THE PARASITES OF THE BOTTA POCKET GOPHER THOMOMYS BOTTAE AND THE TAXONOMY AND BIOLOGY OF RANSOMUS RODENTORUM

by

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A Thesis

Presented to

The faculty of Humboldt State University

In Partial Fulfillment

of the Requirements for the Degree

Master of Arts

June, 1980

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ACKNOWLEDGEMENTS

I would like to thank the members of my committee, Drs. Richard G. Botzler, David L. Largent, and Timothy E. Lawlor, for their guidance and suggestions during my thesis research and text preparation. My additional thanks to Timothy E. Lawlor for allowing me to trap gophers from his land. My special thanks goes to Dr. John D. DeMartini, my major professor, for both his many hours of assistance and guidance, and for allowing me to trap gophers from his land.

I would like to express my appreciation to the following people and agencies: Dr. J.R. Lichtenfels (Leader, Parasite Classification and Distribution Unit, Animal Parasitology Institute) for lending me specimens of Ransomus rodentorum; Drs. John M. Borgerson and Paul S. Kelley (Department of Physics, HSU) for providing rainfall data; Dr. Daniel L. Hauxwell and Carol Anne Hansen (Department of Natural Resources, HSU) for providing soil temperature data; Dr. Richard L. Hurley (Department of Biology, HSU) for providing help in identifying the ectoparasites; Simpson Timber Company for allowing me to trap gophers from their land.

Finally, I thank Cathy, my wife, who provided support and encouragement throughout this project.

INTRODUCTION

Voge (1955, 1956a) and Frandsen and Grundmann (1961), have identified and recorded the distributions of parasites occuring in pocket gophers of the genus Thomomys. Todd et al. (1971) have identified parasites from Thomomys talpoides. There have been no such studies corncening Thomomys bottae in Northern California. Thus, my initial objective was to identify the parasites of T. bottae near McKinleyville, Humboldt Co., California. As my study progressed, questions arose about microgeographic distributions of the helminthofauna. Therefore, determination of microgeographical variation became my second objective. Concurrently, a taxonomic problem arose concerning a strongylid nematode, which was tentatively identified as Ransomus rodentorum Hall 1916. Consequently, my third objective was to clarify the taxonomy of this nematode. Strongylid nematodes commonly infect herbivores (Popova 1964); but R. rodentorum is the only known strongylid parasitizing a subterannean host and the life cycle is unknown. Thus, my final objective was to describe the life cycle and ecology of R. rodentorum.

METHODS AND MATERIALS

For both parasite identification and distributional studies, gophers were trapped from three areas near McKinleyville, Humboldt Co., California that were chosen for both their accessibility and abundance of gophers. All areas are located in range 1E of the Arcata North Quadrangle. Area one is located in the NE corner of sec.32, T7N, 1.2km NE McKinleyville; area two is located in the NW corner of sec.9, T6N, 2km SE McKinleyville; and area three is located in the NE corner of sec.27, T6N, 6km NE McKinleyville. Area one was studied from April, 1979 to February ,1980. Areas two and three were studied only during April and May of 1979. Nematodes and arthropods were fixed and cleared in 70% Ethanol: Glacial Acetic Acid: Glycerine (3:1:1) and were mounted in glycerine jelly. Cross-sections of Heligmosomoides sp. were made using standard microtechnique for identification and were mounted in piccolyte. Cestodes were fixed in Ethanol: Formalin: Acetic Acid (AFA), stained in Delafield's hematoxylin and mounted in piccolyte.

For taxonomic studies, Specimens of Ransomus were obtained from the US National Museum. The cotypes of R. rodentorum (USNM Helm. Coll. No.16181), collected from Thomomys fossor by M.C. Hall and those specimens of Ransomus sp. (USNM Helm. Coll. No. 9072),

collected from Geomys bursarius by O.W. Olsen, were compared to specimens collected locally (USNM Coll. No. 75992).

For life cycle studies, eggs were stripped from females of R. rodentorum, placed in distilled water, and cultured at temperatures varying from 10 to 20 C. Those eggs failing to develop at lower temperatures were subsequentially cultured at 20 C to determine their viability. The eggs of Heligmosomoides sp. were cultured only at 20 C. Eggs and larvae of R. rodentorum were stained with Sudan IV, which is lipid specific, to determine the presence and location of lipid within these life stages. Drawings of life stages were made from wet-mounted specimens and from 35mm color slides of those specimens. Gophers were live-trapped with my modification of Blair's (1941) live trap for small mammals (1941). The gophers were maintained in terraria and fed Purina Dog Chow, carrots and potatoes. Levamisole (Le), obtained from a local veterinarian, was powdered, mixed with water and given orally at 2.6x10-4g(Le)/g host as an antihelminthic. For each infection a gopher was starved for two days before being fed approximately 20 infective larvae placed on pieces of carrots and potatoes. The gopher was initially infected and subsequentially reinfected six days later to establish two size classes of

parasites and to substantiate that infections resulted experimentally.

For ecological studies, area one was studied for approximately eleven months to determine annual quantitative variation in infections of R. rodentorum. Two to four gophers were trapped biweekly from area one and were necropsied either fresh or frozen. Gopher burrows were excavated to observe both the arrangement of nests and seasonal moisture variation. Fecal pellets obtained from nests were mixed with water and checked for infective larvae under a dissecting microscope. Laboratory-reared larvae of R. rodentorum and Heligmosomoides sp. were used to identify wild larvae collected from fecal pellets. Soil temperatures (Carol Hansen, HSU, personal communication) were taken at a depth of 61 cm from two sites, one north and one west, within 4km of study area one for the period January, 1979 to January, 1980 (Appendix, Table 1). Rainfall data were unavailable for the McKinleyville area during the study period, but rainfall averages for Sunnybrae (John Borgerson, HSU, personal communication), approximately 8km S of Mckinleyville, are similar to those of McKinleyville (Paul Kelly, HSU, personal communication). Thus, Sunnybrae data was used for the period January, 1979, to February, 1980 (Appendix, Table 2).

PARASITES AND THEIR DISTRIBUTIONS

Results

Eighty-nine specimens of T. bottae yielded four endoparasitic helminths and two ectoparasitic arthropods (Table 1).

Phylum Platyhelminthes

Class Cestoda

Hymenolepis citelli (Hymenolepididae) occurred only in areas two and three. It inhabited the entire small intestine and infected 10 of the 25 (40%) gophers taken during the spring.

Phylum Nematoda

Class Secernentea

Heligmosomoides sp. (Heligmosomatidae) occurred only in area one. It inhabited the proximal portion of the small intestine and infected 15 of 64 (23%) gophers. Ransomus rodentorum (Strongylidae) occurred in all areas. The worm inhabited primarily the caecum, occasionally the colon, and rarely the small intestine. It infected 67 of 89 (75%) gophers. A larval ascarid (Ascaridae) inhabited the duodenum of one gopher from area two, but not identified.

Phylum Arthropoda

Class Insecta

The flea Foxella ignota (Dolichopsyllidae) and the

Table I Parasites from Thomomys bottae collected in 1979-80 from three sites near McKinleyville, Humboldt Co., California.

PARASITE*	AREA	SAMPLE SIZE	NUMBER INFECTED	%INFECTED
Hymenolepis citelli	1	64	0	0
	2	13	5	38
	3	12	5	42
Heligmosomoides sp.	1	64	15	23
	2	13	0	0
	3	12	0	0
Ransomus rodentorum	1	64	51	80
	2	13	9	69
	3	12	7	58
Foxella ignota present in all areas				
Geomydoecus hueyi present in all areas				

*Larval ascarid not included

louse Geomydoecus hueyi (Trichodectidae) commonly infested gophers from all areas.

Discussion

Hymenolepis citelli was identified in T. bottae from Monterey county (Voge 1955), in ten species and subspecies of rodents from Utah including T. talpoides and Peromyscus maniculatus (Wassom et al. 1973, Frandsen and Grundmann 1961 and Derrick 1971) and in Spermophilus spilosoma from Colorado (Broda and Schmidt 1978).

Hymenolepis citelli is widely distributed both among host groups and geographically, but its distribution is restricted only to areas two and three in this study. While the reason for the absence of H. citelli from hosts from area one is unknown, ecological factors are implicated. Tenebrionid beetles and a camel cricket (Ceuthophilus utahensis) are intermediate hosts for H. citelli (Voge 1956b, Derrick 1971). The occurrence of H. citelli in Peromyscus maniculatus is restricted to locations that are ecologically suitable for the camel cricket (Derrick 1971). The habitat of area one differs from those of areas two and three. Area one was clear-cut and is dominated by azalea (Rhododendron occidentale) and blue brush (Ceanothus thrysiflorus). Areas two and three are pasturelands dominated by grasses and herbaceous annuals. Thus, H.

citelli may be absent in area one due to an unsuitable environment for the intermediate host.

Heligmosomatids in the genus Heligmosomoides are assumed to parasitize only rodents whose distributions include both the Palearctic and the Nearctic regions, especially members of the Microtinae (Durette-Desset 1971). The presence of Heligmosomoides sp. in T. bottae, a strictly Nearctic species, shows that this feature is not valid. Heligmosomoides sp. and H. citelli did not occur in the same area and both parasites inhabit the small intestine. The uncommon occurrance of Heligmosomoides sp. in T. bottae coincides with the absence of H. citelli, a common parasite of Thomomys. Thus, interspecific competition may preclude Heligmosomoides sp. in areas two and three. Because Heligmosomoides is a common parasite of the Microtinae (Durette-Desset 1971), the establishment of Heligmosomoides in T. bottae may be from some microtine reservoir host.

R. rodentorum has been found only in geomyid rodents. It was found in T. fossor from Colorado (Hall 1916), Geomys bursarius from Minnesota (Olsen unpublished), T. fossor from Utah (Todd 1971), and T. talpoides and T. umbrinus from Utah (Frandsen and Grundmann 1961). The identification of R. rodentorum in T. bottae of California represents both a new host

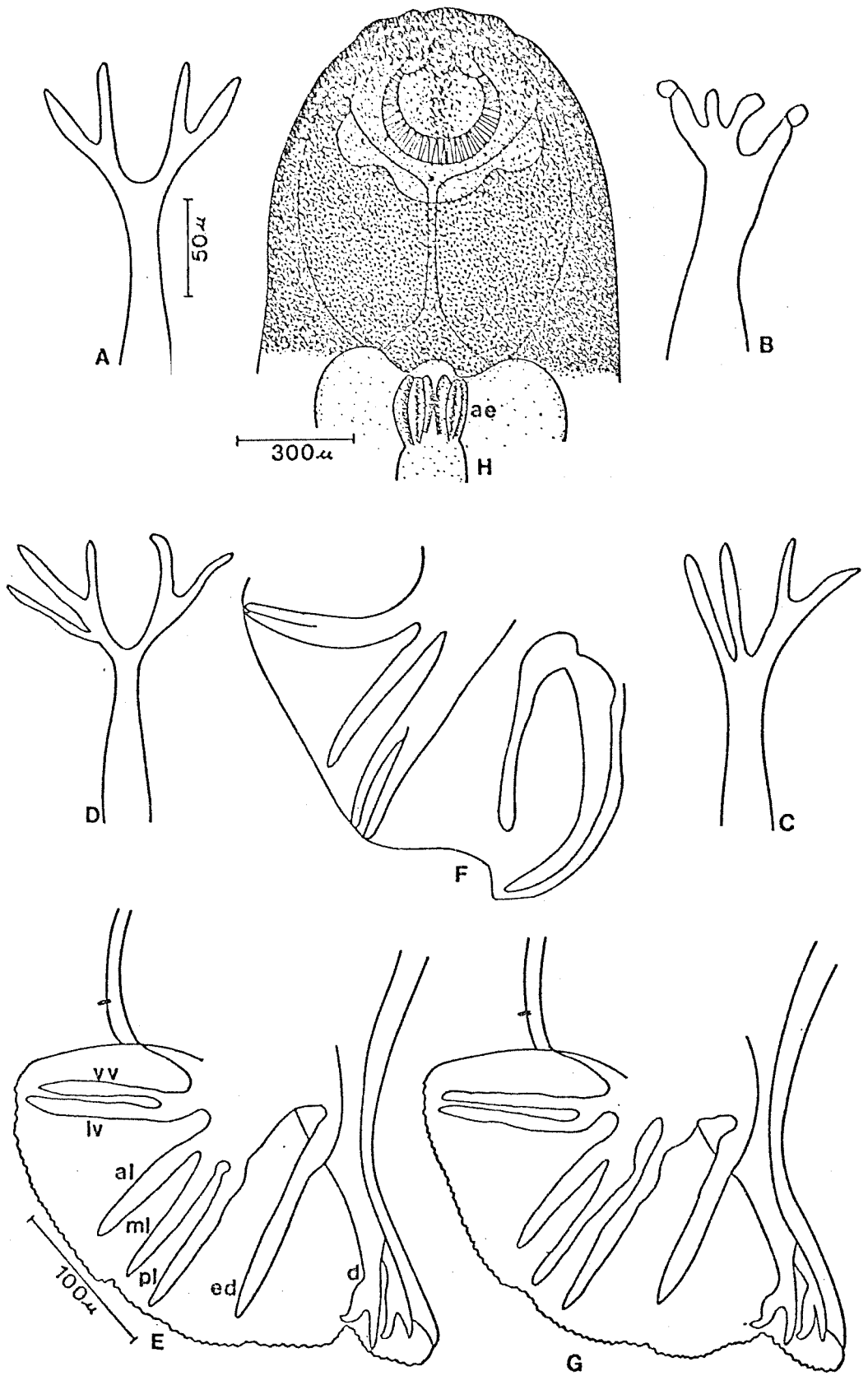
and a new distribution record.

Foxella ignota is common on pocket gophers in the United States and Canada and was identified on T. bottae laticeps in Del Norte Co. (Hubbard 1947) which is adjacent to Humboldt Co. The genus Geomydoecus is found only on geomyid rodents and G. hueyi was recently reported from T. bottae in Humboldt Co. (Price and Hellenthal 1980).

TAXONOMY OF THE GENUS RANSOMUS AND OF THE SPECIES RANSOMUS RODENTORUM HALL 1916

Bursal characteristics of the cotypes and of those specimens collected locally usually agreed, except for occasional variation. However, neither the cotypes, nor my specimens, agreed with Hall's (1916) type description. Variations of the dorsal ray were noted by Frandsen and Grundmann (1961), but they were not documented. I observed variation departing from Hall's description in all bursal rays (Fig. 1A-G). The opaqueness of USNM specimens hindered anatomical studies. However, the female described by Hall differs by two characters from the females collected locally. First, he reported that eggs are uncleaved when oviposited. I observed eggs at the seven or eight cell stage in both the uteri and the ovijectors. Second, uteri are parallel, not divergent as described by Hall,

FIG.1 Cephalic and bursal features of Ransomus rodentorum. A, dorsal ray most commonly observed on USNM and local specimens. B, dorsal ray of type specimen (Hall 1916); C and D, Variations of dorsal ray observed on local specimens; E,F and G lateral view of bursa, E, showing lateral ray configuration most commonly observed on USNM and local specimens, F, of type specimen (Hall 1916), G, as observed on one cotype; H, Dorsal view of Head. ae, anterior esophageal elements; al, anterolateral ray; ed, externodorsal ray; d, dorsal ray; lv, lateroventral ray; ml, mediolateral ray; pl, posterolateral ray; vv, ventroventral ray.


A
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while the ovijectors are divergent. On my specimens, one ovijector extends a short distance posteriorly, then turns anteriorly resulting in parallel uteri. The mouth faces dorsally on all specimens as redescribed by Lichtenfels (1979).

I studied one male specimen collected by Olsen (unpublished) and its bursal features were similar to both those of the cotypes and those collected locally. Thus, I consider all of the specimens that I studied to belong to Ransomus rodentorum.

This report augments the description of Ransomus and of Ransomus rodentorum with the following unreported characters: Location of excretory pore; apparently glandular anterior esophageal elements (Fig. 1H); variation in elements of copulatory bursa; presence of prebursal papillae and of a serrated bursal margin (Fig. 1E and G); ovijectors divergent and uteri parallel; eggs cleaved when oviposited. I have constructed the following redescription of the genus Ransomus and of the species Ransomus rodentorum:

Genus Ransomus Hall 1916

Anterior end of head obliquely truncate, with mouth directed anterodorsally. Buccal capsule large, without interior teeth. Corona radiata composed of numerous small wedge-shaped cuticularizations. Dorsal

lobe of bursa slightly longer than lateral lobes. Ventroventral and lateroventral rays close together and parallel. Lateral rays diverging, with or without a common base. Dorsal ray doubly bifurcate, additional branching sometimes present. Spicules long, tubular and alate. Gubernaculum present. Prebursal papillae located on ventral surface slightly anterior to ventral rays. Vulva slightly anterior to anus. Vagina moderately long. Muscular ovijectors extending anteriorly and posteriorly from vagina; posterior ovijector turning forward after short distance. Uteri parallel. Eggs may be cleaved when oviposited.

Ransomus rodentorum Hall 1916

Worms relatively short and robust. Esophagus clavate, relatively short and robust with six anterior, apparently glandular elements. Rectum distinctly defined. Cuticle finely striated longitudinally. Excretory pore 0.43 to 0.60mm behind ventral edge of mouth.

Male. 4.64 to 8.19mm long and maximum width 0.24 to 0.36mm, at widest part of esophagus. Diameter in plane of buccal aperture 0.69 to 0.75mm, attaining a maximum width of 0.21 to 0.34mm, near posterior end. Nerve ring 0.40 to 0.47mm from anterior end and at middle third of esophagus. Small prebursal papillae 56

to 97u anterior to ventral rays. Bursa relatively short, wide and margin serrated. Tips of anterolateral and externodorsal rays falling appreciably short of bursal margin. Other rays nearly reaching bursal margin. Ventroventral and lateroventral rays approximately equal, may be cleft for varying distances. Lateral rays consisting of anterolateral, mediolateral and posterolateral rays, either sharing common base, or either anterolateral, or posterolateral ray separating from other two rays. All laterals diverging slightly. Externodorsal ray long, thin, arising separately from base of dorsal ray. Dorsal ray extending 70 to 115u, bifurcating, then either bifurcating again immediately, or each branch extending short distance before bifurcating again. Both processes resulting in four terminal digitations. Tubular alate spicules 0.83 to 1.01mm long by 28 to 29u wide proximally, tapering to fine points distally. Cloacal aperture near end of genital cone.

Notable bursal variations are the following: Lateral rays- 1. posterolateral separating from other laterals, 2. anterolateral separating from other laterals, 3. only two laterals present, 4. all laterals sharing common base. Dorsal ray- 1. dorsal bifurcating, one branch extending before bifurcating again while other branch bifurcating immediately, 2.

secondary branching resulting in five or more terminal digitations, 3. dorsal bifurcating, each branch extending for short distance then each bifurcating again, 4. secondary bifurcation at level of first bifurcation.

Female. An addition to Hall's description is that eggs are oviposited at the seven or eight cell stage.

LIFE HISTORY AND ECOLOGY OF RANSOMUS RODENTORUM

Description of Intermediate Life Stages of Ransomus rodentorum

Developmental times of intermediate life stages were recorded for animals at 20 C. Eggs oviposited at seven or eight cell stage, 90 to 100u long by 31 to 50u wide, symmetrically ellipsoidal and blastomeres highly granular (Fig. 2A). First stage larvae evident after day four, internal granules obscuring anatomy. Head rounded, body robust tapering to finely pointed tail (Fig. 2B). Second stage larvae developing within egg by day five, enclosed by first stage cuticle, hatching as early as eight, most hatching after day ten. Granulation still obscuring internal anatomy. Morphology similar to first stage, length and width approximating third stage described below. Third stage larvae developing by day thirteen, occasionally developing within egg, ensheathed in both first and

FIG.2 Intermediate life stages of Ransomus rodentorum. A, oviposited egg with seven blastomeres; B, first stage or second stage larva within egg; C, third stage larva; D, posterior end of fourth stage male larva; E, female fourth stage larva. an, anus; br, bursal rudiments; cl, cloaca; ec, excretory canal; g, gonad; gd, gonoduct; rs, rudimentary spicules; rv, rudimentary vulva; vd, vas deferens.

A
25μ
B
C
an
ec
50μ
D
rs
br
cl
vd
100μ
E
rv
gd
g
250μ

second stage cuticles. First stage cuticle apparently very delicate and sometimes absent. Third stage infective and lethargic. granulation primarily restricted to intestinal cells and to periphery of pseudocoel. Length 320 to 450u by 23 to 34u at widest part of esophagus. Mouth leading into anterior funnel-shaped region apparently representing buccal capsule, then narrowing to join esophagus. Strongyliform esophagus 102 to 125u long, very slender, terminating with end bulb about 15 to 23u wide. Intestine 194 to 328u long consisting of thirteen cell pairs. Anus 43 to 59u anterior to tail, excretory canal 77 to 100u posterior to mouth (Fig. 2C). Fourth stage larvae occurring primarily in cecum. Buccal capsule sub-globular, no corona radiata evident, mouth directed slightly dorsally. Cuticle finely annulated. Sexes distinguishable. Gonads originating near level of anterior third of esophagus, extending posteriorly. Gonoducts extending anteriorly from either cloaca of male or rudimentary vulva of female. Male and female dimensions similar. Length 1.32 to 2.02mm by 0.09 to 0.12mm wide at widest part of esophagus. Esophagus clavate, 240 to 280u long by 69 to 110u at maximum width. Excretory pore 130 to 220u posterior to ventral edge of mouth. Intestine 0.83 to 1.15mm long. Male spicular and bursal rudiments evident.

Female. Anus 90 to 100u anterior to end of tail. Rudimentary vulva 190 to 260u anterior to tail (Fig. 2E,F).

Granules within all free-living stages were diagnosed as lipid. Eggs cultured at 10 C would not develop, but did so when re-cultured at 20 C. Developmental rate was highly variable at all temperatures. Eggs cultured at 15 C produced third stage larvae only after 30 days.

The infected gopher yielded fourth stage larvae that could be grouped into two significantly different size classes ($P<.01$, Student's t test). Fourth stage larvae at sixteen days post-infection were surrounded by loosened cuticles and each male each had a well developed copulatory bursa.

The Ecology of Ransomus rodentorum

Fecal chambers in gopher burrows were always separated from the nest. Chambers usually occurred from 24 to 61cm underground and lacked fresh plant material. Fecal content varied between chambers, but copious amounts indicated their long use. Fecal pellets taken from burrows in area one during July 1979 contained larvae of both R. rodentorum and Heligmosomoides sp. Fresh leaves of composites, presumably the primary food, were found in nests all year. Soil moisture of burrows was high in the rainy

season and low during the dry season.

Infective strongylid larvae are typically transmitted *per os* with ingested vegetation. Since gopher feces are isolated from food sources, movement of larvae to vegetation seems unlikely. Laboratory-reared larvae are either quiescent, or lethargic, indicating that movement away from feces may not occur. Furthermore, during the spring, groups of young worms at similar stages of development were found in high numbers within individual hosts. Thus, worms were probably concentrated in an area and were ingested simultaneously. The fecal chamber is the most logical location for concentration. Coprophagy occurs among various rodents (Geyer et.al. 1974, Barnes et.al. 1957, Bjornhag and Sjoblom 1977) and may supplement their nutritional requirements (Bjornhag and Sjoblom 1977). Coprophagy is not reported for gophers, but I postulate that it effects transmission of infective larvae of *R. rodentorum*.

During the spring, both the relatively high number of young parasites per infected gopher (Fig. 3) and the occurrence of high parasite counts (Table 2) suggest that numerous infective larvae were available to the hosts. Incidence of host infection increased in the spring and remained high for the remainder of the year (Fig. 4), while an increase in adult parasite

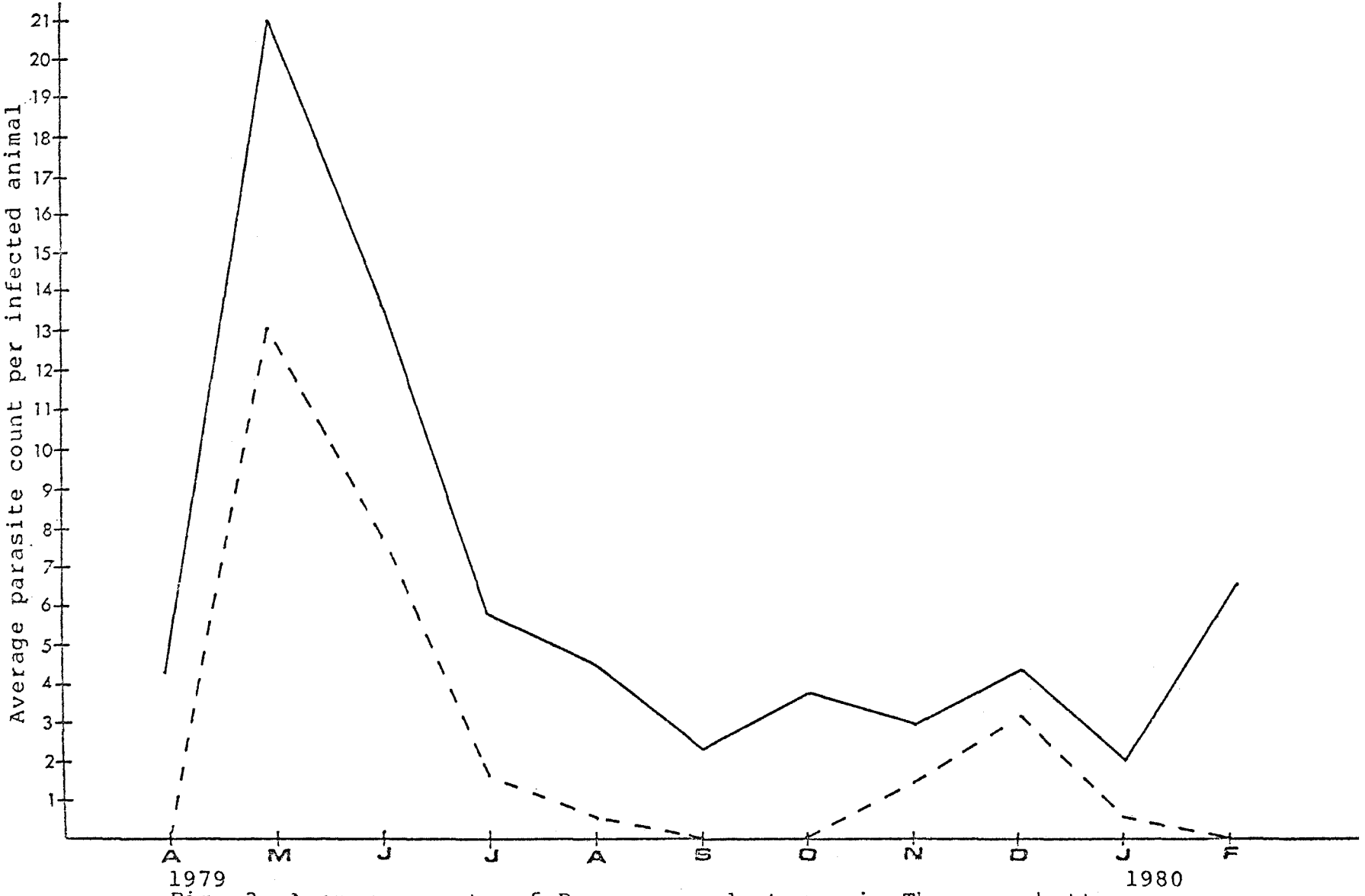

Fig. 3 Average counts of Ransomus rodentorum in Thomomys bottae collected in area one. ——All parasites; - - -Larval parasites.

Table 2 Frequency of counts of Ransomus rodentorum in Thomomys bottae.

MONTH		PARASITE COUNTS 0-10	11-20	21+
Area One				
1979	Apr	7	0	0
	May	3	1	1
	Jun	4	2	1
	Jul	5	1	0
	Aug	8	0	0
	Sep	7	0	0
	Oct	5	0	0
	Nov	3	0	0
	Dec	7	1	0
1979	Jan	6	0	0
	Feb	1	1	0
Area Two				
1979	Apr	3	0	0
	May	4	4	2
Area Three				
1979	Apr	11	1	0

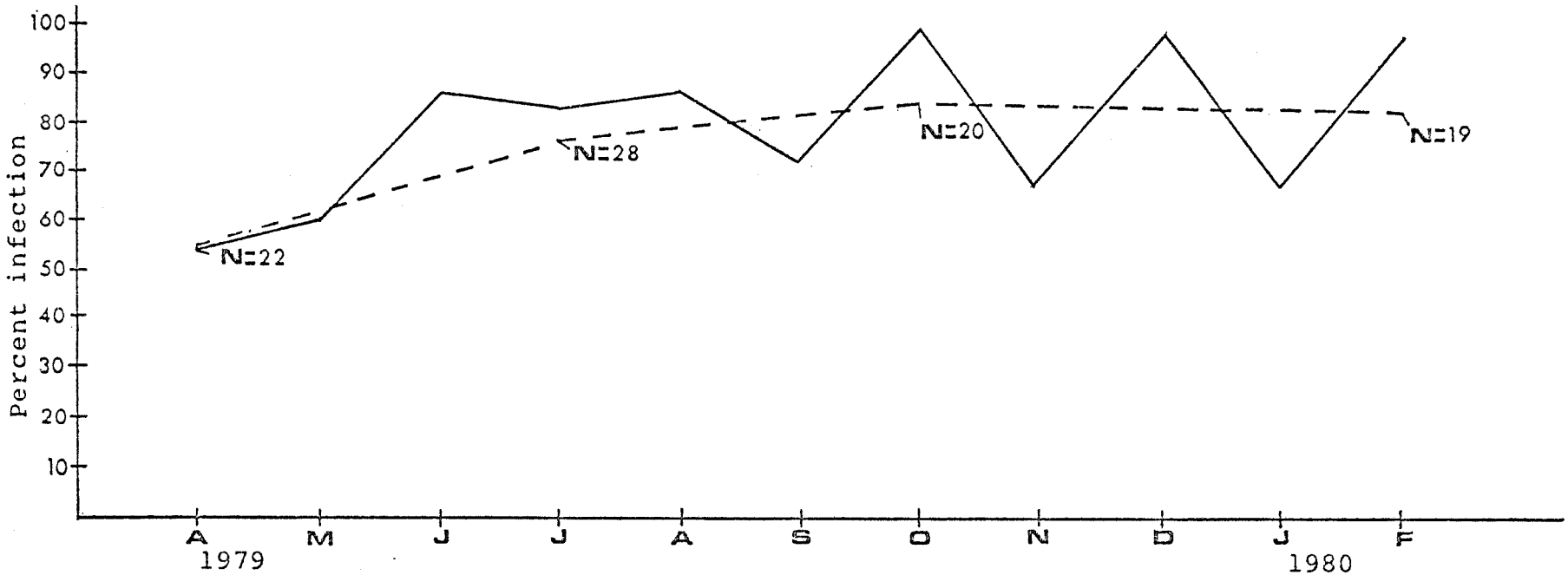

Fig. 4 Incidence of Ransomus rodentorum in Thomomys bottae (all areas).
—— Monthly;- - - - Grouped into sample sizes equal to April.

evels was only transitory (Fig. 3). Thus, the spring infection was most important in increasing the incidence of host infection and this relationship correlates with the presence of young gcphers in the burrows. Gophers are generally solitary (Ingles 1952, Hansen and Miller 1959, and Howard and Childs 1959) and after the young leave the home nest, there are few sources of infection. The parasite's incidence of infection and distribution are favored by the availability of infective larvae prior to the dispersion of young gophers.

The spring infection coincided with soil temperatures increasing above an inhibitory level. Laboratory cultured eggs required temperatures above 10 C to begin development. Soil temperatures from January to April, 1979 ranged from 10 to 13.6 C and either represent inhibitory, or at best marginal temperatures. Temperatures in May ranged from 12.3 to 15 C and approached those required for hatching. Favorable temperatures apparently stimulate the springtime development and lower temperatures are probably a limiting factor during the winter.

The number of young parasites decreased dramatically from July through October when soil temperatures were favorable for hatching. This decrease followed low rainfall totals in late spring.

In December, a slight increase in incidence of infection and the presence of adult parasites was preceded by heavy rainfall in October and November.

Using a correlation of nominal data (Ives and Gibbons in Zar 1974), I found that the combined effects of temperature and rainfall were correlated with monthly larval counts ($r=0.82$, $p<0.05$, Appendix, Table 3). Therefore, a working hypothesis is proposed: In early spring, host infection incidence is low. When the soil temperature increases above inhibitory levels, eggs develop and hatch. Gophers become infected by ingesting infective larvae with feces. Young gophers disperse and establish infections in new burrows. In summer, incidence of host infection is high, but abundance of infective larvae declines due to reduced moisture. In fall, hatching is promoted by rainfall, but may be limited due to decreasing temperatures. Adult parasites resulting from this infection may contribute to winter egg production. In winter, soil temperatures decrease and inhibit larval development. Eggs continue to accumulate and develop when favorable temperatures occur.

Typically, in the superfamilies Strongyloidea and Trichostrongyloidea, a first stage larva hatches from the egg. However, these two superfamilies have examples of advanced larval stages developing within

the egg membrane (referred to as ALD below for brevity) i.e. R. rodentorum (Strongylidae), Uncinaria lucasi (Ancylostomatidae)(Olsen and Lyons 1965), members of the Syngamidae (Ortlepp 1923, Threlfall 1965), Members of the genera Amidostomum (Amidostomatidae) and Epomidiostomum (Leiby and Olsen 1965), and Nematodirus (Trichostrongylidae) (Thomas and Stevens 1960).

ALD is sometimes correlated with hosts whose behavior limits the optimal infection season to a short period of the year. For example, adults of U. .lucasi occur only in pups of northern fur seals (Callorhinus ursinus) and Steller's sea lions (Eumetopias jubata). Eggs pass with feces during the spring, but larvae fail to hatch from the eggs until September (Olsen and Lyons 196 this time susceptible hosts are unavailable, so the free-living larvae must survive until the following spring to complete their life cycle. Similarily, eggs of two sheep-infecting species, N. battus and N. filicollis, require exposure to cold to maximize hatching, resulting in numerous infective larvae coinciding with the arrival of susceptable lambs in the spring (Thomas and Stevens 1960). Finally, the springtime occurrance of infective larvae of R. rodentorum is correlated with favorable temperature and moisture conditions. These conditions coincide with the parturition of gophers. These examples indicate

that maximum exploitation of the host is dependent upon free-living stages surviving through the winter. I now give evidence that ALD may promote survival of free-living larval stages. The relative resistance of both embryonated eggs and third stage larvae among the Strongyloidea and Trichostrongyloidea is well documented (Stewart and Douglas 1938, Kates 1950, Prassad 1959, Silverman and Campbell 1959, Rose 1963, Williams and Mayhew 1967, and Anderson and Levine 1968). Free-living stages of N. filicollis and N. spathiger were "the most resistant to the entire annual complex of climatic conditions" of six genera of strongylines tested and they were especially resistant to cold (Kates 1950). Nematodirus was the only genus utilizing ALD. Protection of delicate first and second stage larvae is apparently enhanced by ALD.

Strongyloid first and second stage larvae typically feed on extrinsic food sources (Levine 1968). In the Ancylostomatidae, these stages produce and store lipid which is subsequentially catabolised by the non-feeding third stage (Giovannola 1936). Giovannola (1936) found lipid in small amounts in first and second stage larvae. Clark (1969) found large amounts of lipid, up to 39% of the dry weight, in third stage larvae of Ancylostoma caninum. However, those strongylines utilizing ALD are ensheathed when they

hatch and therefore cannot feed. Such strongylines must provide an intrinsic energy source within the egg prior to ovipositing. Numerous lipid granules occur in the egg and subsequent larval stages of R. rodentorum. First and second stage larvae of U. lucasi (Olsen and Lyons 1965), N. helvetianus (Herlich 1954), A. skrjabini, A. raillieti, and E. uncinatum (Leiby and Olsen 1965) are very granular and these granules are also probably lipoidal. Incorporation of large amounts of lipid in the oocyte would eliminate the need for an extrinsic food source for developing free-living stages. As a corollary, an intrinsic energy source would preclude the need for hatching, resulting in greater protection of larvae within the egg membrane.

Contrastingly, overwintering eggs of Amidostomum and Epomidiostomum were considered as inconsequential to high springtime infections (Leiby and Olsen 1965). This conclusion was based on Cowan's statement (1955), unsupported by published data, that infectivity decreased in A. anseris when eggs were kept longer than ten days at 6 C. Cowan further stated that a goose maintained over the winter on a plot infected with A. anseris eggs, but allowed to mix with wild waterfowl, became infected by March. Thus, evidence is ambiguous as to the resistance of these eggs. Stradowski (1975) showed that A. anseris eggs were

extremely resistant to cold, but mortality increased over time. Therefore, winter survival may be an important factor in promoting these infections. This is consistent with the postulated adaptiveness of ALD.

SUMMARY

Eighty-nine pocket gophers were trapped from three areas between April, 1979 and February, 1980. The parasites Ransomus rodentorum (Strongylidae), Foxella ignota (Dolichopsyllidae) and Geomydoecus hueyi (Trichodectidae) occurred in all areas. Hymenolepis citelli (Hymenolepididae) and Heligmosomoides sp. (Heligmosomatidae) had geographically separate distributions indicating possible interspecific competition among these species of parasites.

I believe that all specimens of the genus Ransomus examined from the USNM and those collected from T. bottae belong to Ransomus rodentorum. The genus Ransomus and the species Ransomus rodentorum is redescribed.

The life cycle of R. rodentorum is evidently direct. It varies from other known strongylids by second and third stage lavae developing within the egg. Transmission is evidently via coprophagy. Lipoidal material is present within the egg, occurs in all free-living stages and apparently serves as an energy

source. Soil temperature and moisture apparently act synergistically on egg development of R. rodentorum. Rising soil temperatures, associated with abundant soil moisture apparently stimulate egg development in the spring. Spring infections are most important for increasing incidence of host infection and coincide with host parturition. Increased rainfall, associated with favorable soil temperatures apparently stimulate egg development in the fall resulting in minor infections. Advanced larval stages developing within the egg membrane is uncommon in known life cycles of both the Strongyloidea and Trichostrongylidea, and may protect delicate free-living stages.

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APPENDIX

Appendix Table 1. Soil temperature at 61cm deep within 4km of study area.

Month	Site One		Site Two	
station	G1	G2	G1	G2
Jan	9.8	9.0	9.9	9.5
Feb	10.1	9.0	10.4	9.9
Mar	11.5	10.1	12.2	11.7
Apr	12.3	11.0	13.6	12.7
May	13.6	12.3	15.0	14.2
Jun	15.4	13.6	16.7	15.6
Jul	16.7	14.9	18.2	17.1
Aug	16.3	14.9	17.7	16.7
Sep	-	-	18.2	17.7
Oct	15.8	14.6	16.7	16.7
Nov	13.6	12.3	13.6	13.2
Dec	10.1	9.8	10.4	10.4
Jan	11.0	10.1	11.7	11.3

Appendix table 2. Rainfall during study period.

Month	Average Monthly Total		Monthly Totals
	McKinleyville	Sunnybrae	Sunnybrae 1979-80
Jan	6.93	8.86	4.58
Feb	8.41	5.70	8.56
Mar	8.22	7.54	2.64
Apr	4.53	3.22	5.62
May	1.00	1.57	3.57
Jun	0.70	0.73	0.22
Jul	0.12	0.15	0.26
Aug	0.36	0.73	0.05
Sep	2.24	1.45	1.36
Oct	4.92	3.58	7.99
Nov	9.42	7.87	8.14
Dec	9.27	8.77	5.78
Jan	-	-	5.21

McKinleyville data includes five years during the period 1971-1978, Sunnybrae data is for the period 1966-1980.

Appendix Table 3. Correlation of temperature and rainfall to larval abundance of Ransomus rodentorum.

Month	Temp. >12.5 C	Rainfall >5 in.	Sum	Sum Coded	larvae Avg.>1
Apr	-	+	0	-	-
May	+	+	2	+	+
Jun	+	+	2	+	+
Jul	+	-	0	-	+
Aug	+	-	0	-	-
Sep	+	-	0	-	-
Oct	+	-	0	-	-
Nov	+	+	2	+	+
Dec	+	+	2	+	+
Jan	-	+	0	-	-
Feb	-	+	0	-	-

Temp.- soil temperature recorded from one month previous. Rainfall- total for two months previous. Sum- combined temperature and rainfall signs. Sum coded- from Sum coulumn; sum 0=-, sum 2=+. Larvae- average for that month. Correlation: rn= a-b/a+b. Where a=those signs that agree in columns Sum Coded and Larvae Avg.>1 i.e. +,+ or -,-; b=those signs that disagree, i.e. +,- or -,+; Calculation: a=10, b=1; rn = 10-1/10+1 = 0.82 (p=0.05).