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ANALYSIS OF DROPPINGS TO DESCRIBE DIETS OF SMALL BIRDS

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Stomach contents have been the major source of dietary information for small birds and other vertebrates. However, killing specimens to look at stomach contents is not an option in studies of endangered species and often is undesirable in other studies. Emetics, causing regurgitation of stomach contents, can be used successfully with small birds, but can stress and even kill them (Prys-Jones et al. 1974, Radke and Frydendall 1974). Collecting or photographing food brought to nestlings may be possible, but only for a limited season and necessarily few individuals. Droppings are produced by all birds all year and can be collected with little stress on the bird, often with little extra effort by the researcher. Large samples are thus possible. We describe here an efficient method for individually examining large numbers of droppings of small birds for a quantitative description of arthropods and fruit seeds in their diets.

Davies' work with flycatchers (1977b) and wagtails (1976, 1977a), Waugh and Hails' work with a swiftlet (1983), and Tatner's work with a magpie (1983) address the problems of differential digestibility and find good agreement between feces contents and food eaten or stomach contents. The agreement held for softer-bodied groups, such as flies, as well as hard insects, such as beetles. At least in these species the feces provided a fairly unbiased sample, which may be freely analyzed in many ways. Feces from other species may be biased samples, but they still contain much information that can be useful.

We tried methods that used feces of small, insectivorous birds or bats (Lobb and Wood 1971, Bryant 1973, Davies 1976, 1977a, b, Matsuoka and Kojima 1979, Waugh 1979, Whitaker and Findley 1980, Moeed and Fitzgerald 1982, Tatner 1983, Waugh and Hails 1983, and others in progress) and were frustrated because the fecal analysis was only vaguely described, was tedious, or had been short-cut by pooling many droppings. Such a useful technique warrants careful description, especially to allow comparable studies to follow. As part of a study of Hawaiian forest birds we needed to examine more than 1000 droppings. We preferred not to lose data by pooling samples, and our droppings did not disperse in liquid well enough to be mixed and subsampled. Here we detail the method we developed, mention some procedures that did not work, and illustrate or list some arthropod structures we found useful.

METHODS

Collecting and storing droppings.—Droppings were collected during banding operations in native forests of the island of Hawaii from fall

1976 to winter 1981. Birds taken from mist nets were put individually in clean, labeled cotton bags for transport and holding before processing, during which time they usually defecated. Droppings were later scraped individually from bags into paper packets and stored frozen. Some were stored with no ill effects for more than 4 years. We examined droppings from 12 species of passerines, including insectivorous, frugivorous, granivorous, and nectarivorous species.

Preparing droppings.—We needed to develop a method that would effectively and quickly break down a large number of droppings individually so that the contents would be well dispersed and easily viewed. For initial tests, we collected droppings of Common Myna (*Acridotheres tristis*) and House Sparrow (*Passer domesticus*) from the ground under roosts in Honolulu. We usually used about 8 droppings for each treatment.

We tried solutions of various chemicals to soften and disperse the dropping contents: 70% ethanol, 5% KOH, water, liquid dishwashing detergent (5 ml in about 80 ml water), laundry soda (Na_2CO_3; 1 part soda in 2 parts water), detergent and laundry soda combined, sulfuric acid (H_2SO_4; in various concentrations), bath oil powder, and meat tenderizer crystals. Droppings were soaked overnight in all of these and were boiled in all but ethanol and KOH. To boil droppings in these solutions, we tried holding test tubes individually over a flame, placing batches of test tubes standing in beakers in a microwave oven, standing test tubes in a boiling water bath, and standing test tubes in a kitchen pressure cooker. We used the pressure cooker to steam droppings. As containers for the droppings during these procedures, we tried test tubes, shell vials, and for steaming, paper. Usually, each sample was shaken vigorously before it was poured out and examined.

Examining droppings.—The procedure for microscopic examination is described below.

RESULTS

Problems with unsuitable treatments.—None of the chemicals we tried dispersed the droppings better than water. In all solutions, portions of some droppings remained as chunks, just as in all solutions some droppings dispersed nicely. Moeed (pers. comm.) recommended using the laundry soda solution (0.25 M Na_2CO_3), saying this dissolved the uric acid and mucus that held droppings together and clouded the liquid. He also suggested boiling each sample to agitate it thoroughly. Lobb and Wood (1971) used KOH successfully. However, we did not find these solutions helpful, and our samples were not clouded by uric acid. Boiling seemed like a good idea, but required close attention to prevent sudden boiling over. Treatment with acid required subsequent neutralization, and when we then introduced alcohol, a dense flocculant clouded the samples.

Dropping preparation, handling, and storage.—The simplest and quickest method to disperse droppings was to steam them in a pressure cooker

for 30 min, and this was as effective as any other treatment. After being weighed, each dropping was placed in a 1-dram shell vial with one or two drops of water and stood in a shallow tin can with holes punched in the bottom to allow water drainage. We used tuna cans, which held 23 vials. Three cans were placed in a pressure cooker on a platform over the water and loosely covered with a sheet of paper to prevent water dripping from the pot lid into the vials. Following 30 min of steaming under pressure, the vials were cooled, partially filled with 70% ethanol, and corked with neoprene stoppers.

Some droppings treated this way still needed to be broken gently apart while being examined under the microscope. For breaking up these droppings we used a pen point with a broad, circular, flat tip (a speedball type B, size 4 pen nib). A small amount of pressure carefully applied with this "crusher" dispersed the contents but did not damage insect remains. For handling things being viewed in the microscope, two pairs of very fine forceps were indispensable for each worker.

For examination the dropping was poured from its vial into a white porcelain combustion boat (97 mm long × 16 mm wide × 10 mm deep or 100 × 20 × 13 mm, depending on the size of the sample) and examined using a binocular dissecting microscope fitted with an ocular micrometer. Combustion boats are normally used to hold samples being baked or burned in a bomb calorimeter. Their rectangular shape makes it possible to examine a sample completely with a single pass along its length, since the full width is in view under 16 power. Their slight spout facilitates pouring out the sample.

We saved each sample for reference. Each was filtered through VWR Grade 613, smooth, white filter paper placed in a Buchner funnel connected to an electric vacuum pump (10 PSI, maximum vacuum 30 cm [12 in] Hg). The filter paper was wider diameter (5.5 cm) than the funnel, extending up the sides of the funnel to prevent any of the sample from escaping under the edges of the paper. Without the pump, drainage was too slow. The samples were then air dried and stored folded up in the labelled filter paper.

Examination of the sample.—The variety of fragments recognizable in the finely ground matrix of droppings was at first overwhelming. To identify the origins of these, a reference collection of arthropods and fruits from the study area was essential. Well-illustrated entomology texts (Borror et al. 1976; Peterson 1948, 1951) were also useful. Studying droppings of a caged bird fed known insects was a valuable way to gain familiarity with arthropod structure. Some fragments, such as wings and heads, posed only minor problems in identification. Others, the smaller or internal structures, required systematic dissection (e.g., mandibles of each group) or serendipity to match with their owners. Table 1 lists, and Figs. 1–24 illustrate, some of these less obvious structures. These are structures that survive digestion and are diagnostic. They represent cosmopolitan and common arthropod groups. Illustrations of these structures are few or too widespread in entomological literature

TABLE 1. Some little-known structures and features commonly found in droppings.

Group	Structure	Description or comments
Psocoptera	mandibles	Small, translucent, but dark on the two points.
Hemiptera		
Adult	clavus, corium	These were easily recognized without the membrane attached.
Nabidae	foreleg (Fig. 1)	Tibia has two long rows of small, black teeth.
Male	clasper (Fig. 2)	Part of the male genitalia, visible at tip of abdomen.
Homoptera	"rib" (Figs. 3, 4, 5)	The strong, curved apodeme (an internal ridge of the exoskeleton) associated with the hind leg was distinctive for each family (nymphal psyllids lack this).
Cicadellidae	hindleg	Tibia has rows of prominent spines, marked by dark bumps where the spine has been knocked off.
Delphacidae	hindleg	Tibia has a large, toothed, movable spur, or calcar, at apex. Tibia and tarsal segments have several large apical teeth.
Cixiidae	hindleg	Similar to delphacid's, but tooth pattern distinguishable, and calcar lacking.
Psyllidae		
Adult	hindleg	In our species, the tibia has two or four small, dark, apical spurs.
Nymph	wing pad (Fig. 6)	
	abdominal terga (Fig. 6)	For our one species with free-living nymphs, this dorsal covering of the abdomen was the most numerous fragment.
Neuroptera		
Chrysopidae		
Nymph	mandible, maxilla (Fig. 7)	This smooth, sickle-shaped piece (4 per individual) occurs alone, showing the flat surface that matches its dorsal or ventral mate, as well as attached to its mate, forming a rounded sickle.
	tarsus (Fig. 8)	
Coleoptera	mandible (Figs. 9, 10)	Beetle mandibles are so diverse as to defy generalization. They sometimes differ between adult and larva of the same species. They differ from larval Lepidoptera in being usually more elongate and bearing teeth or grinding surfaces somewhere besides the apical edge. They must also be distinguished from Hymenoptera and larval Tipulidae mandibles.

TABLE 1. Continued.

Group	Structure	Description or comments
Carabidae		
Adult	foreleg	Tibia of our species was distinctively notched.
	hind trochanter (Fig. 11)	Found separate, as well as attached to the coxa.
Curculionidae		
Adult	tarsus	In our fauna penultimate tarsal segment has two large, flat lobes.
Lepidoptera		
Adult	wing scales (Fig. 12)	Wings are covered with countless small scales, which may be swallowed even if the bird tears off the wings.
Larva (=caterpillar)	mandible (Fig. 13)	Most commonly shaped like baseball glove, or broad scoop, with one or more teeth along the cutting edge and a spherical knob at one of the basal corners.
	front (Fig. 14)	A triangular sclerite on the front of the head.
	spiracle	A dark, elliptical ring.
	crochet (Fig. 15)	Many occur on each proleg.
	anal comb (Fig. 16)	Only some families have this.
Diptera		
Adult		
Cyclorrhapha (many sturdy, large, black flies)	antenna	Apical segment is acorn-shaped, was often encountered.
	wing	Leading edge has small but stout, curved bristles.
	bristles (setae)	These are numerous, sometimes found still attached to legs. They are strong, black, slightly curved, tapered.
Larva	entire body	Surprisingly, these did occur in droppings.
Tipulidae	mandible	Finger-like teeth.
	head	V-shaped incisions along posterior edge of head capsule.
Hymenoptera		
Adult		
Wasps	head	Hard, hypognathous, with a distinct, round foramen where it connects with thorax.
	mandible (Fig. 17)	Generally longer and slenderer than those of Coleoptera or Lepidoptera, with two apical teeth.
Ants	thorax	The hump or node on the "waist" is distinctive.

TABLE 1. Continued.

Group	Structure	Description or comments
Araneida	chelicera (Fig. 18)	Even when fangs were absent, chelicerae were distinguishable by their slightly asymmetric but conical shape and sometimes an arrangement of spines.
	fang (Figs. 19, 20)	Curved and sharp, this piece sometimes resembled tarsal claws.
	leg (Fig. 21)	Leg segments tend to be straight-sided, whereas those of insects usually taper at the joints. Spiders' also are usually hairy. Simple tarsus with two claws is diagnostic.
Male	pedipalp (Fig. 22)	Male genitalia in the spherical or egg-shaped terminal segment occurred frequently.
Pseudoscorpionida	"pincer" (Figs. 23, 24) (chelate pedipalp)	The movable and stationary pieces were usually found separate. The smaller piece has a finely serrated edge in our species.

to be useful to the ornithologist. To become familiar with the arthropod fauna his avian subjects might be eating, the ornithologist should examine the structures we list here, especially for the groups common in the study area.

Some fragments had to be tallied at first under a code number because we did not know from what prey they came. By the end of the study we had a large collection of still-unidentified fragments, but usually none occurred in more than 3 samples.

We soon discerned which fragments survived digestion most often, but we never felt we could name a "set standard," as did Lobb and Wood (1971), or rely on just wings, as did Davies (1977b) and Waugh and Hails (1983). We felt we needed to watch for a variety of clues for most food items, as did Tatner (1983). The number of a particular fragment, or one half that if it occurred in pairs, was usually our estimate of the number of individuals represented. Sometimes matching pairs of

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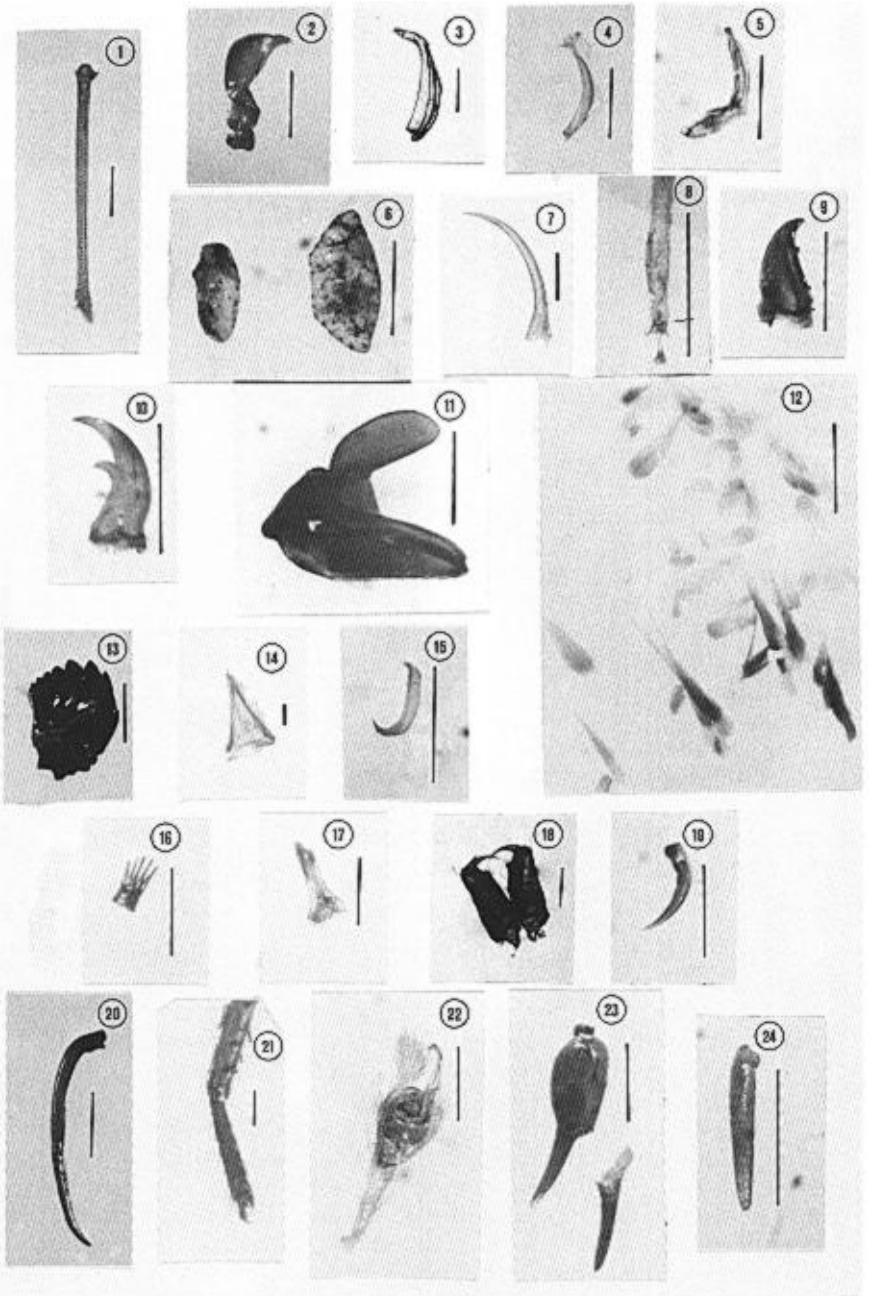
FIGURES 1–24. Arthropod structures frequently encountered in bird droppings. The vertical line to the right of each item is 0.5 mm long. 1. Nabid foreleg tibia, inner surface. 2. Nabid male clasper. 3. Delphacid "rib." 4. Cicadellid "rib." 5. Psyllid "rib." 6. Psyllid nymph wing pad (left) and abdominal terga (right). 7. Chrysopid mandible. 8. Chrysopid tarsus. 9. Carabid adult mandible. 10. Carabid larva mandible. 11. Carabid trochanter (the elliptical portion above). 12. Lepidoptera scales. 13. Lepidoptera larva mandible. 14. Lepidoptera larva front. 15. Lepidoptera larva crochet. 16. Lepidoptera larva anal comb. 17. Hymenoptera adult mandible. 18. Spider



chelicerae, still paired, fangs in place. 19. Spider fang. 20. Spider fang. 21. Spider leg tip. 22. Male spider genitalia on pedipalp. 23. Pseudoscorpion pincer, movable piece separated. 24. Pseudoscorpion pincer, movable piece.

chelicerae, mandibles, wings, or other pieces was possible and provided a slightly larger, though still conservative, estimate. To expedite counting, when a sample contained more than about 50 of an item, it was counted in one quarter of the boat only and the result multiplied by four.

Certain structures, e.g., spider chelicerae, Lepidoptera larva mandibles, and Diptera and Hymenoptera wings, seemed useful as indicators of prey size, so were measured as they were encountered. However, no more than 10 of any item were measured from any one sample.

Overview.—Droppings varied in size and consistency among species. The most nectarivorous species (Iiwi, *Vestiaria coccinea*: Apapane, *Himatione sanguinea*) produced very small droppings containing correspondingly little information. The most granivorous species (Northern Cardinal, *Cardinalis cardinalis*) had very hard droppings that needed to be teased apart with forceps even after steaming. Even then the contents were finely ground. Most species (Akepa, *Loxops coccineus*; Akiapolaau, *Hemignathus munroi*; Hawaii Creeper, *Oreomystis mana*; Common Amakihi, *Hemignathus virens*; Elepaio, *Chasiempis sandwichensis*; Japanese White-eye, *Zosterops japonicus*; Red-billed Leiothrix, *Leiothrix lutea*) were largely insectivorous. Their droppings were the most easily and profitably handled with our method. Droppings of a larger, more frugivorous bird, the Hawaiian Thrush (*Phaeornis obscurus*), also were appropriate for this method, but those of the even larger, frugivorous Hawaiian Crow (*Corvus hawaiiensis*) needed a larger combusion boat and did not require any steaming.

After we recognized arthropod structures and seed types and learned the counting systems, processing one dropping took about 15 to 30 min, depending on the size and type of dropping. Using the method described here, two people (not trained in entomology) working full-time for 3 months analyzed more than 1000 droppings.

DISCUSSION

The two major obstacles in this method were the cohesiveness of some droppings and the fragmented nature of the material to be identified. Droppings varied both within and among species in how tightly the contents were cemented together. We suspect that steaming would be effective in softening droppings of most small, insectivorous and frugivorous birds. It might not be necessary at all for largely frugivorous species. We do not know why Moeed and Fitzgerald (1982) found their samples cloudy unless treated with Na_2CO_3, while we did not.

For identifying arthropod fragments, nothing could substitute for experience, although expertise in entomology did not guarantee a person could identify even the most common fragments. Trainees could quickly learn to recognize the common pieces. This paper should speed this process. Entomological training was useful in tracking down the less common fragments by consulting reference collections and books.

Though undoubtedly somewhat biased in certain ways (Hartley 1948), information from droppings is quite suitable for many kinds of analysis and is infinitely more useful than no information. We suspect that materials we found in droppings probably under-represented some very small food items, such as tiny spiders. However, all arthropods and fruits we expected the birds to be eating showed up in some form in the droppings.

Bird droppings can be collected with little extra effort during other operations, such as mist netting in a field study. With the steaming procedure we suggest here, the droppings are easily prepared for examination, which is the time-consuming step. The equipment and counting procedures we used streamlined the examination without sacrificing too much detail, making it physically and economically possible to analyze a large number of droppings. In some studies, if the droppings are more easily dispersed than ours were, making a homogeneous mixture, they could be pooled and the mixture subsampled, saving some time. Though no procedure can eliminate the tedium in dropping analysis, we recommend this procedure to describe the range of diets and the relative importance of various foods for small birds.

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