Inventory Methods for Moles and Pocket Gopher

Standards for Components of British Columbia's Biodiversity No. 26

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Ministry of Environment, Lands & Parks
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Preface

This manual presents standard methods for the inventory of moles and pocket gophers 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. *Inventory Methods for Moles and Northern Pocket Gopher* is labeled as Version 2.0, in keeping with the rest of the series, even though Version 1.0 has never been published. The CBCB 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 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]. 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.elp.gov.bc.ca/rib/wis/spi/

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:

Conservation Biology Section,
Resource Inventory Branch
Ministry of Environment, Lands & Parks
P.O. Box 9344, Station Prov Govt
Victoria, BC V8W 9M1
spi_mail@victoria1.gov.bc.ca

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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 access the Resources Inventory Committee Website at: http://www.for.gov.bc.ca/ric.

Terrestrial Ecosystems Task Force

The current version of this manual is the result of the hard work of S. Tim Sheehan, Virgil C. Hawkes and Mark A. Fraker. The background information and protocols presented in this document are based on the unpublished government report, *Sampling Techniques for Fossorial Mammals: Moles and Pocket Gophers*, prepared by S. Tim Sheehan and Carlos Galindo-Leal. Figures 1-5, 8 and 9 are taken from the Royal B.C. Museum handbook *Opossums, Shrews and Moles of British Columbia* by David W. Nagorsen; and are reprinted with permission. Figures 6, 7, 10, 12 and 13 are original artwork by S. Tim Sheehan. All decisions regarding protocols are the responsibility of the Resources Inventory Committee.

The Components of British Columbia's Biodiversity series is currently edited by James Quayle and Leah Westereng.

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1. INTRODUCTION

In general, fossorial mammals are small mammals with long pointed snouts that are covered with short, close-set fur. They walk in a plantigrade style and possess short limbs that are usually equipped with a full compliment of five digits. They have a highly developed sense of touch and smell and small eyes and ears that may be barely visible (Gorman and Stone 1990). Most of them are also solitary or live in a simple social structure and survive on an invertebrate diet. This manual will focus on fossorial mammals commonly known as moles and gophers.

In British Columbia there are three mole species in the family Talpidae; the Coast Mole (*Scapanus orarius)*; the Townsend's Mole (*Scapanus townsendii)*, and the Shrew-mole (*Neurotrichus gibbsii)*. Both the Coast Mole and the Shrew-mole are extensively distributed throughout the Lower Mainland. In contrast, the Townsend's Mole is found only within a 15 to 20 km^2 area around Huntingdon and Abbotsford (Sheehan and Galindo-Leal 1996[a]). Both *Scapanus* spp. are considered pests by various landowners because of the damage, real or perceived, created by their tunneling activities; as a result they frequently become the focus of indiscriminate kill-trapping efforts.

There is also one gopher species that exists in BC (Cowan and Guiguet 1973; Nagorsen 1990), the Northern Pocket Gopher (*Thomomys talpoides)* that belongs to the family Geomyidae. It inhabits the dry southern interior and the southern Kootenays and Rocky Mountains of BC (Nagorsen 1990). More specifically, these animals are present from the Manning Park region east to the Alberta border and north from the international border to the Thompson and Fraser Rivers.

Habitat loss and fragmentation are the critical issues facing long term mole persistence in the Fraser Valley. The population of Townsend's Mole on Sumas Mountain is currently threatened from residential development. In the Abbotsford area, a sizable percentage of the land that is occupied by the Coast Mole and the historical sites where the Townsend's Mole specimens have been collected are in the Agricultural Land Reserve. Although this land is not at an immediate risk to urbanization intensive agriculture is a continuous threat to the Townsend's Mole (Sheehan 1999).

Glendenning (1959) reported that moles had only 'recently' multiplied to become pests to Fraser Valley farmers. The increase in mole activity/abundance was attributed to agricultural practices such as intensive cultivation and heavily manured fields. Increased arable land and soil fertility corresponded to a rise in earthworm numbers that provided a growing supply of ideal mole habitat. In fact, Glendenning (1959) suggested that the presence of earthworms in the area prior to agriculture is a 'disputed point' and that their abundance was 'scarce' if present at all, when compared to current concentrations. Therefore, farming practices from the past may have contributed significantly to the problems present-day farmers have with moles in the Fraser Valley.

Moles are credited with being beneficial to agriculture by improving the aeration and drainage of the soil, consuming harmful soil invertebrates, and circulating soil minerals (Kuhn and Edge 1990). Others authors (Glendenning 1959; Stone 1989; Gerber 1995) believe that these advantages have been replaced by modern agricultural practices and that any benefits accruing to agriculture, due to mole activity, are now grossly outweighed by their damage.

Most mole studies relate to the economic damage caused by this small mammal (Glendenning 1959; Pedersen 1963, 1966; Giger 1973; Schaefer 1978). Often, the objective of studying moles is to obtain information to improve the effectiveness of mole control efforts that range from trapping to poisoning to the use of deterrents (Glendenning and MacCarthy 1965; Hawthorne 1980; Gorman and Stone 1990; Gerber 1995). In contrast, this manual is not intended to prescribe techniques for eradicating the focal species but rather to study them with as little intervention as is necessary. The purpose of this manual is to recommend methods and protocols for determining presence and abundance of moles and pocket gophers in BC.

2. INVENTORY GROUP

Below is the current taxonomy as of April 2001. If you are concerned that this may have changed, you will always find the most current taxonomy in manual No. 5, *The Vertebrates of British Columbia: Scientific and English Names* (RIC).

Order Insectivora, Family Talpidae:

Neurotrichus gibbsii (Baird)	Shrew-mole	M-NEGI
ssp: *gibbsii* (Baird)		
Scapanus orarius True	Coast Mole	M-SCOR
ssp: *schefferi* Jackson		
Scapanus townsendii (Bachman)	Townsend's Mole	M-SCTO
ssp: *townsendii* (Bachman)		

Order Rodentia, Family Geomyidae:

Thomomys talpoides (Richardson)	Northern Pocket Gopher	M-THTA
ssp: *cognatus* Johnstone		M-THTA-CO
fuscus Merriam		M-THTA-FU
incensus Goldman		M-THTA-IN
medius Goldman		M-THTA-ME
saturatus Bailey		M-THTA-SA
segregatus Johnstone		M-THTA-SE

The family Talpidae is divided into three subfamilies: Talpinae (New and Old World moles and American and Japanese shrew-moles); Uropsilinae (Chinese or Asiatic shrew-mole); and Desmaninae (the Russian and Pyrenean desman). Only the subfamily Talpinae with its seven species is present in North America: the Hairy-tailed mole, *Parascalops breweri*; the Broad-footed mole, *Scapanus latimanus*; the Eastern American mole, *Scalopus acquaticus*; the Star-nosed mole, *Condylura cristata*; the Coast Mole, *Scapanus orarius*; the Townsend's Mole, *Scapanus townsendii*; and the Shrew-mole, *Neurotrichus gibbsii* (the latter three occur in BC).

Five subspecies of *Neurotrichus gibbsii* are recognized, but only *N. g. gibbsii* exists in British Columbia (van Zyll de Jong 1983; Nagorsen 1990). Both *Scapanus townsendii* and *Scapanus orarius* have two subspecies. However, only *S. t. townsendii* and *S. o. schefferi* occur in this province (Hartman and Yates 1985; Nagorsen 1990; Carraway *et al.* 1993). In Canada, the *Scapanus* and *Neurotrichus* genera are found only in southwestern British Columbia.

Thomomys talpoides belongs to the family Geomyidae and it is the only species of this family that exists in BC (Cowan and Guiguet 1973; Nagorsen 1990). Based upon pelage colour and skull characteristics, six subspecies of *Thomomys talpoides* are recognized in the province (Nagorsen 1990).

2.1 Shrew-mole *(Neurotrichus gibbsii)* M-NEGI

Description

The Shrew-mole is a small mole that has shrew-like features (Figure 1). Its grey to black fur is directed backwards like a shrews and it is not as velvety as the fossorial mole's. Its feet and tail are scaly. It has minute eyes, no external ears and a long, flattened nose. Its front feet are longer than they are wide and have long, curved claws (Nagorsen 1996). Average measurements are: total length 105 mm, tail 35 mm, hindfoot 15 mm and weight 10 g.

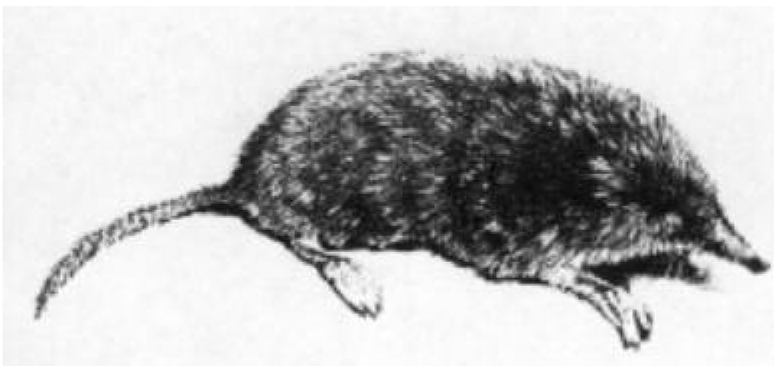

Figure 1. Shrew-mole (Nagorsen 1996)

Distribution

Species Range

In Canada, Shrew-mole distribution is limited to southwestern British Columbia. This species ranges east from the Sechelt region to Hope and Manning Park. In the Unites States it extends southwards along the Pacific slope to central California (van Zyll de Jong 1983).

Provincial Range

The Shrew-mole is widely distributed in BC, occurring across the southwestern portion of the province from the lower Fraser Valley and Cascade Mountains east to Manning Provincial Park (Figure 2). The northern limits of its range are the Sechelt Peninsula and Boise Creek, north of Pitt Lake. Some localized populations may inhabit some of the wet valleys on the east side of the Cascade Mountains (Cowan and Guiguet 1973; Nagorsen 1996).

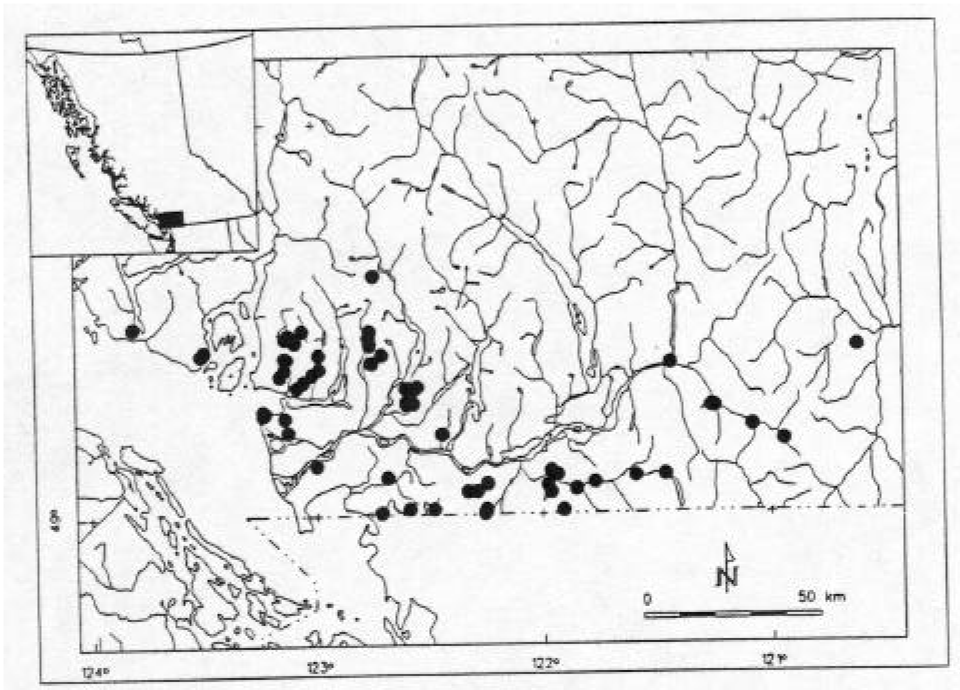

Figure 2. Shrew-mole distribution in BC (Nagorsen 1996)

Density Patterns

Little information is available on population numbers, but densities of 12 – 15 Shrew-moles per hectare have been reported in ideal habitats in Washington (Nagorsen 1996). The Shrew-mole is a shy, secretive animal that is rarely seen. In the Lower Mainland, Campbell and Hochachka (2000) reported nine captures in 932 trap nights, Zuleta and Galindo-Leal (1994) cite capture rates of 0.1 -0 .23 per 100 trap nights and Kremsater *et al.* (1993) reported only two captures after 1100 trap days. Home range and movements have not been determined.

Activity Patterns

The Shrew-mole is frequently active above ground and is surprisingly agile and quick if disturbed (Dalquest and Orcutt 1942). The Shrew-mole has no well defined activity period. However, temperature appears to influence summer surface activity. Campbell and Hochachka (2000) live trapped in early autumn and were successful only between 2300 and 0700 hours. They suggest that during this time of year the surface activity of the Shrew-mole is primarily nocturnal.

Status

The Shrew-mole appears on BC's Yellow list. Nagorsen (1996) states that there is concern that local populations in the lower Fraser Valley may be at risk because of rapid habitat loss and fragmentation from urban development.

Habitat Requirements

The Shrew-mole prefers forested hillsides and valley bottoms with loose soils comprised of high humus, abundant leaf litter, decaying vegetation, rotting logs and stumps (van Zyll de Jong 1983). Ravines comprised of big-leaf maple *(Acer macrophyllum)*, vine maple *(Acer circinatum)*, salmonberry *(Rubus spectabilis),* thimbleberry *(Rubus parvifloris)*, trailing blackberry *(Rubus ursinus)*, sword fern *(Polystichum munitum)*, and mosses provide ideal Shrew-mole habitat (Carraway and Verts 1991). They are also often found where standing water or mud occurs. The Shrew-mole is less numerous in swampy localities and generally avoids dry hard soils and grassy meadows (van Zyll de Jong 1983).

Most Shrew-moles have been captured in second growth forests in the Lower Mainland but have also been caught in clear-cut and old growth habitats (Seip and Savard 1992). Shrew-moles persist in a broad habitat range, from moist, mature forest to shrub habitat (Kremsater *et al.* 1993). Research from the U.S. implies that Shrew-moles are also habitat generalists (Terry 1981; Carraway and Verts 1991). However, other studies suggest a higher abundance of this species in riparian habitats (Anthony *et al.* 1987; Doyle 1990). In the Lower Mainland, the Shrew-mole does not appear to favor riparian habitats (Zuleta and Galindo-Leal 1994).

Ground cover is important to the Shrew-mole in western Oregon (Hooven and Black 1976). Most captures of this species (85%) occurred in a forested control plot comprised of an undisturbed mat of decaying litter. No Shrew-moles were found in the experimental clear-cut plot that was slashed and burned (Hooven and Black 1976). In western Washington, Shrew-mole presence was negatively correlated to open areas and forest edges (Terry 1981). Shrew-moles appeared to be dependent upon closed forests and preferred habitats where the organic component of the soil was high and they avoided areas where the soil was rocky (Terry 1981).

Conspicuousness and Distinctiveness of Sign

There are no definitive habitats that can distinguish the Shrew-mole from the other two mole species in BC where their ranges overlap. However, when identifying Shrew-mole specimens three external traits may be used to distinguish it from the Coast and Townsend's Moles: a small, shrew-like body; a longer, hairier tail (>25% of total length); front claws and feet that are only moderately suited for excavating (Figure 3); and 36 teeth instead of 44. *Neurotrichus gibbsii* is significantly different from *Scapanus spp.* and researchers are strongly encouraged to view all three simultaneously at a vertebrate museum, which will help minimize confusion in the field.

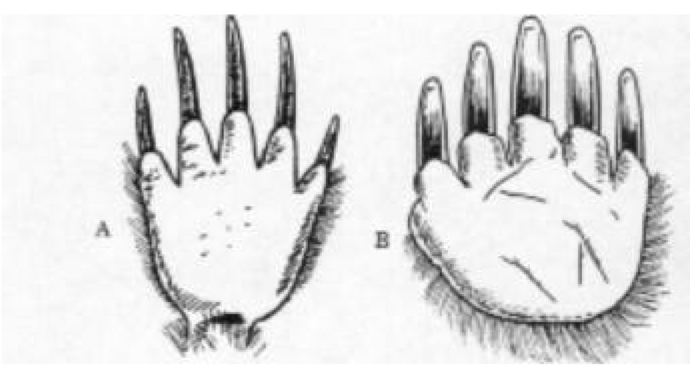

Figure 3. Shrew-mole (l) and *Scapanus* front foot (Nagorsen 1996)

Feeding

The Shrew-mole does not appear to be as dependent upon earthworms for its survival as the other two moles in the province are. Earthworms represent only 42% of its diet, by volume, and isopods (sowbugs) represent 36%. Insect larvae and pupae are also important (van Zyll de Jong 1983). Whitaker *et al.* (1979) examined eleven stomachs and discovered the existence of 11 types of prey. Earthworms were the most prevalent, occurring in 82% of the stomachs, accounting for 49% of the volume, followed by centipedes (Chilopoda), snails and slugs (Mollusca), insects (Diptera and Coleoptera), unidentified insects and unidentified vegetation.

Generally, this species consumes invertebrates that are damaging to trees and plant life. However, its diet does not differ dramatically from that of the fossorial moles. However, *N. gibbsii* appears to vary its diet seasonally. In September, 75-88% of its diet are invertebrates while in July a great proportion is conifer seeds (36%) and lichens (32%) (Carraway and Verts 1991). Reed (1944) kept a Shrew-mole in captivity for two weeks by feeding it a diet of winged adult termites and earthworms. Attempts to feed the animal meal-worms and the viscera of a freshly killed mouse were unsuccessful. Campbell and Hochachka (2000) successfully kept six Shrew-moles in captivity for almost four weeks on a diet of water and a mixture of earthworms, mealworms and sow bugs.

Reproduction

Information pertaining to the life expectancy and reproductive rates of the Shrew-mole in BC is presently unavailable.

Predators

Natural enemies of the Shrew-mole include: weasels (Silver 1933), great horned owls (Maser and Brodie 1966), barn owls (Giger 1965), red-tailed hawks (Silver 1933; Roest 1952), coyotes (Toweill and Anthony 1988), screech owls (Dalquest and Orcutt 1942), long-eared owls (Reynolds 1970) and northern saw-whet owls (Foreman and Maser 1970). Domestic cats and dogs predate upon *Neurotrichus* in urban areas (Sheehan, pers. comm. 2001).

Herons, crows, foxes and mink are known predators of the European Mole *Talpa europa* (Gorman and Stone 1990) and possibly prey upon *Neurotrichus* and *Scapanus* spp. The extent to which these natural predators control mole populations is presently unknown.

2.2 Coast Mole *(Scapanus orarius)* M-SCOR

Description

The Coast Mole has luxurious light to dark grey short velvety fur, which offers no resistance to the touch. Its most striking characteristic are its broad front feet, each possessing five long, flat claws (Figure 4). In contrast, its small hind feet have short, relatively weak claws. The Coast Mole's body is cylindrical; it has tiny eyes which are hidden by fur, a long sparsely-haired snout and a short, almost naked, double-tapered tail. Average measurements are: total length 162 mm, tail 31 mm, hind foot 21 mm and weight 67 g (Sheehan and Galindo-Leal 1996[b]).

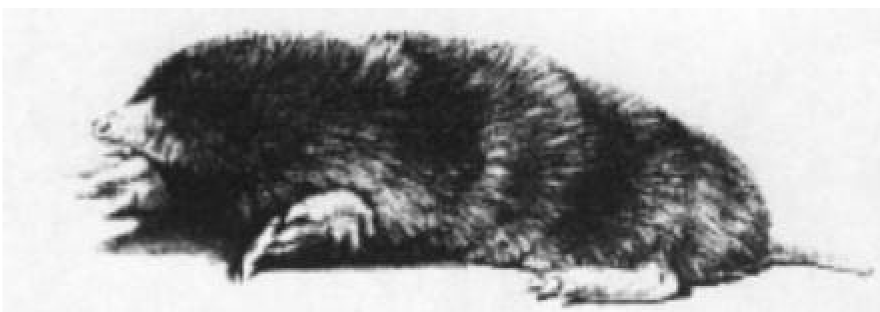

Figure 4. Coast Mole (Nagorsen 1996)

Distribution

Species Range

This mole occupies the Pacific coast north from California to southwestern British Columbia and eastern Washington and Oregon (Nagorsen 1996). The eastern limits of its range are in extreme western Idaho (Hartman and Yates 1985).

Provincial Range

The Coast Mole range is more extensive than that of the Townsend's Mole. The Coast Mole is found throughout the Puget sound Lowlands north to Vancouver and Agassiz on the north side of the Fraser River (Figure 5). Its range extends up the Fraser Valley to the Boston Bar Area (Cowan and Guiguet 1973; Nagorsen 1996).

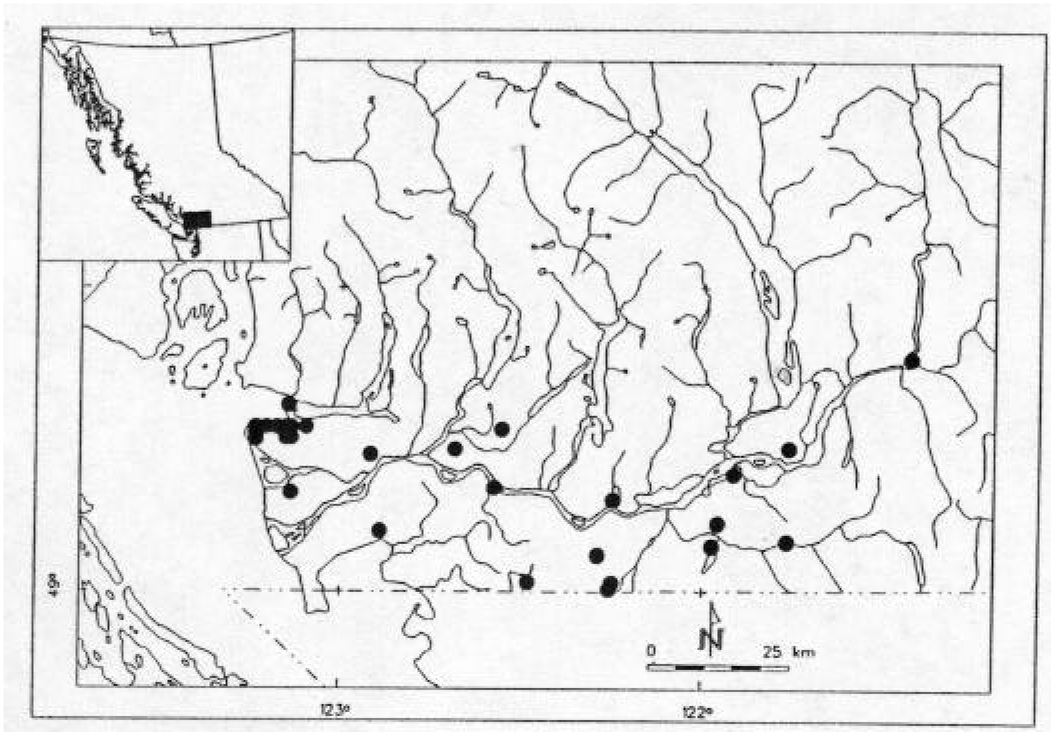

Figure 5. Coast Mole distribution in BC (Nagorsen 1996)

Density Patterns

Home range size for Coast Moles averages 0.12 hectares and density estimates for BC range from less than one mole per hectare to 13 per hectare (Nagorsen 1996). Population numbers correlate with the physical characteristics of the soil and its associated biomass.

Activity Patterns

The Coast Mole is active throughout a 24 hour period. Fresh mole mounds can be found throughout the day and the presence of new mounds in the morning indicates nocturnal activity. Researchers have directly captured and live trapped the Coast Mole during daylight hours, which refutes the notion that this species is primarily nocturnal (Schaefer 1978; Sheehan and Galindo-Leal 1997).

Status

The Coast Mole appears on BC's Yellow list.

Habitat Requirements

Coast Moles are not dependent upon a specific habitat. They can be found in agricultural land, grassy meadows and sagebrush, alder, dogwood, yellow pine and Douglas fir forests; spruce and hemlock woodlands (Hartman and Yates 1985). This mole prefers lighter, better-drained soils than those inhabited by the Townsend's Mole (Hartman and Yates 1985) and can be found in soils with a high-gravel content (van Zyll de Jong 1983). Throughout the fragmented landscape where this mole persists in BC, it is found along roadway shoulders, in recreational fields and residential lawns and gardens.

Conspicuousness and Distinctiveness of Sign

The only mole that can be confused with this species is the larger Townsend's Mole. Physically, the total length of the adult Coast Mole is <180 mm, hind foot <24 mm and the condylobasal length of the skull is < 37 mm (van Zyll de Jong 1983). Also, the Coast Mole is unlikely to exceed 90 g in weight (Schaefer 1978; Sheehan and Galindo-Leal 1996[b]).

Coast Moles produce hundreds of mounds annually in the construction, maintenance and extension of their underground tunnel systems. Sheehan and Galindo-Leal (1997) calculated the average Coast Mole mound to be 30 cm in diameter and 11 cm in height with a corresponding average tunnel diameter (measured from top to bottom of tunnel) of 3.6 cm (Figure 6).

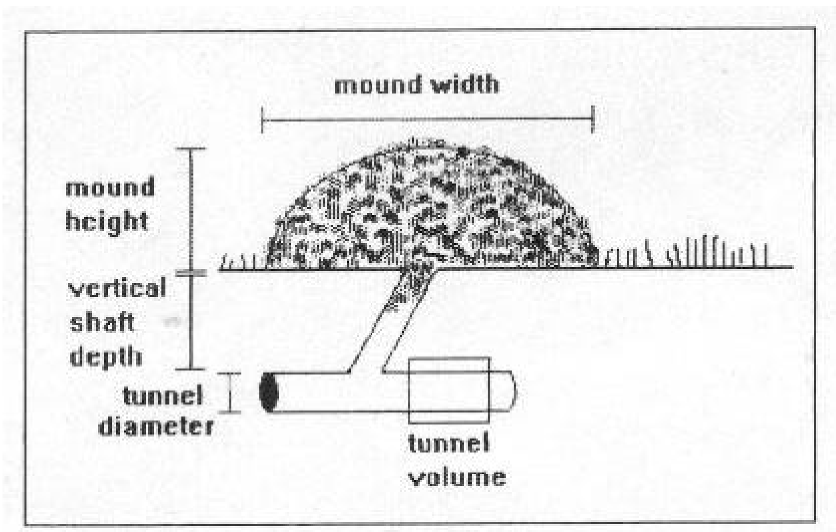

Figure 6. Mound and tunnel measurements for *Scapanus* spp.

Scapanus spp. in BC produce cloddy, conical mounds that are distinguishable from those of the pocket gopher which are generally fan or crescent shaped and comprised of fine, sifted dirt (Figure 7).

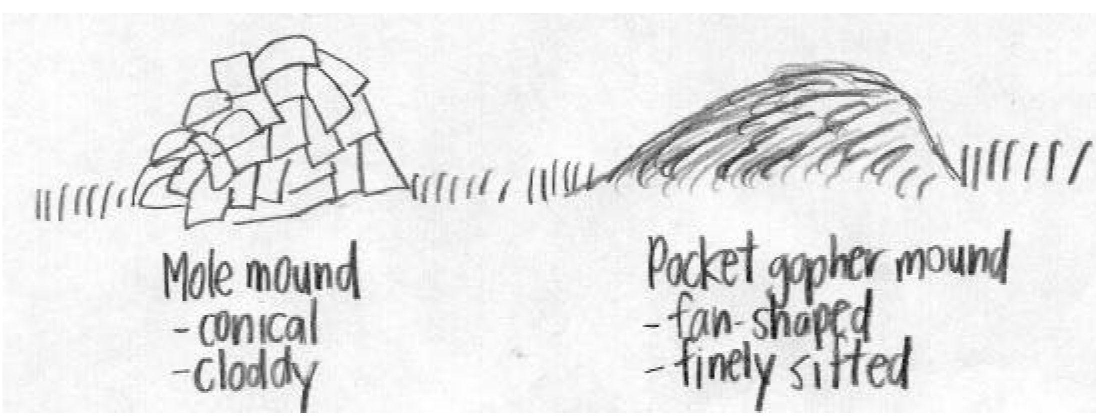

Figure 7. Mole and Pocket Gopher mound differences

Feeding

Coast Moles are principally dependent upon soil invertebrates for their sustenance and naturally prefer soils that are capable of supporting healthy populations of such organisms. Consequently, they seem more abundant in grassland habitats where there is high soil invertebrate biomass. Earthworm biomass concentrations, especially *Lumbricus terrestris,* are greater in pastures than wooded habitats, which partially explains why the former supports higher mole densities.

Coast Moles feed mostly on earthworms. Glendenning (1959) found a highly significant correlation between the density of Coast Moles and the density of earthworms in 157 fields in the lower Fraser Valley. He stated that the Coast Mole eats arthropods, annelids, and molluscans. He examined the stomach contents of 108 kill trapped Coast Moles and found no evidence of any vegetable matter (which, incidentally, is a commonly cited concern of gardeners/farmers). Earthworms were found in 93% of the stomach contents indicating the importance of these invertebrates in the Coast Mole's diet. Van Zyll de Jong (1983) reported that earthworms constitute more than 90% of the food consumed by the Coast Mole while small quantities of plant material (bulbs, grains, peas, potatoes) may be ingested. In fact, Glendenning (1959) estimated that one mole required 150 worms per day, roughly 1 every 10 minutes. This amount would equate to the consumption of an unbelievable number of 52,560 earthworms a year!

Reproduction

During their estimated life expectancy of 4 or 5 years, a female Coast Mole will have from 9 to 15 offspring (Glendenning 1959). Breeding takes place in late winter and the pups are born throughout the spring after a gestation of 4 to 6 weeks. The young are weaned during the summer months and establish their own home range shortly afterwards. They are capable of reproduction during their first year.

Predation

Natural enemies of the Coast Mole include: weasels (Silver 1933), great horned owls (Maser and Brodie 1966), barn owls (Giger 1965), red-tailed hawks (Silver 1933; Roest 1952), coyotes (Toweill and Anthony 1988) and rubber boas on nestlings (Maser *et al*. 1981). Domestic cats and dogs are probably the Coast Moles most significant predators considering the human population in the Greater Vancouver and Fraser Valley region.

Herons, crows, foxes and mink are known predators of the European Mole, *Talpa europa*, (Gorman and Stone 1990) and undoubtedly prey upon both *Neurotrichus* and *Scapanus* spp. The extent to which natural predators control mole populations in BC is presently unknown, however, it is likely insignificant considering the presence and density of Coast Mole mounds throughout the Lower Mainland.

2.3 Townsend's Mole *(Scapanus townsendii)* M-SCTO

Description

The Townsend's Mole is the largest mole in North America and is similar to the Coast Mole except for size (Figure 8). It is cylindrical in shape, varying in color from gray to black. Like the Coast Mole, its fur is velvety soft and unidirectional. The front feet are broad and shovel-like, equipped with 5 long flat claws. In contrast, the hind feet are relatively small with weaker claws. Tiny blue eyes are hidden beneath their facial fur and a layer of skin, which makes them difficult to see. Both their long, flexible snout and short tail are sparsely haired.

Figure 8. Townsend's Mole (Nagorsen 1996)

Distribution

Species Range

This mole is found throughout the coastal areas of California, Oregon, and Washington northwards into extreme southwestern BC (Nagorsen 1996).

Provincial Range

This mole occurs only in the vicinity of Huntingdon and Abbotsford in the Fraser Valley (Figure 9). Sheehan and Galindo-Leal (1996[b]) extensively sampled this region and surrounding areas and concluded that the range of Townsend's Mole in BC is less than 20 km^2.

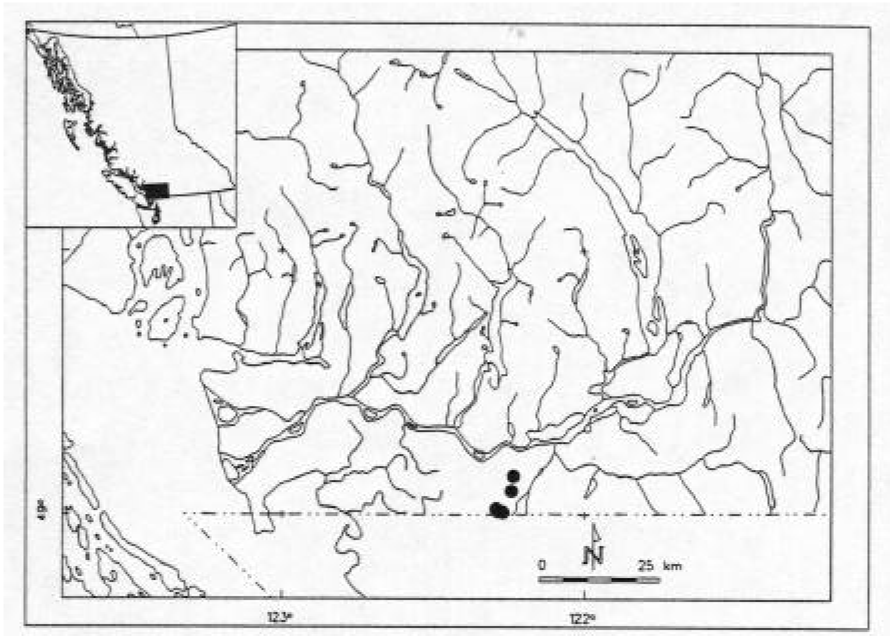

Figure 9. Townsend's Mole distribution in BC (Nagorsen 1996)

Density Patterns

Sheehan and Galindo-Leal (1996[b]) performed crude density estimates based upon mound construction in two fields near Huntingdon and reported densities of 2.1/ha and 5.3/ha. In Oregon, densities may reach 12/ha (Pedersen 1963; Giger 1973) or be as low as 0.4/ha in areas with few earthworms and unsuitable soils. It is unlikely that Townsend's Mole densities in BC would be as high as in Oregon considering it occurs at its northernmost range in the province.

Activity Patterns

Like the Coast Mole, the Townsend's Mole is active both night and day. Sheehan and Galindo-Leal (1997) live trapped this mole day and night and directly captured it during daylight hours. Fresh mounds can be found at any time during a 24 hr period. Active moles can produce upwards of 8 mounds per day; however, less than 3 is average (Sheehan and Galindo-Leal 1996[b]). Mound construction peaks during the period of juvenile dispersal in summer and during the wetter fall months when the soil softens.

Status

The Townsend's Mole appears on BC's Red List (Sheehan 1999) and COSEWIC has designated Townsend's Mole as *Threatened* (Sheehan and Galindo-Leal 1996[a]). This mole has a global ranking of G5 because it is not at risk in California, Oregon or Washington, but has a sub national rank of S1? because it is rare and localized in BC.

Habitat Requirements

The Townsend's Mole prefers damper habitats than the smaller Coast Mole, inhabiting the meadows on the flood plains of low elevation rivers and glacial outwash prairies. However, both Townsend's and Coast Moles do co-exist in some localities (Dalquest 1948) and they are sympatric throughout the larger mole range in BC. Townsend's Mole is known to occupy fir *(Abies)* forests in Washington, but its mounds and tunneling activities are reportedly less visible (Dalquest 1948). This is likely because the Townsend's Mole can compact the excavated dirt against the tunnel walls causing less mounding; as well as any mounds produced are concealed by litter on the forest floor. The deep loose soil of cultivated fields is favored by this mole which avoids areas where the gravel content of the soil is high (van Zyll de Jong 1983). However, Townsend's Mole inhabiting Sumas Mountain persist in rocky, relatively shallow Alderwood Silt Loam as compared to those in the Huntingdon area which enjoy deep, fertile Lynden Silt Loam. An isolated subspecies of Townsend's Mole, *Scapanus townsendii olympicus*, is found only in the rugged terrain of the Olympic peninsula in Washington State at elevations exceeding 1000 m (Carraway *et al.* 1993). This suggests that the Townsend's Mole may be more of a habitat generalist than a specialist. Furthermore, since logging, agriculture and urbanization have significantly altered the natural landscape where Townsend's Mole persists in BC, suitable habitat may include residential backyards, ditches, hayfields or forests.

Pedersen (1963) reported that pastures that received more fertilizer (solid cow manure) also had greater densities of moles because this type of fertilizer provided ideal conditions and habitat for earthworms. The effects of liquid manure on earthworm numbers and mole populations have not been investigated although mole activity appears less in fields where liquid manure is regularly applied (Sheehan, pers. comm. 2001)

Conspicuousness and Distinctiveness of Sign

Adult Townsend's Moles can be differentiated from Coast Moles by: 1) total length >180 mm; 2) hind foot >24 mm; 3) weight >90 g; and 4) condylobasal length of skull >37 mm (van Zyll de Jong 1983). However, sub-adult Townsend's Mole may be confused with the Coast Mole because of the obvious overlapping of measurements that occur during the larger moles first year of growth.

The tunnels and mounds constructed by the larger Townsend's Mole exceed those of the smaller Coast Mole. Sheehan and Galindo-Leal (1997) reported the average Townsend's Mole mound to be 44 cm in diameter, 17 cm in height and a vertical tunnel diameter of 5 cm. Overlap does exist between *Scapanus* mound width and height which leaves vertical tunnel diameter as the most reliable discriminating measure.

Feeding

Townsend's Moles are principally dependent upon soil invertebrates for their sustenance and naturally prefer soils that are capable of supporting healthy populations of these organisms. They seem more abundant in grassland habitats where there is high soil invertebrate biomass. Earthworm biomass concentrations, especially *Lumbricus terrestris,* are greater in pastures and hayfields than wooded habitats which partially explains why this mole is more abundant in these locations.

Townsend's Moles feed mainly on earthworms, but vegetable matter is also part of their diet. Pedersen (1963) analyzed 200 Townsend's Mole stomachs and found that the primary food source was the earthworm (72%). The remaining 28% were composed of roots. One stomach contained 100% slugs and six stomachs had traces of insect skeletons. Wight (1928) examined 306 Townsend's Mole stomachs and found that 97% contained enough vegetable matter to suggest that it was consumed deliberately. Moore (1933) concluded that tulip, tigridia, bulbous iris bulbs, wheat, oats, peas, vetch, corn, carrots, parsnips, and potatoes frequently form a significant proportion of the Townsend's and Coast Mole diet. In addition, he suggested that moles occasionally prefer a vegetarian diet to their natural food of earthworms, insects, and other invertebrates. He mentioned that the Townsend's Mole demonstrates an acquired taste for many cultivated plants even in the presence of its natural food supply. Other authors have confirmed the prevalence of earthworms in the mole's diet (Hall and Kelson 1959; van Zyll de Jong 1983; Forsyth 1985). In fact, Townsend's Mole can be kept healthy in captivity for weeks solely on a diet of earthworms.

Reproduction

The breeding season coincides with the winter months (Pedersen 1963) during which time the females construct shallow underground nests averaging 1.6 L in size (Kuhn *et al.* 1966). An average of 3 pups are delivered during the spring and in 5 to 6 weeks they exceed 115 mm in length and weigh between 60 and 80 g (Kuhn *et al.* 1966). Juveniles are capable of breeding during their first winter. The reproductive potential of this mole is likely similar to that of the Coast Mole, which limits it to 3-4 breeding seasons.

Predation

Natural enemies of the Townsend's Mole include: weasels (Silver 1933), great horned owls (Maser and Brodie 1966), barn owls (Giger 1965), red-tailed hawks (Silver 1933; Roest 1952) coyotes (Toweill and Anthony 1988), and rubber boas on nestlings (Maser *et al.* 1981). Herons, crows, foxes and mink are known predators of the European Mole, *Talpa europa,* (Gorman and Stone 1990) and possibly prey upon *Neurotrichus* and *Scapanus* spp. in BC. The extent to which these natural predators control mole populations is presently unknown.

Domestic cats and dogs are probably the most important predators of Townsend's Mole considering their restricted distribution and the human population they live amongst. Interestingly, dogs and cats do not eat the moles that they kill (Maser *et al.* 1981), possibly because of their unpleasant taste and smell (Glendenning 1959).

Benefits

Unfortunately, a mole's usefulness is directly related to how its existence benefits humans. Moles are credited with being beneficial to agriculture by improving the aeration and drainage of the soil, consuming harmful soil invertebrates and circulating soil minerals (Kuhn and Edge 1990). However, other authors believe that these advantages have been replaced by modern agricultural practices and that any benefits accruing to agriculture, due to mole activity, are now grossly outweighed by their damage (Glendenning 1959; Stone 1989; Gerber 1995). Regardless, moles are unique and benefit our concept of biodiversity with their interesting forms, behaviour and genetic composition.

In the Abbotsford area, a sizable percentage of the land that is now occupied by the Coast Mole and the historical sites where the Townsend's Mole specimens have been collected are in the Agricultural Land Reserve (ALR). Although this land is not at an immediate risk to urbanization, agriculture and industrial activities will continue to threaten the long term persistence of Townsend's Mole in this region. Unfortunately, the population of Townsend's Mole on Sumas Mountain, not in the ALR, is currently threatened by intensive urbanization.

2.4 Northern Pocket Gopher *(Thomomys talpoides)* M-THTA

Description

There are currently six subspecies of pocket gopher described in BC (Table 1). Nagorsen (1991) and Cowan and Guiguet (1973) briefly summarize the distribution of each subspecies. Cowan and Guiguet (1973) describes the Northern Pocket Gopher as follows: about the size of a rat (average of six subspecies is 187-212 mm), tail is about half the length of body, slender, scantily haired, broad head, small eyes, short ears hardly projecting above fur, large fur-lined pouch (for carrying food) extending back inside the cheek from an opening on each side of the mouth. The hair colour varies, but is generally brownish to gray/black with a paler underbelly (Bonar 1995). Its fore claws are strongly curved and sharp-pointed. Many of these morphological traits clearly distinguish this species from moles although many people regularly confuse moles and pocket gophers.

Table 1. Measurements of pocket gophers from different regions. Total length (TL), tail length (T), hind foot (HF) (in mm) and weight (W) (in grams).

Species	TL	T	HF	W	Reference
T. talpoides					
mean		N/A	13.8	m(104.4) f(91.4)	Andersen 1977
range	N/A	N/A	6.0 - 22		
T. t. cognatus					
mean	187	57	26.3	N/A	Cowan and Guiguet 1965
range	175-197	55-64	25-27	N/A	
T. t. fuscus					
mean	192	61.5	25.6		Cowan and Guiguet 1965
range	175-205	56-67	25-27		
T. t. incensus					
mean	204	71	26		Cowan and Guiguet 1965
range	184-277	56-82	24-27		
T. t. medius					
mean	m(195) f(196)	m(67) f(67)	m(25) f(25)	N/A	Cowan and Guiguet 1965
range	m(183-210) f(183-210)	m(60-82) f(61-76)	m(24-27) f(23-27)	N/A	
T. t. saturatus					
mean	m(212) f(203)	m(71) f(65.5)	m(28) f(27)	N/A	Cowan and Guiguet 1965
range	m(196-223) f(186-218)	m(61-79) f(58-73)	m(27-29) f(25-29)	N/A	
T. t. segregatus					
mean	m(208) f(199)	m(74) f(72)	m(27) f(26)	N/A	Cowan and Guiguet 1965
Range	m(193-220) f(190-210)	m(64-81) f(65-86)	m(24-29) f(23-29)	N/A	

For a detailed revision of Northern Pocket Gopher subspecies, *see* Johnstone (1954).

Pocket gophers have fine textured, soft, glossy hair, not really fur (Bonar 1995). Because of their low dispersal rate and subsequently limited gene flow, gopher populations adapt well to local conditions, and therefore exhibit little colour variation within a local population (Case 1983). Several studies have claimed that fur colouration is a result of the environment. Darker species occur in areas with darker soils and lighter species occur in areas with lighter soils (Teipner *et al*. 1983). Pocket gophers annually undergo variable and irregular molts. Semiannual molts result in distinctive summer and winter coats (Teipner *et al*. 1983). Furthermore, early summer molts often result in the presence of a “molt line” that spreads down to the tail.

Distribution and Habitat

Species Range

The Northern Pocket Gopher, *T. talpoides*, has the widest distribution of all pocket gophers, extending from central Alberta and southeastern British Columbia to northern New Mexico and Arizona, and from the western three-fourths of North and South Dakota to eastern Washington, Oregon, and northeastern California (Chase *et al*. 1982).

Provincial Range

In British Columbia, the Northern Pocket Gopher inhabits the southern dry interior, southern Kootenays, and southern Rocky Mountains (Nagorsen 1991). Nagorsen (1991) also gives brief summaries on each of the sub-species occurring in British Columbia.

Density Patterns

Home range size usually corresponds to the burrow system, and a single gopher will inhabit and actively defend individual territories (Teipner *et al*. 1983; Bonar 1995). Although rare, several authors have found that multiple occupancy of a single burrow system exists (e.g., Chase *et al*. 1982). Home ranges of males will typically exclude other males, but will include several females (Bonar 1995). Population densities fluctuate naturally depending on the quality and quantity of food available (Bonar 1995). Ground disturbing activities generally increase the carrying capacity of an area for Northern Pocket Gophers by increasing the supply of preferred foods and allowing a build up of the resident population (Bonar 1995). Gophers will reach their highest densities on friable, light-textured soils with high plant biomass, especially large, fleshy roots, tubers, bulbs, or other storage structures (Bonar 1995). Scrivner and Smith (1981) found that densities of gophers were highest in stands less than 10 years old and greater than 80 years old, compared with stands in the 11 – 79 year old age class. This was attributed to the conditions of the understory vegetation.

Burrow systems can be divided into two parts: shallow feeding burrows located 12-20 cm below ground, roughly 5 cm in diameter; and deep permanent nesting galleries situated at a much deeper depth of 1-3 m which often contain shredded grasses (Anderson and Kluge 1986). One pocket gopher may require 0.5 ha for its burrows (Godfrey 1987). In heavily infested areas, densities may approach 123 adults/ha (Howard and Childs 1959). Pocket gophers can construct more than 50 mounds per year (Anonymous 1984). Gophers will diligently repair any open break in their burrows at ground level by sealing them with an earthen plug. These plugs will also be used to seal off feeding tunnels, and although difficult to see, these plugs can be used to indicate gopher presence (Teipner *et al*. 1983). Tunnel systems can be fairly extensive. Over a five-month period, Richens (1966) followed a pocket gopher and recorded the construction of 146 meters of feeding tunnels. Deep tunnels often lead to nests and may attain depths of 1.5 meters or more (Teipner *et al.* 1983). In winter, gophers can burrow through the snowpack, with tunnels often reaching lengths of 30 meters (Teipner *et al.* 1983). Gophers form soil casts by pushing soil up from their burrow system into tunnels dug into the snow. These cylindrical casts settle into the ground during spring snowmelt, leaving evidence of winter activity (Teipner *et al.* 1983).

Activity Patterns

Pocket gophers are primarily fossorial and live in extensive burrow systems in which they are active throughout the year (Cowan and Guiget 1973; Anderson and Kluge 1986). Increased activity takes place in the fall when food is cached for the winter (Kreps 1909). Arid areas and soils with dense brush and forest-cover are avoided by this species. Where suitable soils occur, the pocket gopher is found from valley bottoms to alpine meadows (Cowan and Guiget 1973). In Alberta, *Thomomys* is associated with the sandy, lighter textured soils of the black soil zones where better quality soils and forage productivity occurs (Nietfeld and Roy 1993). Preference for this soil type may also be accounted for its ability to drain moisture away, allow air to diffuse through the closed burrow systems and the ease with which it can be dug through (Nietfeld and Roy 1993).

Status

The Northern Pocket Gopher subspecies *T. talpoides segregatus* is on the provincial Red List. Globally, this subspecies is listed as *critically imperiled* or *imperiled*

Conspicuousness and Distinctiveness of Sign

Although pocket gophers are often confused with moles they can be differentiated by the mounds they construct. Fresh *Thomomys* mounds are comprised of sifted dirt (indicating kicking) and generally fan-shaped while the dirt in mole mounds is cloddy (indicating pushing) and they are conical in shape. Another distinguishing characteristic of the pocket gopher is that they regularly fill their abandoned runways with dirt, a process rarely undertaken by *Scapanus* spp. (Moore 1939). Also, the existence of observable raised ridges, which result from *Scapanus* spp. surface tunneling are not usually produced by *Thomomys* (Anderson and Kluge 1986). Generally, the pocket gopher inhabits an area further east than either the Townsend's or Coast Mole, although its range may overlap that of the Coast Mole in the vicinity of Manning Park.

Feeding

Pocket gopher food preferences show marked geographical variation (Teipner *et al.* 1983). Pocket gophers are mostly root eaters and not insectivores like the moles. *Thomomys* generally remain underground; however, surface vegetation may be taken in large quantities during the growing season (Ward 1960). Their diet consists of the roots of many species of trees, grasses and flowering plants although legumes and broad-leaved forbs are preferred (Nietfeld and Roy 1993). This vegetation is sometimes stored in underground chambers (Keith *et al.* 1958; Ward 1960; Cowan and Guiget 1973).

Reproduction

Sexual development and fertility depend on nutrition, and gestation and lactation require higher than average energy requirements, an ample supply of nutritious food speeds sexual maturity and promotes large litters and increased survival (Bonar 1995). The breeding season for pocket gophers varies throughout their range and depends on the physical characteristics of the environment (Teipner *et al*. 1983). The major reproductive effort generally takes place in the spring, coinciding with periods of abundant green growth and optimum soils for burrowing (Teipner *et al.* 1983). On western range lands females produce one litter ranging in size from one to thirteen are born sometime in April or May after a gestation period of approximately 28 days (Cowan and Guiget 1973; Teipner *et al*. 1983; Bonar 1995). Approximately 5 to 8 weeks after birth, the young disperse as a result of increasing agonistic behaviour between the mother and her young (Teipner *et al*. 1983; Bonar 1995).

3. PROTOCOLS

When considering sampling designs and methods, the type of information desired, the amount of effort and cost, specific site characteristics and limitations must be considered in light of the objectives of the study and the urgency of the management decisions to be made. Often, several different methods may be needed to address different aspects of a study. As new information is obtained, new questions and hypotheses may be formed, the objectives redefined and the methods modified. Typically, there will be a tradeoff between the amount of time and money an investigator can devote to a particular site and the number of replicate sites that is possible to establish. It is important for researchers to report the details of their research so that the assumptions behind each study are clearly understood. A variety of survey techniques may need to be used to fulfill the study objectives, however, caution should be exercised when attempting to compare results among studies, study areas, or through time (RIC 1999).

Table 2. Recommended methods for inventory of moles and pocket gophers in British Columbia at the three levels of intensity.

Species	Presence/Not Detected	Relative Abundance	Absolute Abundance
Shrew-mole	Direct Observation	Direct Observation	Mark-Recapture
Coast Mole	Direct Observation	Direct Observation	Mark-Recapture
Townsend’s Mole	Direct Observation	Direct Observation	Mark-Recapture
Northern Pocket Gopher	Direct Observation	Direct Observation	Mark-Recapture

3.1 Sampling Standards

The following are guidelines for conducting inventory studies on moles and pocket gophers in the province. Close adherence to these guidelines will permit the collection of reliable data that should satisfy individual and corporate inventory needs, as well as contribute to biodiversity monitoring at local, regional, and provincial scales.

3.1.1 Preliminary Surveys

Researchers should always solicit landowners when searching for information on the presence of local species. Haeck (1969) was successful in appealing to the public for information pertaining to dead moles found on the roads in Holland. The collation of this data provided enough information to uncover the peak period of *Talpa europaea* juvenile dispersal in that country.

The period of juvenile Townsend's Mole dispersal in Oregon was obtained from information acquired from road kills (Pedersen 1963; Giger 1973). Although the effectiveness of this type of research is dependent upon the co-operation of usually unsympathetic landowners that consider moles pests, valuable knowledge can be the amassed. Existing information on the restricted distribution of Townsend's Mole could be refined with the use of road kill data. If undertaken, this information would also provide researchers with an understanding of the juvenile dispersal period, increase the public's awareness of this presently red-listed species and provide specimens for laboratory analyses and museums. If this approach is undertaken, it is important to provide specimen bags and instructions on how to collect and preserve mole carcasses to those willing to assist (see manual No. 4a, *Voucher Specimen Collection, Preparation, Identification and Storage Protocol: Animals* (RIC 1999b)).

3.1.2 Selecting Study Areas

The presence of molehills is the only criterion used by all researchers when selecting study areas. Schaefer (1978) extensively searched for farmland throughout the Lower Mainland with the largest number of molehills (qualitatively assessed) before selecting his 10 study sites to trap the Coast Mole. Only the area within each study site that contained the most molehills was then marked off for his research purposes. Pedersen (1963) explained that the bulk of his Townsend's Mole trapping took place within a five mile radius of Tillamook, Oregon, because the farms are centrally located where this species' habitat is abundant which would reduce travel time between study areas. Giger (1973) chose a 150 ha area hedged by mountains, river and ocean to study the movements and homing of the Townsend's Mole in Tillamook, Oregon. Sheehan and Galindo-Leal (1997) used voucher specimen data and information from professional mole trappers to begin their sampling of the Central Fraser Valley for Townsend's Mole. Therefore, there is no standard method followed when selecting a study area other than the search for signs of mole activity.

Unlike the *Scapanus* spp. whose mound construction indicates their presence, signs of Shrew-mole activity and presence are less obvious. Therefore, surveying efforts should be directed towards areas where research indicates there is an association between this species, other species, and habitat. Shrew-mole runways can be located by raking away the surface cover of dead litter to expose shallow troughs that are roughly one and one-half inches wide by three-quarters of an inch deep (Dalquest *et al.* 1942). Since such runways may be traveled regularly by the animal in its search for food, they may also provide an ideal site to place pitfalls or other traps.

It is advisable to select a study area that consists of a single ecological association which is relatively uniform (Blair 1940). A particular species that occurs in two or more ecological associations is likely to behave differently in each (Blair 1940). For example, Pedersen (1963) sampled moles in two pastures of identical soil type, the only difference was in the application of cow manure to one pasture. The manured plot contained a higher density of moles attributed to an association between soil fertility and earthworm abundance. This situation helps to illustrate why, if more than one habitat type is selected for trapping, the results from each type should be evaluated separately: even within a single association population densities may vary.

3.1.3 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 terrestrially-based wildlife, standard attributes from the terrestrial Ecosystem Field Form developed jointly by MELP and MOF (1998) will be used. Appendix E of the manual, *Species Inventory Fundamentals* (No.1), contains a generic discussion of habitat data collection as well as a list of standard attributes, which may be relevant to mole and pocket gopher surveys (RIC 1998).

3.1.4 Time of Year

Mound Count Surveys

During the autumn months, mounds are more conspicuous and the year's juveniles temporarily increase population numbers. Mole activity and mound formations are visibly reduced during periods of cold weather when the soil freezes. Mound activity increases as the ground softens which generally occurs after prolonged cold spells or heavy rainfall.

Owl Pellets

If this method is used, the collection of owl pellets should coincide with the period of aboveground juvenile mole dispersal. This period is estimated to occur somewhere between May and June for juvenile Townsend's Moles in Oregon (Giger 1965). BC researchers should consider the possible effects that the latitudinal gradient (affecting soil temperature and food availability) between Tillamook, Oregon and the Fraser Valley has on this period of dispersal. The breeding season of moles come later in the year as one moves north due to climatic conditions. For example, breeding periods for *Talpa europaea* vary from mid-February in central Italy to late June in north-east Scotland (Gorman and Stone 1990). *Scalopus aquaticus* exhibits a two month latitudinal variation in its breeding season between the 32nd and 45th N parallels which would also result in a different timing of juvenile dispersal (Eadie and Hamilton 1956).

Open-Burrow Surveys

Plan for late summer / early fall because the young-of-the-year pocket gophers have dispersed by this time (Bonar 1995).

Trapping

Mole activity and mound formations are visibly reduced during periods of cold weather when the soil freezes. In the Fraser Valley, the winter temperatures are rarely low enough to freeze the soil, but if the ground does freeze trapping should be temporarily ceased until it thaws.

Trap-outs have been used successfully to determine the absolute abundance of pocket gophers (Ingles *et al.* 1949; Howard and Childs 1959; Richens 1965; Reid 1973). The basis of the trap-out method is to capture all of the animals in a given unit to determine the size of an active pocket gopher population. Fall trapping will produce the most accurate numbers as the young-of-the-year will have attained trappable size by this time (Teipner *et al.* 1983). The advantages of this technique include count accuracy and rodent availability for immediate examination (Reid 1973; Teipner *et al.* 1983).

3.1.5 Handling

To avoid prolonging stress while handling study animals, take standard measurements as soon and quickly as possible. However, moles are difficult to handle because of their hyperactive nature. The use of the 'mole holder' speeds up this task by freeing up both hands of the researcher (Figure 10). Wearing gloves hampers the deft handling of moles although wearing them is advisable to avoid being bitten or scratched. Work on the mole or pocket gopher over a pail half filled with dirt so that if the animal falls, it will not get hurt and can be easily retrieved. A mole or pocket gopher dropped on the ground will burrow immediately and escape unless it is quickly unearthed with a shovel.

A clip, identical to those on the end of electrical test leads, can be fixed to a 20-inch L-shaped rod with light wire and attached to the tail of the mole. This frees both of the researcher's hands, allowing the vertical specimen to be measured relatively easy over a pail. Individual mole measurements require only a minute with this device and the specimens are all released unharmed. Temporary discomfort to the mole can not be denied. However, this device speeds up the process and eliminates the need to grasp the moles firmly to reduce their struggling, which may result in more permanent injuries.

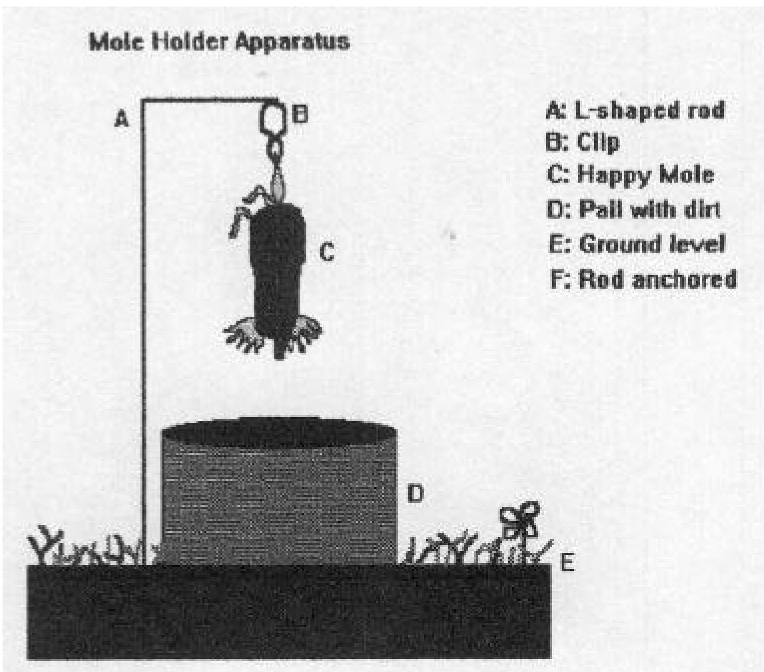

Figure 10. Mole holder

Handling live moles and pocket gophers for measuring purposes is difficult because they continually struggle to escape. Another option is to use ether, which has been used successfully to anesthetize Townsend's Mole (Pedersen 1963). Moles are placed into a can with a piece of cotton saturated with ether and then sealed with a tight lid. The moles are subdued in three to four minutes and then can be removed from the container. In this way, weighing, measuring, and the collection of ectoparisites is much easier. Moles can be returned to their tunnel after 10 minutes and when later recaptured have no visible ill effects.

The Coast Mole appears to be mild mannered. When it bites it feels like a harmless pinch. However, the larger Townsend's Mole can be tenacious when removed from a trap for measuring and its more powerful bite can be detected through leather gloves. This animal's bite would certainly sting, and possibly break, bare skin. Both *Scapanus* spp. occasionally emit a loud high-pitched 'Eeek! Eeek! Eeek!' when handled which appears more as a protest to being handled than reflecting physical pain.

Following data collection return the mole to a five-gallon pail that contains a few centimeters of soil and provide five to six earthworms to help restore its energy prior to release. When the worms have been ingested (10-15 minutes) release the mole into the study plot. If the decision is made to keep the mole for further observation, half-fill the pail with soil and add another five worms to satiate its appetite during transport. Do not put more than one mole in a pail at one time because they will fight violently.

Retained Coast and Townsend's Moles can survive in small terrariums (10 by 20 inches) for short periods and can exist solely on a diet of earthworms. The amount of worms allotted to each mole should be no less than 2/3 of their body weight. However, because they can consume over two dozen earthworms/day (18 worms cost $3.85), moles are costly to keep in captivity. The only way to avoid this cost is to have access to an area where at least one dozen earthworms can be dug up daily for each mole in captivity. The subterranean environment that the moles have adapted to is relatively stable and every attempt should be made to duplicate it. It is important to ensure that the moles temporary lodgings are not exposed to extreme temperatures, direct sunlight or precipitation. The soil should be replaced in the terrariums at least weekly and secure, ventilated lids will keep your mole from escaping.

If long-term captivity is required several authors have provided suggestions and more detailed information (Reed 1944; Glendenning 1959; Gorman and Stone 1990; Campbell and Hochachka 2000).

Hantavirus is transmitted to humans from persistently infected rodents and other small mammals (National Research Council 1991). Multiple species may serve as hosts within a particular area and the strain of virus and its likelihood of causing disease in humans varies from region to region (National Research Council 1991). Although no research can be found that implicates either moles or pocket gophers as hantavirus hosts, the fact that mole tunnels are often communal, used by mice and voles, suggests a potential risk to workers handling fossorial mammals. Deer mice *Peromyscus maniculatus* have been identified as hantavirus hosts in BC and have transmitted the virus to humans. Direct animal contact is not necessary as the virus is spread to humans primarily through the aerosols of infected urine, feces, or saliva (National Research Council 1991). Wearing a mask when excavating the moles and pocket gophers dirty and dusty lateral shafts and tunnels may help prevent the field worker from inhaling these potentially deadly aerosols. The virus can also be transmitted to humans from the bite of an infected animal (National Research Council 1991). Researchers should take precautions and wear gloves when handling moles and pocket gophers to help avert any potential health problems associated with bites from these animals.

3.1.6 Identification of Moles

Distinguishing *Scapanus townsendii* from *Scapanus orarius*

Scapanus townsendii can be distinguished from *Scapanus orarius* by the following measurements [Hartman and Yates (1985) and Carraway *et al.* (1993) figures provided first followed by van Zyll de Jong (1983) in parentheses]: total length larger than 200 mm (> 175 mm), length of hind foot larger than 24 mm and the condylobasal length of the skull is greater than 40 mm (>37 mm).

Distinguishing *Scapanus townsendii* from *Neurotrichus g. gibbsii*

Differentiating the Townsend's Mole from the Shrew-mole is much easier as the former possesses 44 teeth (i3/3, cl/1, p4/4, m3/3 dentition) while the latter has 36 (i3/3,cl/1, p2/2, m4/4 dentition). *S. t. townsendii* has forefeet as wide as long, a total length larger than 130 mm, it's tail is smaller then 25% of total length and it's hind feet are larger than 18 mm (Carraway *et al.* 1993). Also, unlike either of the two *Scapanus* spp. in the province, *Neurotrichus* possesses forefeet that are only moderately adapted to digging. This species can therefore climb small bushes and move with speed and agility above ground (Carraway and Verts 1991). Both *Scapanus* species have powerful forefeet that are permanently oriented outwards to facilitate their lateral-thrust type of digging activity (Hartman and Yates 1985).

Table 3. Standard measurements of moles in British Columbia.

Total length (TL), tail length (T), and hind foot (HF) in mm and weight (W) in g.

Species	TL	T	HF	W	Reference
N. g. gibbsii					
mean	105.4	34.5	15.1	10	van Zyll de Jong 1983
range	98-116	30-40	14-17	9.5-10.5	
mean	113.6	37.1	16.6	10	Carraway & Verts 1991
max	125	42.0	17.5		
mean	112	37		11.1	Nagorsen 1996
range	98-125	29-50		8.0-14.5	
S. o. schefferi					
mean	162	33		62.8	Nagorsen 1996
range	145-181	28-41		45.6-78.0	
mean	162.5	33.	21.7	m(74.3) f(69.8)	van Zyll de Jong 1983
range	154-173	28-37	20-24	m(64-91) f(61-79) m(65.3) f(57.2)	Schaefer 1978
mean	168.7	34.7	23		Hartman & Yates 1985
range	165- 171	34-35	23		
S. t. townsendii					
mean	203	38		116.8	Nagorsen 1996
range	156-237	29-45		106.0-126.5	
mean	203.3	36.2	26.7	m(l47) f(117)	van Zyll de Jong 1983
range	179-237	33-41	25-29		
mean	m(214) f(203)			m(l41.7) f(119)	Pedersen 1963
range	195-237	34-51	24-28		Carraway *et al.* 1993

3.1.7 Sexing Moles

Research on the European Mole indicates that sexing this species can be accomplished by careful external examination (Gorman and Stone 1990). Measuring the distance from the anus to the urinary papilla has revealed that this measurement is always greater in the male (Gorman and Stone 1990). This method has not been validated with *Scapanus* spp., but it would provide an excellent measure for researchers attempting to sex live trapped moles in the field.

Body weight can be used by researchers to sex moles because females are generally lighter than males (Pedersen 1963; Gorman and Stone 1990). Pedersen (1963) recorded the standard mammalian measurements for 300 Townsend's Moles from Oregon and found only slight differences in the external measurements. However, body weights were discovered to reveal the greatest difference between the sexes; males averaged 141.65 grams and females 119.03 grams (Pedersen 1963). Unfortunately, considerable overlap exists between these average weights, but Townsend's Moles that weigh more than 140 grams are probably male. Glendenning (1959) also reported that male Coast Moles were heavier than females although significant overlap with this species also exists. A Coast Mole that weighs more than 79 grams is likely a male (Glendenning 1959); however, pregnant females can exceed this weight (Sheehan, pers. comm. 2001).

Moles examined during the breeding season may be more easily sexed because the males are likely to have enlarged testes and the vaginas in females become perforated and visible. This procedure is difficult and should only be performed by experienced researchers to ensure accuracy. Also, moles captured during the spring should be examined for signs of lactation.

3.1.8 Ageing Moles

Glendenning (1959) trapped 940 Coast Moles in Aggasiz, BC from November to April between 1935 and 1945. Based on weight categories, fur condition (no criteria provided) and the number of embryos in gravid females he classified moles into four age classes: young of the year (40-60 g), one year old adults (65-70 grams), two year old adults (71-75 g) and aged adults (>75 g). A constant 45% of the specimens were adults older than one year and 6 % were over three years old. This methodology is problematic and its usefulness can only help to identify Coast Moles less than one year old.

Schaefer (1978) developed a key based upon tooth wear for aging the 23 Coast Moles he kill trapped in Aldergrove, BC. Tooth wear is influenced by habitat and therefore the ages derived from this method should be regarded only as estimates. Schaefer (1978) estimated that 18% were one year old, 52% were two years old, 26% were three years old and 4% were four years old.

The most accurate assessment of a mole's age can be derived from histological sections made through jaw and teeth samples. In these sections the dense annual growth lines can be counted with the use of a microscope (Gorman and Stone 1990). Obviously, this requires that animals be killed which is inappropriate for any species at risk. Thus, weighing appears to be the quickest method for roughly ageing Coast Moles, but it lacks the accuracy possible from histological sections. At present there is no ageing criteria for either the Shrew-mole or Townsend's Mole.

3.1.9 Marking Moles

Mole research requiring repeat captures (*e.g.,* growth rates, dispersal distances) will require the implementation of some type of system whereby individual moles can be marked and subsequently identified. Some researchers have found toe-clipping in Townsend’s Mole to be satisfactory and reported no detrimental side-effects (Pedersen 1963; Kuhn *et al.* 1966). Giger (1973) found that size 5 bands (16.0 by 3.5 by 0.5 mm) fastened to the proximal end of a mole's tail resulted in necrosis and tail loss in 7 of 40 (18 %) recaptured Townsend’s Moles marked this way. Monel butt-end bands (National Band and Tag Company, Newport, Kentucky) of size 6 (19.0 by 3.5 by 0.5 mm) that were attached above the hind foot were found to be the most effective means of marking adult and sub-adult Townsend’s Mole (Giger 1973).

The use of Monel butt-end bands appears to be the best available method for marking Townsend’s Mole. This method should be investigated with different sized bands, on both the Shrew-Mole and Coast Mole. Researchers will have to experiment to establish an effective methodology for marking moles. Perhaps tattooing or permanent marker applied to the backside of the forefeet may also work for recapture studies.

3.1.10 Mole Trapping

Live Traps

1) Victor-based live trap

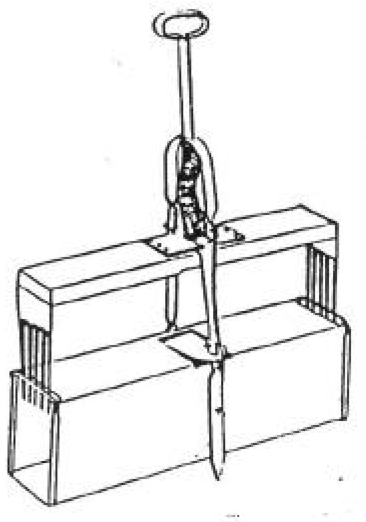

Figure 11. Victor-based live trap

After a straight section of tunnel has been identified dig a rectangular plot 35 by 20 cm (cut the top section of sod in one piece so that it can be replaced when the trap is pulled) keeping the approximate location of the mole tunnel in the center. When the sod has been lifted off (about 10 cm) carefully scrape the ends of the plot with a hand trowel and locate the mole tunnels before continuing to dig. It is extremely important that the trap be packed firmly with dirt below the inside trigger paddle (making a dirt ball with the hands and placing it under the trigger paddle works). This will ensure that the moles tunneling will force the trigger paddle to ‘spring’ the trap. The ends of the trap are then packed with dirt to the middle section. A piece of rebar or dowel (< 2 cm) can be reamed into both ends of the trap up to, but not through, the harder-packed center to create a tunnel. Center the ends of the trap with the exposed mole tunnels in the plot and set the trap in place. Ensure the ends of the trap are sealed to the mole tunnels with dirt so that no light can enter as this will repel the moles. Mark the trap site with high visibility flagging. Traps should be checked every 12 hours to ensure captured moles do not starve. Baits are not necessary with this trap.

Sheehan and Galindo-Leal (1996[b]) reported an efficiency rate of 14% with this trap. Giger (1973) experienced an efficiency rate of 31% using a similar, but better designed trap. Although these traps are cumbersome and can take up to 30 minutes to set properly they can catch both *Scapanus* spp. Considering the central Fraser Valley has been extensively sampled, it is doubtful that their use for future distribution research would be cost-effective (Sheehan and Galindo-Leal 1996[b]).

2) Pipe Trap

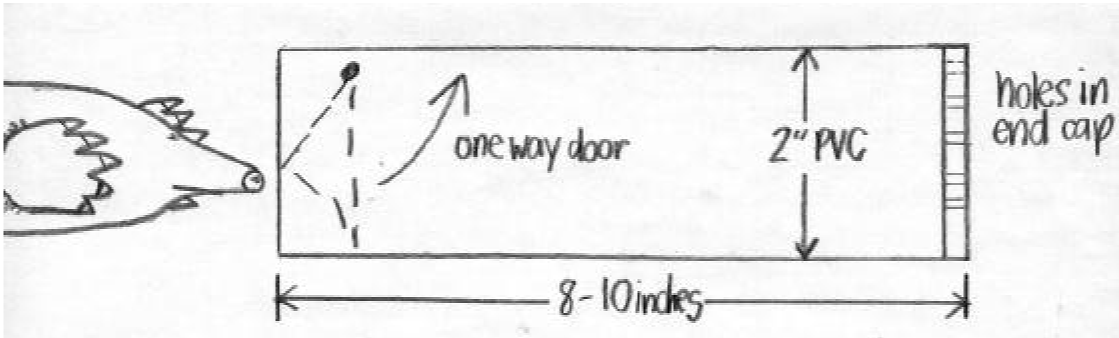

Figure 12. Pipe trap

The pipe trap can be constructed quickly and cheaply out of PVC or ABS (Figure 12). The dimensions can be modified and those provided are suitable for the Coast Mole. This trap has been used for decades to capture the Coast Mole in BC.

The pipe trap is best suited for shallow tunnels. These are identified as raised ridges and will require two traps per site. Find a relatively straight section and cut into the ground exposing the two tunnel openings. Cover each tunnel opening with a pipe trap making sure that the interface is sealed with dirt so that the only light that enters the trap is through the ventilated end cap. Mark the trap site with high visibility flagging and check the traps at least every 12 hours. Set up time is less than 10 minutes for 2 traps.

3) Direct Capture

The most commonly cited method of capturing moles alive is with a shovel (Glendenning 1959; Kuhn *et al*. 1966; Giger 1973; Schaefer 1978; Gorman and Stone 1990; Sheehan and Galindo-Leal 1997). This method is referred to as 'direct capture' (Figure 13).

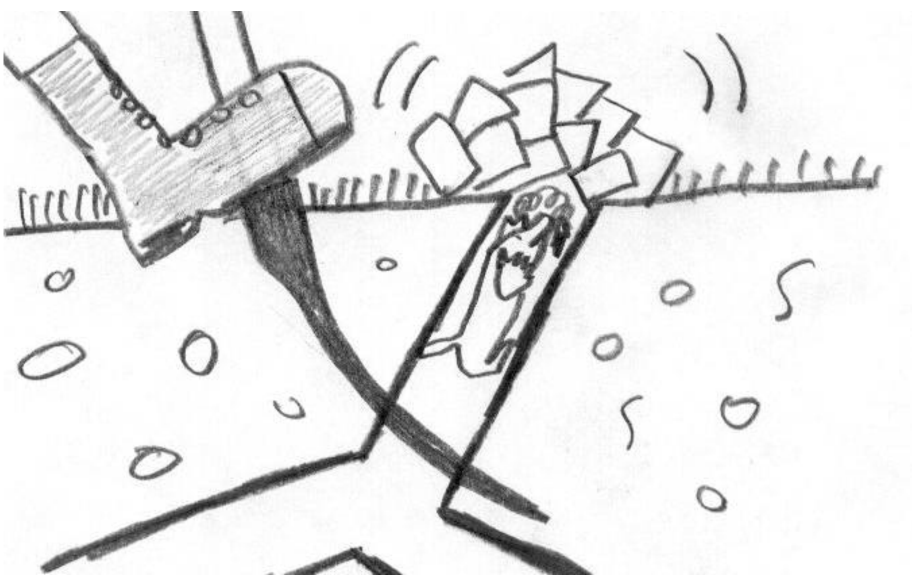

Figure 13. Direct capture technique.

The method requires that a person approach the active mound slowly and quietly, to avoid detection by the mole. If you feel your movements have alerted the mole and mound construction ceases, do not leave, but be patient and remain motionless by the mound with your shovel ready. The mole will often continue to work after a brief pause of less than five minutes.

The difficulty with this approach is estimating the location of the mole and thus where to plunge the shovel. Moles can be pushing dirt up into the mound from either inside their tunnels or just below the surface in the lateral shaft. Observations of mound building indicate that the mole forces the sheared soil up into the mound in a series of distinct, successive pushes which brings both the voided earth and the mole slowly closer to the surface. Each period of activity is followed by a brief pause when it is assumed that the mole goes back down the tunnel to collect more sheared soil from tunnel construction for expulsion. Waiting until the mole nears the surface before plunging the shovel is safer and more effective than attempting to dig out a mole deep within its tunnel.

The best opportunity to safely unearth the mole comes when it is engaged in the last push of a series, which reveals the mole is close to the surface. Sometimes it is possible to detect from which direction the mole is expelling the dirt by observing the angle and position of the voided plug with respect to the mound. It is more effective if the mole is dug up from the side it approaches the mound because the shovel will sever the tunnel it is occupying thereby eliminating an escape route. When the time is right, quickly force the shovel into the ground with your foot, ensuring enough room is left for the shovel to clear the retreating mole. It is important to immediately pop out the shovel-full of sod from the ground because the mole may appear in the excavation and will speedily burrow to safety if not grabbed immediately.

Direct capture may be effective, but it is also a potentially dangerous live capture method because the timing is so critical. If the shovel is misdirected into the ground the mole could be sheared in two, crippled, or mortally wounded. However, if the shovel is plunged into the ground too slowly the mole will have time to retreat into its tunnel and escape. Sheehan and Galindo-Leal (1997) directly captured eight Coast Moles and seven Townsend’s Moles during nine months of fieldwork. They estimated the efficiency rate of this method to be 30%, which is, double that of the live trapping efficiency. Researchers should be familiar with mound construction by viewing it in progress for a period of time prior to attempting direct capture. This method should not be attempted by inexperienced biologists, especially if a species at risk is concerned.

4) Pitfalls

Small mammals that characteristically travel in burrows and that orient primarily by non-visual senses can be trapped with pitfall traps (Williams and Braun 1983). Shrew-moles have been successfully captured in pitfalls in BC (Seip and Savard 1992; Kremsater *et al.* 1993; Campbell and Hochachka 2000) although their effectiveness on *Scapanus* is negligible (Sheehan and Galindo-Leal 1995).

Tin can pitfall traps 1.4 L in size buried in the forest floor every 15-m apart along transects or grids will be effective. Traps should be checked at least every four hours to minimize starvation, predation and freezing. Pitfalls should be closed if frost or rainy weather is expected. Shrew-mole trap mortality can be high and is usually caused by starvation.

3.1.11 Pocket Gopher Trapping

Live Traps

A variety of live traps designed to capture pocket gophers *(Thomomys)* have been described in the literature (Scheffer 1934; Ingles 1950; Howard 1952; Sargeant 1966; Hart 1973).

The most important variable to consider when choosing which trap to construct is the soil condition present at the proposed research sites. Other factors such as season, weather, animal size, animal residency status, and odours of previous animals captured in the trap will effect the efficiency of the trap (Allen *et al.* 1997). If the land is sandy it is advisable to select the Hart's trap because it was designed specifically for use in this type of soil. Hart's successfully captured *Thomomys talpoides*, the species of gopher that does exist in BC. In addition, Hart (1973) stated that his trap captured both *Geomys* and *Thomomys* genera in all types of soil.

Trapping sites composed of stable soils are less likely to require specifically designed traps. The Hart trap is recommended when working with strict budget constraints, as it is cheaper to construct than the Bolten trap. Also, it has been field tested on *Thomomys talpoides* and it is effective in a wide variety of soil conditions (Hart 1973). When budget is not a concern, the Bolten trap should be used, as it has been field tested in BC, and is considered a versatile, successful trap (Fraker pers. comm. 1998). It is recommended that when possible the Bolten trap be favoured over the Hart trap due to the success of the Bolten trap in BC.

1) Bolten Trap

Formally known as the Longworth live trap, this trap has been used extensively for the capture of small mammals in BC. This trap has several advantages over other traps and is considered to be the trap of choice for the live capture of pocket gophers (Fraker, pers. comm. 1998). The Bolten trap has several features that make it appealing. First, it has a door that can be locked open when not trapping. This is particularly useful if you are trying to desensitize an animal to the trap. Second, the trap comes apart into two pieces, the nest box and the tunnel, therefore the trap is easily maintained. And third, the trap is small, easily transported, and relatively easy to operate. The only drawback with the Bolten trap is its significant cost. At around $50.00 per trap, these traps may not be suitable for those working with a limited budget. However, these traps have been proven to work in BC, and have been very successful when used to capture pocket gophers (Fraker pers. comm. 1998).

Generally, Bolten traps are used in pairs, with the opening of each trap facing opposite directions. This increases the capture probability and eliminates potential trap avoidance behaviour. The traps should be covered with soil and cotton batten to prevent any light from entering the tunnel system. When trapping within the tunnel system of a pocket gopher, it is advisable to disturb the tunnel as little as possible. Placement of the traps should occur such that the trap is angled slightly downward, allowing for complete door closure.

2) Hart Trap

Hart (1973) reported from field-testing that his trap captured over 200 gophers (trap nights not revealed) and resulted in only one injury and one death (causes not disclosed). This trap successfully captured *Geomys bursarius* throughout its range in all types of soil, even those approaching pure sand, as well as *Thomomys talpoides* (Hart 1973). Although no figures are provided, this trap is also claimed to be as effective as the Macabee kill trap when set under optimal conditions (Hart 1973). In 1973, the cost of materials for one trap was estimated at $0.40. Traps can be constructed in 45 minutes to one hour (Hart 1973). Despite Hart's (1973) confidence in his trap design he suggested adjustments to the trap diameter, length and sensitivity of the trigger wire, the placement of supporting loops, and the tension and suspension of the door.

This trap should be placed into an excavated cavity adjacent to the main burrow at an angle. It is important not to break into the main burrow, which requires careful digging down the lateral shaft to ensure that the trap can be positioned horizontally at the junction of the lateral shaft and the main tunnel (Hart 1973). A natural tunnel floor can be simulated in the trap by gently twisting and pushing the trap into the enlarged excavation which will push soil up into the bottom of the trap (Hart 1973). The trap is then carefully covered, not filled with dirt. If the soil is sandy, a piece of sod placed on the trap will help prevent excess dirt from entering the trap (Hart 1973). Do not cover the top perforation in the tin can with dirt. Leave it open to allow light and air into the trap to entice the gopher to enter and trip the trap with its plugging reflex.

Although Hart (1973) was successful with the use of one trap per site, with its door opening flush with the main burrow, he suggested that two traps facing in opposite directions in the main tunnel might also be effective. Baits are not necessary with this trap.

3.2 Inventory Surveys

The table below outlines the type of surveys that are used for inventorying moles and pocket gophers for the various survey intensities. These survey methods have been recommended by wildlife biologists and approved by the Resources Inventory Committee.

Table 4. Types of inventory surveys, the data forms needed, and the level of intensity of the survey.

Survey Type	Forms Needed	*Intensity
All	• Wildlife Inventory Project Description Form • Wildlife Inventory Survey Description Form-General	• PN • RA • AA
Field data forms for methods recommended in this manual are still under development.		

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

3.3 Presence/Not Detected

Recommended method: Sign Sampling for moles and pocket gophers.

3.3.1 Sign Sampling

The presence of molehills is the only criterion used by all researchers when selecting study areas. Areas with active moles are easily identifiable by fresh molehills. Mounds are more conspicuous during the autumn months when the year's juveniles temporarily increase population numbers. Mole activity and mound formations are visibly reduced during periods of cold weather when the soil freezes.

Until recently, sympatric *Scapanus* spp. could not be differentiated without trapping. A less invasive approach using mound and tunnel measurements after Sheehan and Galindo-Leal (1997) is recommended as a way to differentiate Townsend's Mole from the Coast Mole. Pocket gopher mounds can be differentiated from mole mounds by shape and appearance (see section 2.4, heading Conspicuousness and Distinctiveness of Sign).

The collection and investigation of the remains in owl pellets provides researchers with another method of gathering data on the distribution of both *Scapanus* spp. and *Neurotrichus* in BC. One regurgitated barn owl pellet usually represents one meal (Giger 1965) which provides physical evidence of prey species when analysed.

Giger (1965) found that although *Scapanus* spp. were preyed upon by the barn owl, very few are consumed. Also, Pedersen (1963) stated that neither dogs or cats were observed consuming the moles that they killed. This fact should encourage researchers to seek the assistance of landowners considering the less that are eaten the more specimens there should be available. Campbell (1983) examined over 11,000 barn owl pellets, some coming from the 13 km^2 area surrounding Huntingdon believed to be the range of the Townsend's Mole in Canada. He found the remains of 317 Coast Moles and only one Townsend's Mole. Campbell (1983) concluded that *Scapanus* spp. accounted for less than 0.5% of the barn owl's diet. Kremsater *et al.* (1991) examined 976 barn owl pellets from the Fraser Valley, 303 from the Townsend's Mole range. They found 29 Coast Moles and no Townsend's Mole remains.

Owl pellets have also been examined for the Shrew-mole (Cowan 1942; Campbell 1983; Kremsater and Kremsater 1993). Similar to *Scapanus* spp., the Shrew-mole made-up less than 0.5% of the barn owl's diet in BC (Campbell 1983). Cowan (1942) attributed the low number of Shrew-mole remains in the barn owl pellets he examined (one in 300 pellets) to differences in the habitat hunted by the barn owl (open meadowlands) and the mainly wooded habitat preferred by Shrew-moles. Kremsater *et al*. (1993) collected 976 barn owl pellets, from Delta, Surrey, Langley, and the Abbotsford area, and discovered the remains of six Shrew-moles. Interestingly, all six remains were recovered from Langley and all but one from a single site. This suggests that although Shrew-moles comprise a very small percentage of the average barn owl's diet, some owls may predate upon this species more than others. Several authors have reported the percentage that shrew moles represent in the diet of different predators: Screech Owl, 5% (Dalquest and Orcutt 1942), Great Horned Owl, 2.3%, and Barn Owl, 0.98% (Maser and Brodie 1966). These percentages indicate Shrew-moles are a more significant food source for owls than either the Coast or Townsend's Mole. However, Kremsater *et al.* (1993) believe that live trapping *N. gibbsii* offers more potential than Barn Owl pellet analysis in acquiring information on this species.

Mound-oriented and owl-pellet methods may be used independently or combined when conducting and animal sign survey.

Office Procedures

- Review the introductory manual No. 1 *Species Inventory Fundamentals.*
- Obtain maps for Project and Study Area(s). Any map, which is used to record data, should be referenced to NAD83.
- Outline the Project Area on a map. Due to the limited distributions of moles and gophers, the scale of this map will probably be no smaller than 1:50,000.
- As appropriate, determine Biogeoclimatic zones and subzones, Ecoregion, Ecosection, and Broad Ecosystem Units for the Project Area from maps.
- Delineate Study Areas within this Project Area using the presence of molehills as the criterion for selecting study areas. Study Areas will generally be fairly small (no more than a few hectares) for this species and as “closed” as possible to movement over the study area boundary.
- 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.
- Draft a letter to landowners whose properties are to be surveyed. This letter should outline who will be working in the area, what they will be doing, the objectives of the research, and the dates and duration that the property would be visited if access is granted.

***Scapanus* differentiation based on mound and tunnel characteristics**

More data are desirable to support the use of mound and tunnel characteristics as a tool for differentiating the presence of one *Scapanus* species from another. The protocol below provides a simple method to further test this method before using it to determine species present in a new area where either or both species may occur.

- Choose study areas where mole sign is present. Study areas should possess boundaries over which movement will be minimized (e.g. roadways). Ensure that study areas are small enough that they can be thoroughly searched, or if larger study areas are desired, these should be stratified to ensure that “likely” habitats are thoroughly searched. Include control areas that contain only the Coast Mole considering this mole is extremely abundant and widespread (e.g. Surrey, Langley and Vancouver).
- Completely search each study area. Transects which cover the entire study area can ensure that it is searched thoroughly. Roughly map and number each encampment in the delineated study area.
- Randomly select a number of encampments to sample within each study area. If there are only a few encampments, sample them all.
- For each encampment sampled, roughly map and number the mounds.
- Randomly select mounds for measurement (preferably 3-5 **fresh** mounds per encampment).
- Measure and describe attributes of each randomly selected mound (as described in Field procedures below).

- Perform an Analysis of Variance based on these measurements to investigate significant differences in mound measurements and possible occurrence of Townsend's mole, characterized by significantly larger mounds and tunnel diameter, in a study area. See *Data Analysis*.
- Perform additional Townsend's Mole live trapping in areas known to support this species (e.g. Farmer and MacKenzie Road west of Huntingdon and Ledgeview Golf Course area on Sumas Mountain). This is necessary to create a robust Townsend's Mole mound and tunnel measurement sample size.

Sampling Design

- There is no standard sampling design to determine presence other than concentrating in areas that have a high density of activity and sign for the target species. Search appropriate habitat areas within the restricted ranges of each species.
- If the survey is attempting to evaluate the presence of mole fauna in an area where sympatric mole species may occur, it is recommended that the sampling design follow the approach described above (*Species differentiation based on mole mound and tunnel characteristics*).
- Owl pellets: As above, collect any pellets that are found.

Sampling Effort

- The amount of effort and time required for a survey will depend largely on the size and the number of study areas.
- If more than one study area is to be sampled, sampling efforts should remain constant if a valid comparison of species richness between areas is to be made.

Personnel

- Field personnel should be familiar with identifying mole remains or signs, especially the differences between *Scapanus* spp. mound and tunnel characteristics.
- At least one person should be familiar with the collection of habitat data.

Equipment

- Maps
- Gloves
- Hand trowel
- Wire flags (orange)
- Probe (rebar)
- Field book with data forms and pen
- Indelible ink marker (for flagging)
- Golf balls (4.3 cm diameter)
- Calipers (tunnel diameters, mole measurements)
- Tape measure (mound diameters, heights)
- Bendable ruler
- Weight scale (up to 200 grams)
- Specimen tags/bags
- Field guide to mammals
- Binoculars
- 5 gallon pails (carry gear) and long handled shovels

Field Procedures - Mounds

- It is best if fresh mounds can be found, as this supports the conclusion that moles or gophers presently occupy the study area. This approach works well if the study area is a large pasture or hayfield that could be chain harrowed with a tractor and then observed for signs of fresh activity. Also, a freshly cut hayfield would provide an excellent site because the machinery would have leveled most molehills, flattened numerous surface runs and cleared the field to permit the observation of the latest diggings. However, the window of opportunity is limited to the growth rate of the grasses. Therefore, researchers should be in regular contact with farmers during the haying season to take advantage of this opportunity.
- Draw a sketch map of the sample area that indicates directions to the site, outline of the study area, prominent features within the site, and the location of transect lines (if used) so that other investigators may relocate the appropriate site for future sampling.
- Follow either a systematic or random sampling design (see section Sampling design above).
- Record any mole or pocket gopher observations or sign detections on the appropriate dataforms.
- Determine whether a detected mound belongs to a pocket gopher or a mole:
    - *Thomomys* mounds are constructed of finely sifted dirt and are generally fan-shaped while mole mounds are cloddy and conical in appearance (see Figure 7).
    - Pocket gophers regularly fill their abandoned runways with dirt, a process rarely undertaken by *Scapanus* spp. (Moore 1939).
    - The existence of observable raised ridges which result from *Scapanus* spp. surface tunneling are not usually produced by *Thomomys* (Anderson and Kluge 1986).
- For each selected mole mound record the following attributes (see Figure 6):
    - Mound width = greatest diameter at the base of the molehill (measuring tape)
    - Mound height = distance from the ground to the apex of the mound (ruler)
    - Perform the ‘Golf Ball’ test to determine whether the tunnel is large enough for a golf ball (~ 4.3 cm diameter) to fit into. Expose tunnels by digging through mound until the vertical shaft reaches the permanent horizontal tunnels. Dig the tunnels squarely back about 10 cm to ensure a consistent measure before inserting the golf ball. Drilling through the middle of the golf ball and pulling a string through and tying it off on either side will eliminate losing the ball down a tunnel.
    - Vertical tunnel diameter = taken approximately from 10 cm from either side of the fork where the vertical shaft originates (use calipers).
- Determine from mound and tunnel measurements which mole species are present (see data analysis calculations).
    - Base decisions on your own calculations from earlier field testing (see section, “Species differentiation based on mole mound and tunnel characteristics”) and/or
    - on the results of studies by Sheehan and Galindo-Leal (1997) in southwestern BC. They determined that encampments containing mounds, which exceed 15 cm in height and 40 cm in width with shallow tunnel diameters greater than 4.5 cm (approximately the size of a golf ball) strongly indicate the presence of Townsend’s Mole.
- If it is not clear from mound and tunnel measurements which *Scapanus* spp. is present live-capture may be necessary.

Field Procedures - Owl pellets

- Draw a sketch map of the sample area that indicates directions to the site, outline of the study area, prominent features within the site, and the location of transect lines (if used) so that other investigators may relocate the appropriate site for future sampling.
- Transects may be used to ensure complete coverage of the study area. If this is not feasible, it may be most efficient to include portions of the study area where perching and roosting trees for raptors are present.
- Record any mole observations or sign detections on the appropriate dataforms.
- Collect any owl pellets found.
- Examine pellets (or have them examined if you are not skilled at identifying prey remains) to determine prey items, specifically whether any moles were consumed.

Data Analysis

- Compile lists of the sampled species indicating which were present at each study area. It is important to note that not detecting a species does not necessarily indicate that it is absent from the study area. However, the greater the sampling effort, the more confidence one has about which species are present and which are absent.
- Summarize attributes of measured mounds, if appropriate, by study area.
- If appropriate, perform Analysis of Variance by study area and by encampment (if sample size permits – as discussed in *Species differentiation based on mole mound and tunnel characteristics*). This may provide a valuable clue as to where Townsend’s Mole, which is characterized by significantly larger mounds than Coast Mole, may occur. Beware of the effects of “diluting” measurements from Townsend’s Mole mounds with those from Coast Mole mounds in study areas where the two species’ distributions overlap. This will result in less discernable differences between mound measurements collected from study areas occupied exclusively by Coast Moles, and those that also contain Townsend’s Mole.

3.4 Relative Abundance

Recommended method: Sign Sampling for moles; and Ground Counts of Mounds and Earth Plugs and/or Aerial Photographic Surveys for pocket gophers.

3.4.1 Sign Sampling

Whereas section 3.3.1 contains protocols for sampling mounds to determine presence, it is necessary to sample new mound activity to evaluate relative abundance of fossorial mammals. Many of the procedures for sampling mounds will be the same as those described in section 3.3.1.

Mound Activity

At present, there is no accurate means of calculating the number of moles from the number of mounds in an area. However, indirect indices of mole abundance can be acquired by physically destroying existing mole activity and recording the subsequent mound and surface tunnel production. This approach would be ideally suited to a large pasture or hayfield that could be chain harrowed with a tractor and then observed for signs of fresh mole activity. Concentrations of new mole mounds when observed on a daily basis and isolated from others (20-30 m) would indicate distinct mole encampments. Daily flagging each area of new activity will provide the researcher with a rough estimate of mole numbers, in less than a week, for a particular study plot.

Sampling Design

- Search in areas that have a high density of mole activity and signs.
- Unlike methods described for *Presence/Not Detected*, it is very important to attempt to keep bias constant between the different units sampled when evaluating relative abundance.

Sampling Effort

- Sampling efforts must remain constant if a valid comparison is to be made among study areas, or over time at a study area.
- Depending on the density of the target species, surveyors may choose to standardize their results by unit effort (e.g. mounds per linear distance of transect searched) or per square units (e.g. meters) of study area searched.

Data Analysis

- Be explicit in written discussion that mound activity values do not equate to actual animals.
- Comparisons of relative abundance among sites is only possible if biases (e.g. effort, detectability) can be maintained at a constant level when sampling within each of the study sites. Surveyors should be clear in their written discussion as to where unequal biases may lead to possibly erroneous results.
- To determine relative abundance, summarize the data in terms of observations per unit effort, *i.e.*, area or time.

3.4.2 Ground or Aerial Photography to Count Sign

Ground Counts of Mounds & Earth Plugs and/or Aerial Photographic Surveys for pocket gopher sign counts.

Relative abundance has been shown to only be useful if it can exhibit a reasonably consistent relationship with actual abundance over time and/or space. Teipner *et al.* (1983) describe several techniques which may be used when determining relative abundance of pocket gophers. Richens (1965) found that a positive correlation existed between gopher density and fresh gopher mounds counted bimonthly in a given area; he did not find a positive correlation when counting in 72 hour intervals. Conversely, Reid *et al.* (1966) found that a positive correlation between gopher densities and mound counts done in 48-hour intervals did exist. They combined mound counts with a session of kill trapping immediately following a count session. In this way they were able to determine the number of active pocket gophers present in a given area. Teipner *et al.* stated that ground counts and aerial photography were the most favourable methods to use as they provided the highest level of accuracy. Table 4 lists seven techniques that have been used to determine relative abundance of pocket gophers.

Table 5. Population estimation techniques that have been used to obtain relative abundance of pocket gophers (After Teipner *et al.* 1983).

Technique	Timing/Method	Recommended	References
Surface Sign **Surface sign and kill trapping**	• Bimonthly throughout summer • Visually count mounds and earth plugs • Visual sign every 48 hours. Trapping: best results in late summer, early fall	• Yes • With caution • (invasive)	• Richen 1965 • Reid *et al*. 1966
Winter Soil Casts	• Winter, after snowmelt	• With caution	• Richens 1965; Reid *et al.* 1966; Reid 1973; Reid 1981.
Open Hole	• Spring/Summer/Fall • Open marked burrows, then check the number of plugged holes 24 hours later for indications of activity	• Yes • (inclement weather can result in conservative estimates)	• Miller and Howard 1951; Miller 1953; Richens 1968; Barnes *et al*. 1970; Hungerford 1976; Birch 1978.
Aerial Photography	• Whenever mounds are visible • Photograph mounds from air and count from photo	• Yes	• Driscoll and Watson 1974.
Nearest Neighbor	• Seasonally	• With caution	• Hansen and Remmenga 1961

Bonar (1995) also describes three types of pocket gopher surveys that can be used to establish a relative abundance:

Visual Reconnaissance

This method is used to determine the presence of pocket gophers and determine the relative density of gopher activity. Recently created mounds and winter casts are used to estimate a rough percentage of a particular area that has an active gopher population associated with it (Bonar 1995). These surveys can be conducted using a predetermined survey design such as circular or linear plots selected systematically or randomly in such a way that the area in question is sufficiently sampled.

Mound-Count Surveys

These will provide an approximate population density and are easy to do. Using a plot survey that samples 1 to 5 percent of the area, one is able to ascertain the locations of concentrated gopher activity. This will also enable one to approximate the number of gophers per unit area as well as the range of gopher activity over the entire area. Because earth plugs do not affect the reliability of only counting mounds, they can be ignored in any tally of gopher sign (Bonar 1995). Population indices based on mound-counts alone are more reliable than those based on winter soil casts alone. Reid (1973) reports that winter soil casts may have utility in estimating relative abundance in early summer before the young disperse.

Mound-count surveys can be done quickly and efficiently using linear transects selected randomly or systematically throughout the area in question. One possible survey design may incorporates 2m linear transects selected randomly from a random numbers table such that each transect spans the width and/or length of the sample plot. The sample area may be a fraction of the desired area or may very well include the entire sample area. It is suggested that linear transects be used as they are easy to set up and can be repeated readily. To increase the randomness of the survey, one may choose to select different compass bearings and/or transect lengths from a random numbers table.

Open-Burrow Surveys

The open-burrow method gives a more accurate relative abundance method than does the mound-count survey. This is in part due to the pocket gopher’s solitary nature and to their tendency to plug any hole in their burrow system. To obtain an estimate, you expose a main runway in a representative sample of the burrow systems in a unit, and check the holes within 48 hours to see if the gophers closed them. If a hole remains open, the burrow is presumed to be unoccupied. The same procedure that is used for mound-count surveys can be applied here, with the addition of a probing device, which can be used to create the holes in active burrows.

Bonar (1995) found that the most effective method to determine relative abundance of pocket gophers was (in decreasing order): Open-burrow surveys, Mound-count surveys, and Visual reconnaissance. Therefore it is the recommendation of this manual that if the most precise relative abundance is required, then the Open-burrow systems be used when ever possible to determine relative abundance. When determining presence/not detected, Visual reconnaissance can be used quite successfully, perhaps in combination with Mound-count surveys. It is also possible to use a combination of ground counts and/or aerial photography, as it is non-invasive, efficient, and economical. Furthermore, surveys should be planned for late summer / early fall because the young-of-the-year have dispersed by this time (Bonar 1995).

3.5 Absolute Abundance

Recommended method: Mark-recapture sampling.

Live trapping should be performed only if sufficient precautions have been taken to minimize the risk of death to the captured animals (see below, Field Procedures).

Prior to inventorying study species, researchers must first decide the size of the study area(s) they require to accomplish the objectives of their research. The next step is to identify locations where there is animal activity and to assess the suitability between this area and your research goals (see section 3.1.2). Locating study sites to live trap the Shrew-mole and Coast Mole in BC is relatively easy because of their widespread distribution as compared to Townsend's Mole. Researchers undertaking the live capture of Townsend's Mole should look to previous fieldwork, voucher specimens and professional mole trappers for assistance in delineating a study area.

Information pertaining to the absolute abundance of a species in different habitat types is useful for the development of wildlife management policies. Unfortunately, acquiring this knowledge for moles is difficult because they are rarely observed; however, several techniques have been developed to assist researchers in determining the population levels of moles and pocket gophers.

Although the sampling methods described below focus primarily on moles they can be applied to pocket gophers, provided the sample size is adjusted accordingly.

3.5.1 Mark-recapture Sampling

Absolute abundance of a population can be determined by utilizing a mark-recapture methodology. Captured animals can be marked and then released. Despite the disturbances this method will cause to the local population it would be considerably less than that caused by kill trapping the population. Mark-recapture sampling requires significantly more labour to execute and the success rate in recapturing the wary, elusive mole is probably low despite some past success. Townsend's Moles were recaptured using Victor-based live traps and one was re-captured a total of eight times (Giger 1973). Attempts to establish absolute abundance figures for *Scapanus* spp. in BC by live trapping would be costly and extremely difficult considering low capture rates.

There are several concerns to incorporate into a researcher's mark-recapture study design, such as: length of trapping schedule, time of year, biased trapping of males and varying trapping skills amongst personnel must be addressed when live trapping. Weather conditions, differences in the distances between traps, and the varying ability of certain traps to capture moles are variables that will also influence population estimates when trapping.

There are numerous variations on the MRR method. However, all have common assumptions that must be met, or approximated, in order for subsequent data analysis and abundance estimates to be valid. These assumptions are:

1. Demographic and geographic closure (*i.e.*, The sample population is not significantly altered by births, deaths, immigration or emigration during the time of sampling).

2. All members of a population must have an equal or known probability of being captured (*i.e.,* any sample should be representative of the population being sampled).

Only in rare situations are both of these assumptions ever met; however, it is important that continued efforts be made to approximate them, if accurate estimates of population size are to be attained. When trapping *Scapanus* researchers should be aware that during the breeding season males will leave their encampments in search of receptive females and that males are more frequently live trapped (Pedersen 1963). Also, females with pups to nurture do not venture far from their nests. It is possible to minimize the effects of violating assumptions by modifying the general approaches discussed below. It can be useful to review some modifications to the basic formulas in literature such as Eberhardt (1969), Cormack (1972), O'Farrell *et al.* (1977), Pollock (1982), Krebs and Boonstra (1984), Nichols *et al.* (1984), Kenneth and Anderson (1985), Wilson and Anderson (1985), and Chao (1988). However, the computer program CAPTURE integrates all of these formulae into one comprehensive package, and researchers may find it easier to consult documents which deal with the suite of models used in CAPTURE, such as Otis *et al*. (1978), White *et al*. (1982), or Rexstat and Burnham (1991). See also the introductory manual, *Species Inventory Fundamentals, No. 1.*

Office Procedures

- Review the introductory manual No. 1 *Species Inventory Fundamentals.*
- 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). Any map, which is used to record data, should be referenced to NAD83.
- Outline the Project Area on a small to large scale map (1:250,000 – 1:20,000).
- Determine Biogeoclimatic zones and subzones, Ecoregion, Ecosection, and Broad Ecosystem Units for the Project Area from maps.
- Delineate one to many Study Areas 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.
- Based on map interpretation, identify transects and/or grids for census.
- Draft a letter to landowners whose properties are to be surveyed. This letter should mention: who the researchers are, what species is under study, the objectives of the research, dates and duration that property is be accessed.
- Acquire necessary permits for trapping.

Sampling Design

- Sampling designs for mark-recapture will vary with the objectives of a study. Traps are generally arranged systematically along straight or meandering transect lines or in a grid fashion. Length of transacts, dimensions of grids, and distance between trap stations will vary upon topography and home range size of the inventory species.

Time of Day

- Late afternoon to early morning may be the most humane time to live trap moles or pocket gophers during hot weather. During the hot summer months exposed traps should be closed during the day.

Sampling Effort

- It is difficult to recommend appropriate sample sizes (number of animals sampled) for the different trapping techniques described in the 'sampling standards' section. In general, the larger the sample size, the more precise the abundance estimate will be. Trapping should be performed consistently, *i.e.,* trap sessions should be scheduled uniformly among the different study areas.

Personnel

- Field personnel should be familiar with identifying mole or pocket gopher signs and the differences between *Scapanus* spp. mound and tunnel measurements.
- At least one person should be familiar with proper trap setting techniques.
- At least one person should be familiar with the collection of habitat data.

Equipment

- Trapping license(s)
- Maps
- Traps (ensure enough are available and that they are in working condition)
- Bait
- Shovel
- Gloves
- Hand trowel
- Wire flags (orange)
- Probe (rebar)
- Mole Holder
- Field book with data forms and pen
- Indelible ink marker (for flagging)
- Calipers (tunnel diameters, mole measurements)
- Tape measure (mound diameters, heights)
- Five-gallon pails (2)
- Earthworms
- Weight scale (up to 200 grams)
- Bendable ruler
- Specimen tags/bags
- Field guide to mammals
- Binoculars

Field Procedures

Place traps at predetermined capture (trap) stations.

Plot size for small mammals is generally 10-20 times the expected size of the species average home range, or no less than 2 ha (Blair 1940). Schaefer (1978) tracked one Coast Mole and calculated its home range to be 39 m by 39 m. Giger (1973) calculated the mean of the greatest distance between points of capture for 14 Townsend's Moles of both sexes, captured three or more times, to be roughly 41 m. If we accept these figures as workable home range values for each species and multiply them by 15 (average of 10-20) then a minimum plot size would be between 2.3 and 2.5 ha for trapping the Coast Mole and the Townsend's Mole respectively. These are only approximations and should only be used as a guide.

Once a study site has been selected it can be marked off into a 1 ha study plot that will allow the researcher to uniformly subdivide the plot (*e.g.,* 10 by 10 m). Schaefer (1978) subdivided his study plot so that he could make comparisons between molehill and earthworm numbers, within each study site.

There is no information pertaining to the Shrew-mole's home range (van Zyll de Jong 1983), but their population density has been estimated to be 12 to 15 animals/ha (Dalquest and Orcutt 1942).

Grids and transects provide a systematic approach to trapping. They allow the researcher to compare results from different trap sites. Terry (1981) successfully trapped Shrew-moles in Washington by setting up parallel traplines in 10 seral forest stages with internally homogenous vegetation. The parallel traplines were 12 m apart and each individual trapline was comprised of 10 stations, at 6 m intervals, containing two Victor mouse traps baited with a mixture of peanut butter, bacon grease and rolled oats.

Paired (Calhoun) traplines were also used by Anthony *et al.* (1987) to investigate the presence of small mammals in riparian zones (1 m from the stream and 15-25 m from the stream) in young, mature, and old-growth coniferous forests. However, only one Shrew-mole was captured after 900 trap nights. Each trapline consisted of 25 trap sites and positioned every 15 m were two museum special traps and one Victor rat trap that were baited with rolled oats and peanut butter.

Gashwiler (1959) used small-mammal grids to investigate mouse populations in different forest habitats that could be also be used when trapping Shrew-moles. Seip and Savard (1992) established grids of pitfalls and other traps throughout the north shore mountains of the Fraser Valley and after 13,722 pitfall trap nights, 48 Shrew-moles were captured.

Hooven and Black (1976) established permanent grids for live-trapping small mammals in western Oregon to investigate the effects clearcutting practices had upon their populations. Each grid was comprised of 10 rows that were spaced at 20 m intervals with two Sherman-type metal live traps of 22-gauge steel (9 by 9 by 25 cm) set at each station. These researchers sheltered each trap with bark and limbs to protect the captured animals from the elements. The traps were baited with a mixture of rolled oats, Douglas-fir seed, commercial bird seed and opened for three consecutive days per month from April to October 1968-1970. Three years of trapping (31,500 trap nights calculated) resulted in the 'infrequent' capture of 26 Shrew-moles and four Coast Moles (Hooven and Black 1976). Unlike the Shrew-mole, Coast Moles were captured in all three plots; the undisturbed forest, the clearcut forest, and the burned clearcut plot (Hooven and Black 1976). These results suggest that the Sherman metal live trap may be an effective trap for Coast Moles and Shrew-moles since it successfully captured both species.

Setting & Checking traps

- Please see ‘Sampling Standards’ section for details on trap types and trap placement. Ensure that enough traps are available and that they are in good working condition.
- During the hot summer months exposed traps should be closed during the day unless personnel are available to check them regularly. Late afternoon to early morning may be the most humane time to live trap moles or pocket gophers during hot weather. If daytime trapping must be undertaken, attempts should be made to shelter the traps from direct sunlight.
- When trapping in cold weather the traps should be baited and supplied with nesting material and checked at least every 12 hours. A caged mole will die in 12 hours if left in a live trap without food (Glendenning 1959; Pedersen 1963; Schaefer 1978).

Mark & measure unmarked animals

- See ‘Sampling Standards’ section for details on species’ measurement and marking.

Data Analysis - General

There are numerous ways of analyzing data from a mark-recapture study. Absolute abundance is determined by means of various estimators. The following is a short list of some useful texts that can be consulted for specific details for mathematical formulas:

1. Begon (1979). Investigating animal abundance: capture-recapture for biologists.
2. Davis (1982). CRC handbook of census methods for terrestrial vertebrates.
3. Krebs (1989). Ecological methodology.
4. Schemnitz (1980). Wildlife management techniques manual.
5. Seber (1973). The estimation of animal abundance and related parameters.
6. White *et al.* (1982). Capture-recapture and removal methods for sampling closed populations.

If a density estimate is required then at least four sessions should be conducted in a brief time period (to minimize violations of the assumption of closure). Many small mammal studies trap for four or five successive nights to get density estimates. The effect of trapping on animals should be considered when deciding the interval between trapping sessions; it will be less disruptive to the local population to allow for breaks in a trapping schedule. Data from this design can be used with program CAPTURE (see White *et al*. 1982).

As an alternative, the Jolly-Seber model will allow calculation of survival estimates, as well as a population size; however, biologists should be aware that this is not a true measure of density even though it provides a good measure for comparison over time or among areas. Sampling sessions can be conducted with a longer duration between trapping periods (*i.e.*, a few sessions each month). Many analysis options exist for the Jolly-Seber model; these are discussed in *Species Inventory Fundamentals, No. 1.*

Both of the designs above can be combined to allow density and survival estimates. This design is called "Pollock's robust design" and is also discussed in *Species Inventory Fundamentals, No.1*.

For systematic sampling, one trap per capture (trap) station is usually sufficient. Within dense populations, however, 2 or 3 traps per capture station may be necessary to avoid competition for traps.

Data Analysis - Sampling Effort

It is difficult to recommend sample sizes (numbers of observations or animals sampled) which are appropriate for every situation, as this will depend on the level of precision needed. Of course, the larger the sample size is, the more precise the abundance estimates will be. In general, sample sizes can be increased by increasing sampling effort. However, there are always limits to the amount of sampling effort that can be afforded during a study. The solution then becomes a compromise between available resources and levels of precision required to meet the objectives of the study. The simulation modules provided in CAPTURE and NOREMARK can be very helpful in determining the sampling effort needed to get adequate estimates. In addition, Pollock *et al.* (1990) provide sample size tables for the Jolly-Seber model.

The "robust" study design of Pollock (1990) is recommended if density estimates, and survival, and other demographic rates are an objective of inventory efforts. With this design, a series of five-day samples are conducted at equal intervals (*i.e.*, every month) during the time period of interest. The data from the five-day sessions is used to estimate density using program CAPTURE. (See section on data analysis, and *Species Inventory Fundamentals, No. 1*, Appendix G for more details on program CAPTURE.) In addition, these data are pooled and used with the Jolly-Seber model to estimate survival and other demographic parameters (using JOLLY or JOLLYAGE). This design has the following advantages:

1. Theoretically robust estimates of population size and survival are possible

2. Temporary emigration from the study area can be estimated from the data set allowing for further demographic inference, and less biased survival estimates if a subset of the population is not available for capture in a given trapping period. A new program, RDSURVIV, has been designed for this purpose when the robust design is used (see *Species Inventory Fundamentals, No. 1*, Appendix G).

3. The data should also allow further demographic inference and model fitting of survival rates using programs MARK, SURGE, and POPAN (see *Species Inventory Fundamentals, No. 1*, Appendix G).

Methods are available for biologists to determine appropriate sample sizes for the various mark-recapture estimators. It is recommended that project biologists consult the following sources for sample size calculations (Table 3).

Table 6. Sources for sample size calculation

Estimator	Source for optimal sample size calculation:
Lincoln-Peterson estimator.	Krebs (1989 page 22)
Jolly Seber estimates	Pollock (1990, page 72) Simulation: POPAN (Arnanson and Schwarz, 1987)
CAPTURE	White *et al.* (1985) Simulation: CAPTURE

The above references include graphs, and discussions of needed sample sizes for estimators. The determination of optimal sample sizes for program CAPTURE is complex. An easy to use simulation module is available as part of the program CAPTURE to allow biologists to explore sample size issues.

Data Analysis - MRR

The project biologist should be familiar with the different methods of data analysis for mark-recapture inventories before data collection begins. Different assumptions and requirements of the various models will have great bearing on sample design, effort and overall approach.

Below is a cursory discussion of mark-recapture models. This is included to provide biologists with an overview; however, a greater depth of knowledge will be required to actually carry out a mark-recapture inventory. Prior to commencing, it will be necessary to consult *Species Inventory Fundamentals No. 1* as this manual provides descriptions of many techniques, which are generic to species inventory. In addition, the following is a short list of some useful texts and articles. For complete citations see Literature Cited.

1. White *et al.* 1982. In some opinions, this is by far the most readable reference on mark-recapture that is available. Available at:
2. http://www.cnr.colostate.edu/~gwhite/software.html
3. Buckland *et al.* 1993. Good text for distance and transect sampling.

4. Krebs 1989, (also 1998, 2nd Edition). Good all round discussion of study design, but Chapter 2, Estimating abundance: Mark-and-Recapture techniques, is especially appropriate).
5. Pollock *et al*. 1990. A good discussion of the Jolly-Seber model.
6. White and Garrot 1990. A good discussion of study design for radio-telemetry estimation studies.
7. White 1996. A good discussion of mark-resight estimation procedure.
8. Schemnitz 1980. *Wildlife management techniques manual* (especially chapter 14 - Estimating the numbers of wildlife populations. pp 221-246).

There are numerous ways of analyzing data from a MRR program. The level of confidence placed in any estimator is largely dependent upon sample size, sample effort and how well the assumptions of the analysis methods are met in the field. Some common methods of analyses found in the literature are summarized below to provide some background information. Many sophisticated and robust methods of analyzing MRR data are available as part of the programs CAPTURE, JOLLY and MARK; all of these are discussed in *Species Inventory Fundamentals*.

Minimum-Number-Alive Estimator (MNA)

One of the easiest ways of estimating the abundance of a population from a mark-release-recapture (MRR) program is called the minimum number alive method (MNA). MNA (also called the calendar count or enumeration) is an estimate based on the sum of all individuals known to be alive during a particular capture (trapping) session. An individual is known to be alive during a given capture session if it was captured during that session, or if it was captured before and after that capture session. For example, if an individual is captured during capture session #1 and #3, it can be accurately stated that it was missed (but alive) during session #2.

Although the MNA method is simple to use, this estimator has been criticized as being negatively biased in most situations. For this reason, in a summary, Ritchie and Sullivan (1989) suggest that the MNA estimate should only be used when the trappability of animals is >70%. Several articles have been written on the use of the MNA estimator (Hilborn *et al*. 1976; Jolly and Dickson 1983; Nichols and Pollock 1983; Boonstra 1985; Efford 1992; Hilborn and Krebs 1992). Most of these papers recommend the use of the Jolly-Seber estimator over MNA if trappability is low or unknown.

This approach which is also referred to as “saturation trapping” or “enumeration” is generally not the best means of achieving a statistically valid estimate, and is not recommended. The reasons for this are:

- In many cases, a large amount of effort is needed to fully trap a population. In contrast, a valid estimate of population can be gained with less effort by using a ratio estimator, or a closed CAPTURE mark-recapture estimator.
- The assumption that an entire population is trapped or marked cannot be validated and therefore population estimates can be negatively biased (Pollock *et al*. 1990).
- To get an unbiased estimate of density, a population should be geographically closed. To minimize violation of this assumption, sampling should occur in a relatively small amount of time. Saturation trapping usually takes long periods of time, and therefore closure assumptions will be violated unless the researcher is working on an entirely closed system, such as an island.

Estimation by Asymptotic Capture

Population abundance can be estimated by intensively trapping and marking a population until no new (unmarked) individuals are captured. This method is essentially a modified (*i.e.*, non-lethal) version of kill trapping where animals are removed until no animals remain. It is generally not recommended as it is subject to criticisms similar to those described above.

Ratio estimators

The Lincoln, Petersen, and Schnabel estimators are based on the ratio of marked to unmarked individuals within a population. These estimators assume that the population is "closed" to immigration and emigration. The formulas are based on the assumption that the population size is related to the number of marked and released animals in the same way that the total caught at a subsequent time is related to the number recaptured (Davis and Winstead 1980). White *et al*. (1982) offer excellent discussion of closed models which many can be calculated using the program CAPTURE.

The Petersen (or Lincoln-Petersen) estimate is the most basic MRR method. It is based on two sample periods only (*i.e.*, one period of marking animals, followed by a single period of recapture). It is described using the following formulas:

$$\frac{N}{M} = \frac{C}{R} \qquad (1)$$

therefore:

$$N = \frac{CM}{R} \qquad (2)$$

where:

N = Population Estimate

M = Number of marked and released animals

C = Total number of animals captured

R = Number of marked animals that were recaptured

Lincoln-Peterson estimates are easy to calculate, and the estimator has been shown to be robust to time variation in capture probabilities. However, there are important assumptions associated with this estimator such as equal probabilities of capture between animals, population closure, and no net loss of animal marks between samples. If relative abundance is the objective then violations of assumptions may not be as significant provided that the degree to which assumptions are violated is similar between studies and over time, and therefore the estimator will show a consistent, comparable bias. If absolute abundance is the objective of methods, and animals can be marked individually then the use of the estimators in program CAPTURE is recommended.

Numerous variations on the Petersen Estimate have been developed. The Petersen Estimate is biased in that it tends to overestimate the actual population, especially if the sample is small. In response to this bias, Seber (1982) offers a variation on Petersen's formula that is less biased, and nearly unbiased if there are at least seven recaptures of marked animals. Another variation, the Schnabel estimate was developed to allow investigators to analyze data from multiple (>2) marking sessions.

The Jolly-Seber Estimator

Like the Lincoln, Petersen, and Schnabel estimators (above), the Jolly-Seber estimator is also based on the ratio of marked to unmarked individuals within a population. However, the Jolly-Seber estimate differs from others in that it recognizes, and attempts to incorporate, the fact that biological populations are generally not “closed”. This “open” model will not provide a true estimate of density, but rather of abundance, as the population is not defined in terms of area. This estimator requires that *at least three* sampling periods be carried out in order to calculate certain variables. Pollock *et al.* provide good discussion of Jolly-Seber models, and the program JOLLY is very useful for simulating MRR or analyzing data.

The formula for the Jolly-Seber estimate of population size is given below.

$$N_t = \frac{M_t}{\alpha_t} \qquad (3)$$

where:

N_t = Population estimate just before sample t

t = Sample period (1,2,3,4,5,......t th sample)

α_t = proportion of animals marked

$$\alpha_t = \frac{m_t + 1}{n_t + 1} \qquad (4)$$

m_t = Number of marked animals that were recaptured during sample t

n_t = Total number of animals captured during sample t

M_t = Estimated number of marked animals just before sample t

$$M_t = \frac{(s_t + 1)Z_t}{R_t + 1} + m_t \qquad (5)$$

s_t = Number of animals released

s_t = (n_t - accidental deaths)

R_t = Number of animals released during sample t, or s_t that were recaptured during a later sampling period

Z_t = Number of animals that were not captured during sample t, but were captured before and after sample t

The Jolly Seber model is also susceptible to biases if unequal capture probabilities are exhibited in the trapped population; however, the survival rate estimate of the Jolly Seber is robust to most forms of capture probability variation, and is therefore a useful alternative for monitoring populations. In addition, there are many modifications to the Jolly-Seber to accommodate age-specific capture probabilities and survival rates (program JOLLY JOLLYAGE and POPAN). If the robust design is used then program RDSURVIV can be used to estimate temporary emigration, and allow more precise survival estimates. Also, the Jolly Seber approach to survival modeling has been modified to allow the testing of biological hypothesis using various model fitting procedures as documented in programs SURGE, and MARK. However, many of the programs mentioned (with the exception of JOLLY and JOLLYAGE) require advanced statistical knowledge, and project biologists are urged to seek the advice of a qualified biometrician. A summary of useful software is available in *Species Inventory Fundamentals, No. 1*, Appendix G.

3.5.2 Removal Trapping

Although this sampling method is viable, kill trapping is not a recommended method for studying red-listed species such as Townsend's Mole or the Northern Pocket Gopher. It is also generally not recommended due to the possible by-catch of other species at risk. The following review is provided for interested readers.

Trap-outs have been used successfully to determine the absolute abundance of pocket gophers (Ingles *et al.* 1949; Howard and Childs 1959; Richens 1965; Reid 1973). The basis of the trap-out method is to capture all of the animals in a given unit to determine the size of an active pocket gopher population. The advantages of this technique include count accuracy and rodent availability for immediate examination (Reid 1973; Teipner *et al.* 1983).

It is possible to conduct a mole or pocket gopher census by trapping out and counting all of the individuals within a certain delineated region. Pedersen (1963) used this technique and trapped out two pastures (6.07 ha and 8.1 ha) in Oregon to calculate the population density of Townsend’s Mole which he calculated to be 4.8 and 11.8 moles per hectare. Schaefer (1978) kill-trapped 23 Coast Moles from a field in Aldergrove with the English scissor trap and calculated their density to be 6.36 moles per hectare. Glendenning (1959) calculated a density of 2.4 Coast Moles per ha after trapping out 157 moles from a 65 ha field in Agassiz.

This approach assumes that all of the moles are trapped to the same degree and that all of the animals within the delineated area were indeed trapped. However, unless this method is carried out for a sufficient period of time there is no validity to these assumptions. Trapping effectiveness varies between animals of the same species. For example, male European moles are more frequently captured than females, especially during the breeding season (Stone 1989). Similarly, Pedersen (1963) trapped 300 Townsend’s Moles over two years and found that males were captured more regularly than females, by a 2 to 1 ratio

It is important to consider the social organization of a particular mole species when estimating population density from trapping since the number of captures expected at each site will vary if the species is solitary or social, territorial or not (Stone 1989). Giger (1973) found the Townsend’s Mole to be solitary and no overlapping movements of adults in established burrows were discovered except during the mating season. However, Dalquest and Orcutt's (1942) trapping results indicate that the Shrew-mole is possibly gregarious as 11 animals were found in a set of traps positioned along a 50 foot log, at one check. During the course of a mole census it is difficult to ensure that no moles leave the study site or enter from adjacent areas.

Operator control, the way the traps are handled and set, varies between researchers and is another variable that can interfere with population measurements (Stone 1989). Some researchers feel that seasoned traps are more effective than new traps (Stone 1989) while others found no difference in capture rates (Sheehan, pers. comm. 2001).

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.

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, DC. 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.

CONDYLOBASAL LENGTH: The length of the skull, measured from the front of the premaxillary bones to the rear surface of the occipital condyles.

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.

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.*

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

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.

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: 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.

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 inventories. Sampling for species generally takes place within smaller, representative study areas so that results can be extrapolated to the entire project area.

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/]

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.

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.

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

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