examined. Count data generated through cluster sampling usually follow one of three distributions: the Poisson (Normal), overdispersed Poisson or negative binomial. In general the species covered in this manual tend to have clumped populations and clumped populations do not fit normal distributions but rather conform better to negative binomial distributions.

Choice of the approach to estimating sample sizes needed for counts is dependent on the nature of the distribution because sampling properties of these distributions differ. Methods of determining sample sizes for different data distributions are covered extensively in Krebs (1989) and fine details of this process will not be covered here.

Another issue related to sampling effort is the number of traps placed at each sampling capture station on an index line (transect). If the population densities of local species are sufficiently high, then trap saturation may occur. That is, if all traps are filled with animals before they are checked and there are still animals at large and potentially trappable, then insufficient numbers of traps have been set out. Sufficient traps have been set out to enumerate a population if >20% of the traps remain empty following a night of trapping (Southern 1973, Gurnell 1976).

3.6.5 Equipment

In addition to the equipment outlined for conducting presence/not detected surveys, material and equipment required to mark animals is necessary.

3.6.6 Field Procedures

The field procedures for relative abundance are largely the same as those for presence/not detected surveys. The only addition to the field procedures is that animals need to be marked upon capture using a method that will last as long as the sampling survey. Captured animals do not necessarily have to be marked with unique identifiers as fate of individuals over time is not an issue when determining relative abundance.

The frequency of trap checking should be such that chances of trap mortality is minimized. Note that leaving pitfalls unchecked for 24 hours will likely result in the mortality of a significant proportion of animals captured; therefore, traps need to be checked at least twice daily when pitfalls are used in conjunction with live-traps. If shrews are targeted, pitfall traps should be baited and checked more frequently.

3.6.7 Analysis Methods

The analysis of count data can be statistically complex given that such data are rarely normally distributed, which is a standard assumption of commonly used parametric analysis methods. Where data do not meet the assumptions of parametric tests (normality, independence of observations, equal variances) parametric tests may still be robust to minor departures from normality providing that *n* (sample size of counts) is large ($n>20$ in each sample) and equal between samples (Zar 1996).

Where data do not meet the assumptions required for parametric tests, nonparametric tests such as the Kruskal-Wallace test can be used. These methods make fewer assumptions regarding the distribution of data, but they also have much lower statistical power than parametric methods.

This manual is not intended to provide an in depth guide to the use of statistics in analyzing inventory data, but rather direct biologists towards proper methods of analysis. Table 5 provides some insight into a selection of methods for analyzing count data. You are encouraged to review these and other potential methods in more depth. For instance, robust data-based methods of analysis (negative binomial comparisons, randomization, bootstrap, jackknife) have been developed for circumstances where parametric methods are not suitable and may provide better statistical power than nonparametric methods.

Table 5. Inventory objectives and analysis methods for relative abundance data.

Objective	Analysis Methods
Comparison in abundance between areas	ANOVA or nonparametric method Power analysis
Determine whether habitat alterations have altered population size	T-test or nonparametric method Power analysis

3.6.8 Assumptions of analysis methods

The main assumptions of relative abundance methods as applied here are:

1. Identical or statistically comparable methodologies are used when the objective of inventory effort is to compare between areas. (Sampling methods and sample sizes are the same in all areas).
2. Environmental, biological, and sampling factors are kept as constant as possible to minimize differences in survey bias and precision between surveys
3. Surveys are independent; one survey does not influence another.
4. Capture probability (see section 3.2.2) is similar between areas.

3.7 Absolute Abundance

3.7.1 Recommended Method: Mark-recapture

Absolute abundance for the species covered in this manual can be best estimated by using a mark-recapture approach. In addition to estimates of abundance, mark-recapture studies can provide information on fundamental demographic parameters such as reproductive rates, immigration rates, mortality rates and emigration rates. These methods have been used in ecological research for several decades and a large body of literature on applications and theoretical development exists (see Otis *et al.* 1978, Seber 1982, White *et al.* 1982, Seber 1986, Krebs 1989, and Pollock *et al.* 1990 for good reviews). Typically, these methods are quite intensive and require relatively complicated analysis methods.

Population models used in the estimation of abundance using mark-recapture data have been classified as those suitable for open or closed populations. Choice of model and study design will depend on objectives of the survey and how the various models will fulfill the objectives (Table 6). Ultimately, the choice of model will depend on how well the assumptions of the models are met by the study design and associated data to be analyzed. Before describing the field procedures required for mark-recapture studies the objectives, models of estimation and assumptions of these models will be briefly discussed.

3.7.2 Objectives of surveys

Although the fundamental focus of many inventories is to determine the population level, population performance, as affected by birth rates, immigration, mortality and emigration, is an equally important parameter, especially when comparing populations. For the most part population size can best be determined by closed population models, and population performance parameters can only be assessed through open population models (Table 6). The choice of which models are used to analyze data will affect study design and field procedures.

Table 6. Inventory objectives and analysis methods for absolute abundance data.

Objective	Analysis Methods
Estimate population density at one particular point in time	Mark-recapture analysis using a closed population model
Estimate demographic parameters such as survivorship	Mark-recapture analysis using an open population model

3.7.3 Open vs closed populations

It is important that biologists attempting to use mark-recapture methods have a thorough understanding of the difference between an open and a closed population. Open populations are those that may experience additions (births and/or immigration) as well as deletions (deaths and/or emigration) over the duration of the survey. Closed populations are those that do not experience any permanent additions or deletions during the survey, and by this definition, remain theoretically constant in size over this period (see Section 3.7.4 for an explanation of the term "survey").

3.7.4 Models of estimation and methods of analysis

The concept of open versus closed populations forms the basis for two fundamentally different approaches to determining absolute abundance and related demographic parameters through mark and recapture of animals. The most commonly used closed models are the Lincoln-Peterson ratio estimator (see Seber 1982:59) and a series of models collectively referred to as CAPTURE (White *et al.* 1982). The most common open model used in mark-recapture studies is the Jolly-Seber model. Each of these will be briefly discussed below. These models are ultimately used to analyze the mark-recapture data generated by the methods outlined below.

Lincoln-Peterson ratio estimator - for closed populations

The Lincoln-Peterson estimator is the simplest form of mark-recapture model and has been used in animal population ecology since the early part of this century. The premise of this method is as follows:

> If a sample of animals (n_1) is captured, marked and released into the population at some point in time (t_1), and a subsequent sample of the same population (n_2) (some of which (m_2) will be marked) is captured at some later date (t_2), then the ratio of marked (m_2) to unmarked (n_2) animals in the second sample will be the same as the ratio of the animals caught in the original sample (n_1) to the total population size.

Assumptions of the Lincoln-Peterson method are as follow:

1. The population is closed.
2. All animals are equally likely to be captured in each sample (equal capture probabilities, see section 3.2.2).
3. Marks are not lost or overlooked by the observer.

In some cases some of the assumptions can be relaxed. For example, if there are only additions to the population between t_1 and t_2, then the additions are all unmarked, and the Lincoln-Peterson estimate of population size is valid at t_2. Deletions from the population between t_1 and t_2 are not as easily handled by the method. Furthermore, variations in individual capture probabilities will either negatively or positively bias estimates depending on the nature of this variation (heterogeneity or behaviour). If marks are lost in the population, then the population estimate will be positively biased because the ratio of m_2 to n_2 will be too small. Seber (1982: 70-104) provides a detailed account of the Lincoln-Peterson assumptions, effects of deviations from these assumptions, and tests for detecting such deviations.

Although this method clearly has limitations, a modified version of it is central to all open population models and is fundamental to the Jolly-Seber model (discussed below).

CAPTURE Models- for closed populations

As discussed, Lincoln-Peterson estimates will be biased if unequal capture probabilities exist in the population being trapped. For this reason, robust models incorporating various sources of capture probability variation have been developed in relatively recent years (Otis *et al.* 1978, White *et al.* 1982). Models within the program CAPTURE have been developed to specifically account for variability associated with heterogeneity, behaviour, and time bias and some combinations of these.

Although the assumption of equal capture probability is relaxed within these models certain assumptions still apply. These include:

1. The population is closed.
2. Marks are not lost or overlooked by the observer.

Even with these more robust population models, there are critical study design factors that need to be considered. Three principle issues should be considered when conducting a study using CAPTURE:

1. *Obtaining adequate sample sizes* - The population on the trapping grid, population capture probability and the number of successive sampling nights (CAPTURE sessions) all influence sample size. Typically, trapping over four or five trap nights (CAPTURE sessions) should be conducted. The number of sample trap nights may have to be increased if population densities or capture probabilities are low. See White *et al.* (1982) and Menkins and Anderson (1988) for discussions of sample size issues with CAPTURE.
2. *Meeting the assumption of closure* - Trapping should be conducted over a brief time interval so that additions and deletions to the population are minimized. Trap mortality must be minimized during surveys by choosing the appropriate type of trap and checking it with the required frequency. Loss of significant numbers of individuals due to trap death will violate the closure assumption.
3. *Minimize capture probability variation* - Trapping surveys should be designed to minimize heterogeneity, time and behaviour bias, if it logistically possible. For instance, heterogeneity bias may be minimized by conducted trapping on grids that are large relative to the home range size of the target species. Behaviour bias may be minimized by limiting the length of time animals are stressed by reducing the physical handling time at the capture (trap) station. As surveyors become familiar with handling animals the handling time will decrease. Using soft cotton gloves as opposed to leather gloves will make it easier to process animals and also reduce handling time. Time bias may be limited by conducting the capture (trapping) sessions in a short period of time and under relatively constant weather conditions (see point 2 above).
4. *Minimize chance of loss of marks* - If applied properly, ear tags remain attached to animals for considerable periods of time. However, there is always a chance that tags may fall off or get ripped out of the ear. Double tagging of captured animals (one tag in each ear for instance) will allow for much longer mark retention. It is unlikely that both tags will fall out between successive sampling capture sessions, but if one does, a second new tag can be attached.

Jolly-Seber Model - for open populations

In some instances it is difficult to ensure that populations remain closed over the duration of inventory studies. Also, when information on population performance and associated demographic parameters is required to meet study objectives, open population models are needed to analyze mark-recapture data. Under this circumstance, the Jolly-Seber model is recommended. This model is based on the Lincoln-Peterson model and therefor is constrained by similar assumptions except that of population closure. One additional assumption of the Jolly-Seber model is that every marked animal present in the population immediately after a sample has the same probability of survival until the next sampling time.

For the most part, the Jolly-Seber is susceptible to biases if unequal capture probabilities are exhibited in the trapped population. Newer versions of the model have accommodated age-specific capture probabilities and survivorship rates which can reduce some biases in due to heterogeneity. The Jolly-Seber approach to survival modeling has been extended to allow for testing of biological hypotheses using generalized linear models. These methods allow for more flexibility and allow for the use of covariates (such as weather factors) to minimize bias of estimates.

3.7.5 Recommended Models

Either of the two models discussed above are appropriate for estimating absolute abundance of a target small mammal species: the Jolly-Seber for an open population and the Lincoln-Peterson for a closed one. A third “robust” method that incorporates both open and closed models as outlined by Pollock *et al.* (1990) may also be applied; however, the intensity of survey required for this method may not be affordable for most inventory projects. All three approaches involve area based trapping (within a two dimensional grid of traps as opposed to the one dimensional index lines used for presence or relative abundance surveys) and successive capture - mark - recapture sessions. Despite this similarity, it is useful to understand that all three models differ fundamentally in their requirements for trap nights and sessions.

For the purposes of inventory projects, trapping events can be divided into trap nights and capture sessions (Figure 2). A session represents a series of days over which trapping occurs, usually, without interruption. It is composed of trap nights, each which represents an individual trap checking event or combinations of several trap checking events if trap checking frequencies are high (e.g. for shrews). For small mammals, a trap night is 24 hours long. Using the Jolly-Seber model, there are a number of sessions (k) consisting of two trap nights (l) over the duration of the inventory. Using the CAPTURE models, there is five sessions consisting of one or more consecutive trap nights. And for the combined method of Pollock *et al.* (1990) there are a number of sessions consisting of five or more trap nights (Table 7).

Figure 2. Diagramatic representation of study designs for RIC mark-recapture studies.

Table 7. Study design criteria for mark-recapture methods of inventory for small mammals.

Model	**Number of Sessions**	**Number of Trap Nights per Session**	**Interval between Sessions**	**Grid Size (stations per axis)**	**Inter-station distance**
Jolly-Seber Open model	8 or more	2	3 - 4 weeks	7 x 7 or <	14.29 m
CAPTURE Closed model	5	1	continuous sessions	16 x 16 or <	7.5 - 15 m
Pollock's Robust model	8 or more	5 or more	3 - 4 weeks	16 x 16 or <	7.5 - 15 m

3.7.6 Office Procedures

- Review the introductory manual No. 1 *Species Inventory Fundamentals.*
- Determine final objectives of the study (see Table 4 for examples)
- Determine species to be studied
- Determine other species expected in the project area
- Obtain maps for project and study area(s) (*e.g.,* 1:50 000 air photo maps, 1:20 000 forest cover maps, 1:20 000 TRIM maps, 1:50 000 NTS topographic maps).
- Outline the project area on a map and determine Biogeoclimatic zones and subzones, Ecoregion, Ecosection, and Broad Ecosystem Units for the project area from maps.
- Determine approximate location of study area(s) within this project area. Study areas should be representative of the project area if conclusions are to be made about the project area. For example, this means if a system of stratification is used in the Sampling

Design then strata within the study areas should represent relevant strata in the larger project area.

- Stratify study areas based on habitats. This will allow you to distribute your effort in manner to maximize your ability to detect all possible species in the study area.
- Determine sampling area dimensions, trap spacing, trapping intervals
- Obtain permits
- Order and organize gear

3.7.7 Sampling Design

The sampling designs outlined in Table 7 represent a range of designs that can be applied in absolute density studies. Finer details than are presented in Table 7 should be considered when assembling your final study design. Remember, the study design should be chosen to minimize any violations of assumptions with respect to the estimation model you will ultimately be using.

Grid layout and capture session length

Open population sampling

The following grid sampling design is recommended for monitoring changes in small mammal populations (Ritchie and Sullivan 1989) and has been applied during many small mammal population studies in B.C. (Sullivan 1979, Sullivan 1980, Sullivan and Krebs 1981, Sullivan and Sullivan 1981, Sullivan and Sullivan 1982, Sullivan *et al.* 1983). During their studies, Sullivan *et al.* used grids consisting of 7 rows of 7 traps in a square grid with capture (trap) stations spaced at 14.29 m with one Longworth live trap (or more where animal densities are greater than 50/ha) per station. This trap spacing allows the grid to cover 1 ha. It should be noted that these grids were used specifically to monitor rodent species. Capture (trapping) sessions should last at least two trap nights, and should be conducted at least 8 times at 3 week intervals during the active season from May to October.

Closed population sampling

To estimate absolute abundance of a closed population at one static point in time, a 16 x 16 grid is recommended (see White *et al.* 1982). A maximum trap spacing of 15 m is recommended (which for 'typical' small mammals would include at least 4 home ranges). One live trap is set for each of 256 stations unless population densities are high, in which case more can be set to avoid competition for traps. At least five trap nights of data are recommended: these will be treated as five sessions by program CAPTURE. This method is suggested as a one time sampling procedure which may be undertaken at any time during the active breeding season (White *et al.* 1982). In contrast, the same five nights would be treated as one of 8 five night sessions by Pollock's model (Table 7).

Choice of traps and trap checking frequency

Live-traps have to be used for absolute abundance inventories. In inventory studies that do not include shrews, the layout should not include pitfall traps. This will alleviate the high frequency of trap checking that is required to ensure minimal trap mortality when pitfalls are used. When doing an inventory of only shrew species, the layout will be composed of pitfall traps only and pitfall traps could be constructed with drift fences (see section 3.1.4). If the study objective includes shrews and other species groups, both traps types (live and pitfall traps) should be used and pitfall traps should be checked every 1.5 to 2 hours during trapping. If this high rate of trap checking is not logistically possible, pitfalls can be deactivated for periods longer than 2 hours provided that your sampling still yields appropriate numbers of captures for shrews.

3.7.8 Sampling Effort

There are numerous methods available to determine sample sizes necessary to ensure reasonable estimates of abundance for mark-recapture studies. The references outlined in Table 8 include graphs and discussions of appropriate sample sizes for the various models used. The determination of optimal sample sizes for CAPTURE models can be quite complex. An easy to use simulation module is available as part of the computer program CAPTURE to allow biologists to explore sample size issues.

Table 8. Sources for sample size calculations for mark-recapture population estimates.

Estimator	Source for methods of sample size calculations
Lincoln-Peterson	Krebs (1989:22)
Jolly -Seber	Pollock *et al.* (1990: 72)
CAPTURE	White *et al.* (1982)

3.7.9 Equipment

The equipment required to conduct mark-recapture studies on small mammals is the same as for relative abundance surveys with the following exception. Given that animals need to be marked with unique identifiers, alternative methods of marking are needed. Serially-numbered animal ear tags are ideal for rodent species; however, they should not be attached to shrews. Individual shrews can be given unique identity markers by dying their fur (V. Craig pers. comm.). These marking methods are able to last between capture sessions and throughout the trapping season (May to October) and may work for absolute abundance inventories of shrew populations each year. Please note that marking methods for shrews are still very much under-development. They are mentioned here to provide possibilities that may work, but it is strongly suggested that procedures be tested prior to implementation to determine that inventory objectives will be achieved.

3.7.10 Field Procedures

Once you have chosen the trapping grid layout and trap type(s) to be used the ensuing field procedures should be followed.

Single or multiple species inventory: live trapping grids:

1. Select a site that is homogeneous in habitat
2. Establish a fixed tie point (reference point which is a known distance and bearing from the first capture (trap) station on the trapping grid). Prepare detailed notes on access and the trapping grid for other field crews.

3. Establish a regularly spaced two dimensional grid of traps at the study site, according to the standard grid design (7 x 7 grid: see Ritchie and Sullivan 1989 and other papers by T. Sullivan; 16 x 16 grid: see White *et al.* 1982). Stations on the 7 x 7 grid should be spaced at 14.3 m intervals, stations on the 16 x 16 grid should be spaced between 7.6m and 15m. All points in the grid should be away from habitat edges or sites of disturbance if at all possible.
4. Use flagging tape to mark capture stations at intersection points (stations) of the grid as well as above the traps once placed. Use trees or sturdy shrubs for the flags, or strong, well-placed sticks or re-bar in open or grassland habitats. Mark the capture station number on the flagging tape at each station.
5. Place 1 trap within 2 m of the capture station (2 or more live traps may be required for sites where animal densities are >50 per ha or where <20% of the traps remain empty after a night of trapping). Live traps should be placed at microsites which might be attractive to mice or voles. Appropriate sites include positions along or under woody debris or rocks, under bushes, along worn travel trails.
6. Each trap should be baited. Slices of carrots will help avoid dehydration of some animals. Sufficient cotton bedding material should be provided. In Sherman traps it is essential that no grains, seeds, vegetable matter or bedding is allowed to hinder the function of the trap door trigger. A cover of debris, vegetation or lightweight board over the trap will decrease chances of hypothermic or hyperthermic captures. Attach a piece of coloured flagging tape to the trap or place one in branches above it for easy relocation.
7. A pre-baiting period of 2 weeks should precede sampling. Baited traps should be left at the sampling site with the doors locked open to allow the animals to familiarize themselves with the novel object in their environment. Traps should be left open and baited at the site between sampling capture sessions if possible.
8. Set the traps in the evening when the pre-baiting period is complete.
9. Check the traps the following morning as early as possible to minimize mortalities and trap stress. Under hot or cold or wet weather conditions, the trapline should be checked once in the late afternoon as well. Shrews, because of their high metabolic rates, do not survive long in live traps, if mortality is to be minimized more frequent trap checks are necessary. If endangered or threatened species are the focus of the study or are expected to be captured in the area, this afternoon check is mandatory to prevent unnecessary mortality. More frequent checks are recommended if mortality of shrews is of concern.
10. Captured individuals should be tagged or marked, identified to species, and age, sex class and reproductive status determined if appropriate to the study. Standard morphometric measurements can also be taken if necessary. These include: total length, tail length, hind foot length, ear length, and weight. Not all measurements are necessary for all animals however, weight should be taken for all. Records of each capture should include capture station number along with the biological data.
11. Trap mortalities should be described as above, and placed in individual plastic bags with a detailed data label. Mortalities should be frozen as soon as possible and prepared as voucher specimens. Check with the Royal BC Museum whether these vouchers should be added to their collection.
12. Continue these procedures till the required number of trap nights has been reached.
13. When the capture session is complete, lock open the traps and leave them at the site ('pre-baited') unless you do not intend to return. Between multiple capture sessions, traps should be left open and pre-baited. If traps must be removed from the site between

sessions (for example, because of limited numbers available), leave a similar item, such as a tin can with a little bait at each capture station to simulate the ‘pre-bait’ trap.

14. On completion of the survey, remove all flagging tape, markers, ‘pre-bait’ tin cans, or other debris from the study site. Researchers wishing to repeat your study should be able to find your trapping grid from tie point and site description notes.

Glossary

ABSOLUTE ABUNDANCE: The total number of organisms in an area. Usually reported as absolute density: the number of organisms per unit area or volume.

ACCURACY: A measure of how close a measurement is to the true value.

ALPHA (α): The probability of making a Type I error (level of significance).

BETA (β): The probability of making a Type II error.

BIAS: The mean difference from the real value of a measure by estimators of that measure. The result of systematic error in data collection.

BIODIVERSITY: Jargon for biological diversity: “the variety of life forms, the ecological roles they perform, and the genetic diversity they contain” (Wilcox, B.A. 1984 cited in Murphy, D.D. 1988. Challenges to biological diversity in urban areas. Pages 71 - 76 in Wilson, E.O. and F.M. Peter, Eds. 1988. Biodiversity. National Academy Press, Washington, D.C. 519 pp.).

BLUE LIST: Taxa listed as BLUE are sensitive or vulnerable; indigenous (native) species that are not immediately threatened but are particularly at risk for reasons including low or declining numbers, a restricted distribution, or occurrence at the fringe of their global range. Population viability is a concern as shown by significant current or predicted downward trends in abundance or habitat suitability.

CBCB (Components of B.C.’s Biodiversity) Manuals: Wildlife species inventory manuals that have been/are under development for approximately 36 different taxonomic groups in British Columbia; in addition, six supporting manuals.

CLUSTER SAMPLING: Simple random sampling in which each sample unit is a collection, or cluster, of elements.

CREPUSCULAR: Active at twilight.

DESIGN COMPONENTS: Georeferenced units which are used as the basis for sampling, and may include geometric units, such as transects, quadrats or points, as well as ecological units, such as caves or colonies.

DIURNAL: Active during the daytime.

EWG (Elements Working Group): A group of individuals that are part of the Terrestrial Ecosystems Task Force (one of 7 under the auspices of RIC) which is specifically concerned with inventory of the province’s wildlife species. The EWG is mandated to provide standard inventory methods to deliver reliable, comparable data on the living “elements” of BC’s ecosystems. To meet this objective, the EWG is developing the CBCB series, a suite of manuals containing standard methods for wildlife inventory that will lead to the collection of comparable, defensible, and useful inventory and monitoring data for the species populations.

INVENTORY: The process of gathering field data on wildlife distribution, numbers and/or composition. This includes traditional wildlife range determination and habitat association inventories. It also encompasses population monitoring which is the process of detecting a demographic (e.g. growth rate, recruitment and mortality rates) or distribution changes in a population from repeated inventories and relating these changes to either natural processes (e.g. winter severity, predation) or human-related activities (e.g. animal harvesting, mining, forestry, hydro-development, urban development, etc.). Population monitoring may include the development and use of population models that integrate existing demographic information (including harvest) on a species. Within the species manuals, inventory also includes, species statusing which is the process of compiling general (overview) information on the historical and current abundance and distribution of a species, its habitat requirements, rate of population change, and limiting factors. Species statusing enables prioritization of animal inventories and population monitoring. All of these activities are included under the term inventory.

IRRUPTION: A sudden surge in population numbers.

MARK-RECAPTURE METHODS: Methods used for estimating abundance that involve capturing, marking, releasing, and then recapturing again one or more times.

MONITOR: To follow a population (usually numbers of individuals) through time.

NEGATIVE BINOMIAL DISTRIBUTION: A spatial distribution in which the variance is always greater than the mean. Used to describe the distribution of aggregated populations (which are common in ecological study)..

NOCTURNAL: Active at night

NORMAL DISTRIBUTION: A spatial distribution approximated by a symmetric bell-shaped curve.

OBSERVATION: The detection of a species or sign of a species during an inventory survey. Observations are collected on visits to a design component on a specific date at a specific time. Each observation must be georeferenced, either in itself or simply by association with a specific, georeferenced design component. Each observation will also include numerous types of information, such as species, sex, age class, activity, and morphometric information.

PARAMETRIC METHODS: Methods of statistical analysis that assume that the population has a normal distribution.

POPULATION: A group of organisms of the same species occupying a particular space at a particular time.

PRECISION: A measurement of how close repeated measures are to one another.

PRESENCE/NOT DETECTED (POSSIBLE): A survey intensity that verifies that a species is present in an area or states that it was not detected (thus not likely to be in the area, but still a possibility).

PROJECT AREA: An area, usually politically or economically determined, for which an inventory project is initiated. A project boundary may be shared by multiple types of resource and/or species inventory. Sampling for species generally takes place within smaller, representative study areas so that results can be extrapolated to the entire project area.

PROJECT: A species inventory project is the inventory of one or more species over one or more years. It has a georeferenced boundary location, to which other data, such as a project team, funding source, and start/end date are linked. Each project may also be composed of a number of surveys.

RANDOM SAMPLE: A sample that has been selected by a random process, generally by reference to a table of random numbers.

RED LIST: Taxa listed as RED are candidates for designation as Endangered or Threatened. Endangered species are any indigenous (native) species threatened with imminent extinction or extirpation throughout all or a significant portion of their range in British Columbia. Threatened species are any indigenous taxa that are likely to become endangered in British Columbia, if factors affecting their vulnerability are not reversed.

RELATIVE ABUNDANCE: The number of organisms at one location or time relative to the number of organisms at another location or time. Generally reported as an index of abundance.

RIC (Resources Inventory Committee): RIC was established in 1991, with the primary task of establishing data collection standards for effective land management. This process involves evaluating data collection methods at different levels of detail and making recommendations for standardized protocols based on cost-effectiveness, co-operative data collection, broad application of results and long term relevance. RIC is comprised of seven task forces: Terrestrial, Aquatic, Coastal/Marine, Land Use, Atmospheric, Earth Sciences, and Cultural. Each task force consists of representatives from various ministries and agencies of the Federal and BC governments and First Nations. The objective of RIC is to develop a common set of standards and procedures for the provincial resources inventories. [See http://www.for.gov.bc.ca/ric/]

SAMPLE: A collection of sampling units drawn from a population.

SAMPLING UNITS: Non-overlapping collections of elements which are intended to be representative of a population. For RIC small mammal surveys, these will be index trap lines or grids.

SESSION: One or more trap nights grouped together for the purpose of statistical analysis, usually involving mark-recapture methods.

SPI: Abbreviation for ‘Species Inventory’; generally used in reference to the Species Inventory Datasystem and its components.

STRATIFICATION: The separation of a sample population into non-overlapping groups based on a habitat or population characteristic that can be divided into multiple levels. Groups are homogeneous within, but distinct from, other strata.

STUDY AREA: A discrete area within a project boundary in which sampling actually takes place. Study areas should be delineated to logically group samples together, generally based on habitat or population stratification and/or logistical concerns.

SURVEY: The application of one RIC method to one taxonomic group for one season.

SURVIVORSHIP: The probability of a new-born individual surviving to a specified age.

SYSTEMATIC SAMPLE: A sample obtained by randomly selecting a point to start, and then repeating sampling at a set distance or time thereafter.

TERRESTRIAL ECOSYSTEMS TASK FORCE: One of the 7 tasks forces under the auspices of the Resources Inventory Committee (RIC). Their goal is to develop a set of standards for inventory for the entire range of terrestrial species and ecosystems in British Columbia.

TRAP NIGHT: A 24-hour period in which a trap is active i.e. set and capable of capturing the focal species or tripped and occupied by the focal species.

TYPE I ERROR: The null hypothesis is rejected even though it is true.

TYPE II ERROR: The null hypothesis is accepted even though it is false

YELLOW-LIST: Includes any native species which is not red- or blue-listed.

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