SECTION 1

Fecal Examination in the Diagnosis of Parasitism

THE PROPER examination of the feces will provide evidence of, or an accurate identification for, most of the parasites which inhabit the alimentary canal. Also, certain parasites of the respiratory tract may be diagnosed by fecal examination, because most of the sputum of animals is swallowed (Figs. 29, 30, 65, 66, 78, 79, 92, 93, 118, 119, 161, 162). Mange or scab mites may be licked or nibbled from the skin, thus accounting for their appearance in the feces (Fig. 130). Fecal examination may also reveal, to a limited extent, the status of digestion, as is shown by the presence of undigested muscle (Figs. 143, 144), of starch, or of fat droplets.

Animals may swallow certain objects that resemble parasite forms. These are known as *pseudoparasites;* they include such things as pollen grains, plant hairs, grain mites, mold spores, and a variety of harmless plant and animal debris (Figs. 67, 132 to 138, 141, 142, 171, 172, 189, 190). *Spurious parasites* are encountered in feces. For example, parasite eggs or cysts from one species of host may be found in the feces of a scavenger or predator host as the result of coprophagy (Figs. 131, 139, 140).

Collection of Fecal Samples

Fresh feces should be used whenever obtainable. Old samples may become dehydrated, making suspension difficult; also worm ova or coccidial oocysts may undergo development, hatching, or disintegration to such a degree as to interfere with diagnosis.

Animal owners may submit fecal samples in all sorts of containers, suitable or not suitable. It is suggested that clients be supplied with clean, wide-mouthed, screw-capped or stoppered jars of at least 60 ml. (2 oz.) capacity. One or two wooden tongue blades are convenient for picking up samples, after which they are discarded. Formed droppings may be transported for a few hours when well wrapped in waterproofed paper.

At least several grams of feces should be collected for an examination. Because of the roughage content, larger samples should be secured from herbivorous than from carnivorous animals.

If defecation does not provide sufficient material, it may be taken directly from the rectum, or, defecation may be induced quickly by inserting a suppository made from bar-soap or a paper match from an ordinary book match folder. Plain water enema-samples may be obtained, but the dilution factor makes them undesirable as a rule. Soapy or oily enemas should not be used. Fecal specimens removed from rectal thermometers are seldom satisfactory in quantity.

If fecal material is to be transported for more than a few hours, it must be preserved. A 10 per cent formalin solution may be added to saturate the sample. Refrigeration will also keep samples in good condition for several days.

Fecal samples to be shipped by postal service, express, or by other means, should be enclosed in leak-proof containers. Proper identification of each sample by means of a label or a tag is necessary.

Gross Examination of Feces

Gross examination should always be made for the detection of living or dead worms or for the detection of the segments of tapeworms. Oily or soapy substances in samples will indicate that the microscopic examination will be difficult or even impossible.

Microscopic Examination of Feces

This may include several techniques such as: (A) The simple smear method; (B) Qualitative concentration methods; and (C) Quantitative concentration methods.

A. The *simple fecal smear method* of microscopic examination is better than no examination at all, but it has many disadvantages. It should be used only when very small samples are available or when lack of equipment or time prevents the use of a more accurate technique. The simple smear is carried out as follows:

1. Place a microslide on a small piece of newspaper.
2. Place a drop of tap water on the center of the slide.

3. With a toothpick, or some similar instrument, detach from the fecal mass a small sample, about the size of a grain of wheat.
4. Mix the sample into the drop of water on the slide until the suspension is cloudy, but not too much so to read the newspaper printing through it. By means of a finely pointed forceps, remove any larger bits of debris that may be present.
5. Gently lower a square 18 mm. or 22 mm. glass or plastic coverglass onto the specimen on the microslide.
6. Examine systematically under low power (x 100) of the microscope, using the high dry power (x 400) for the observation of details (Fig. 14).

B. *Qualitative microscopic concentration methods of fecal examination.* Techniques of this type will be of greatest value in routine clinical diagnosis. They will detect most alimentary-canal parasitisms and, in addition, certain of those from the respiratory tract. They may also serve to diagnose skin mange of the dog, fox, and cat (Fig. 130).

The method to be described is reasonably rapid and its usage is increasing in veterinary diagnosis. It is of value particularly in the field of small animal practice, although it may be very useful in the detection of certain parasitisms of horses, cattle, sheep, goats, swine, and poultry. Animal owners are interested, usually, in seeing parasitic forms under the microscope. Animal surgery is made more safe by postponing operations on parasitized patients until such hosts are de-parasitized. Veterinary hospital contamination, and the transfer of many parasite species from patient to patient, may be avoided through the isolation and treatment of those animals whose feces show evidence of a parasite burden.

A parasitized animal not exhibiting clinical symptoms may enter a veterinary hospital. Should parasitism develop to the clinical stage after that patient returns home, the owner may unjustly conclude that the animal acquired the parasites while in the hospital. Routine examination for parasites of all hospitalized patients would avoid such criticism.

Fecal examination methods can, and should, be conducted in such a manner as to avoid contamination of the laboratory. To prevent the dissemination of odors, keep the samples covered as much as is possible. Various commercial products are available for the masking or for the neutralization of odors.

Concentration of parasitic ova or oocysts from feces may be accomplished in a number of ways. All methods depend upon mixing the fecal sample with a liquid, the specific gravity of which is greater than that of most of such forms, yet less than the specific gravity of most of the fecal debris. Thus the parasite forms rise to the top of the flotation fluid by gravity — a process that may be hastened by centrifugation.

Flotation fluids may be of various composition. Those most commonly recommended include heavy solutions of sodium chloride, sucrose (cane or beet sugar), glycerine, zinc sulfate, zinc acetate, sodium nitrate, sodium acetate, or magnesium sulfate. None of these solutions is ideal for this purpose. Glycerine has too high a viscosity, hence flotation is slow. The saline solutions are low in viscosity but they tend to dehydrate and thus distort parasite forms; also they crystallize rather quickly on the microslide. Solutions of high specific gravity (sp. gr. 1.400) will float too much debris, thus defeating the purpose for which they are intended.

MODIFIED SUGAR FLOTATION TECHNIQUE

Sheather (1923) first proposed heavy sugar solution for fecal flotation technique. Our experience has shown that sugar solution (sp. gr. 1.200 to 1.300) is the most satisfactory flotation fluid available for routine qualitative clinical fecal examinations employing centrifugation. This solution will fail to float most of the ova of tapeworms, flukes, and thorny-headed worms. This is not a serious objection because tapeworm ova usually leave the host enclosed within the worm's segments which may be seen grossly on or in the feces; and, except in certain localities, flukes and thorny-headed worms are not highly important parasites of domesticated animals. A technique for finding fluke eggs in feces will be found on page 16.

PREPARATION OF SUGAR FLOTATION SOLUTION

1. Materials:
   Granulated sugar .....454 gm. (1 lb. avoir.)
   Tap water .....355 ml. (12 fluid oz.)
   Liquefied phenol crystals .....6.7 ml. (1.8 fluid dr.)
2. Place the tap water in the upper half of a double boiler.
3. Dissolve the sugar in the water by stirring. The water in the lower half of the double boiler should be close to the boiling point (do not dissolve the sugar by means of direct heat).
4. Place phenol (carbolic acid) crystals in a small graduated glass cylinder. Dissolve the crystals by immersing and rotating the graduate in water near to the boiling point.
5. Add the required quantity of liquefied phenol to the sugar solution while stirring the latter. The phenol acts as a preservative and prevents the growth of molds.
6. Store the sugar solution in stoppered 8-oz. (237-ml.) bottles.

APPARATUS FOR A QUALITATIVE MICROSCOPIC CONCENTRATION METHOD OF FECAL EXAMINATION (FIG. 1)

1. *The microscope.* Magnifications of approximately x 100 and x 400 are most suitable for fecal examinations. Therefore, the optical equipment should include an 8X or 10X Huyghenian ocular, 16-mm. and 4-mm. achromatic objectives, and a substage condenser of 1.25 numerical aperture. A mechanical stage and a binocular body tube with matched oculars are not essential, but they will save the examiner's time and help to reduce eyestrain. The addition of an oil immersion objective will equip the microscope for all the important clinical procedures that require microscopy.
2. *Lens paper.* This is essential for keeping optical lenses clean. Squares of about 8 cm. (3 in.) may be stored in a covered dish or can. They should be used once, then discarded.
3. *Xylene.* This is the only safe lens-cleaning solvent except water. Xylene should be dispensed from a dropper-bottle.
4. *Microscope lamp.* Daylight should not be relied upon. There are many suitable types of microscope lamps. A simple type

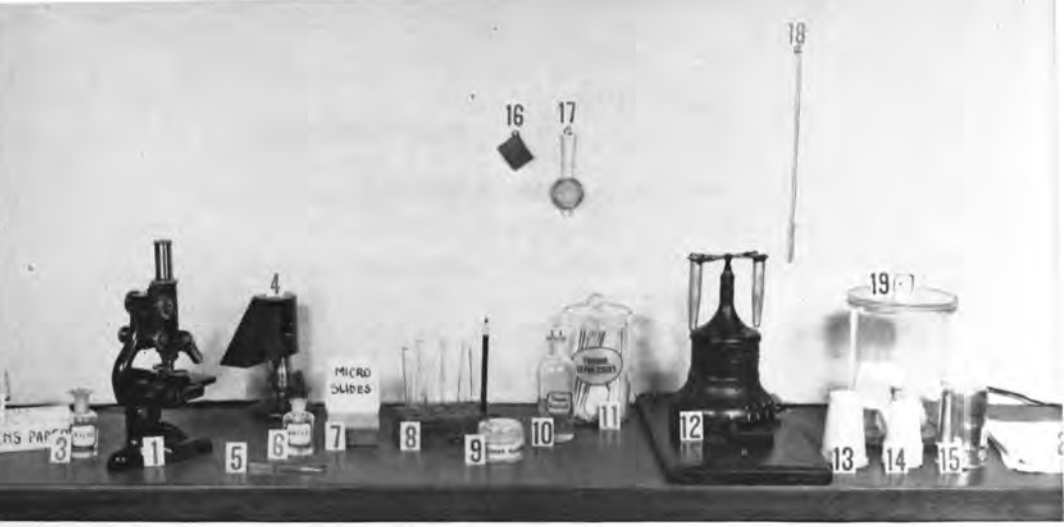

FIG. 1—Apparatus for microscopic examination of feces:

1. Microscope
2. Lens paper
3. Xylene
4. Microscope lamp
5. Coverglass forceps
6. Water-dropping bottle
7. Microslides
8. Test tube block with tubes, headed glass rods, and glass-marking pencil
9. Coverglasses
10. Flotation solution
11. Tongue depressors
12. Centrifuge
13. Large paper cups
14. Small paper cups
15. Aluminum beakers
16. Rubber test tube closure
17. Sieve
18. Test tube brush
19. Jar for waste
20. Towel

to be recommended consists of a metal shade enclosing an inside-frosted 60-watt blue bulb.

5. *Coverglass forceps.* These should always be used when handling micro coverglasses.
6. *Water-dropping bottle.* Any bottle of 30 to 60 ml. (1 to 2 oz.) capacity is suitable, when provided with a medicine dropper. Fresh tap water should be used.
7. *Microslides.* These are the standard 75 x 25 mm. (3 x 1 in.) glass slides. They should be washed and dried before using, and they may be reused repeatedly.
8. *Test tube block with tubes, headed glass rods, and glass-marking pencil.* The test tube block may easily be made by boring 12 mm. (½ in.) holes in a 4 cm. (1½ in.) thick piece of wood. Corresponding 6 mm. (¼ in.) holes are bored to hold

the headed glass rods; and a 10 cm. (3/8 in.) hole is bored to accommodate a glass-marking pencil.

The test tubes recommended are 10 cm. (4 in.) long by 12 mm. (1/2 in.) outside diameter (Fig. 2).

The headed glass rod (Fig. 2) is 5 to 7 mm. (3/16 to 1/4 in.) in diameter by 13 cm. (5 in.) in length. In making the

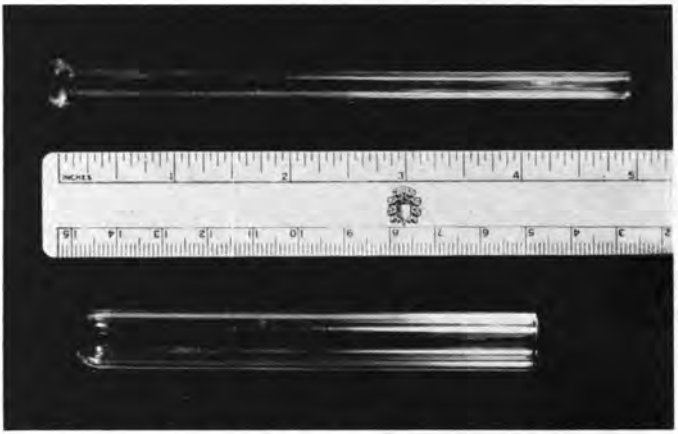

FIG. 2—Test tube **(below)** and headed glass **(above)** shown for comparative size.

head portion, one end of the rod is heated to redness in a Bunsen burner flame. The heated end is then quickly pressed against a warm, flat metal surface such as the head of a hammer, until the head portion of the rod spreads to a diameter of approximately 10 mm. (3/8 in.). After the rod has lost its softness by cooling, it is smoothed by rotating it in the flame. The glass-marking pencil is a standard laboratory item.

9. *Coverglasses.* Any 18 or 22 mm. (3/4 or 7/8 in.) square, glass or plastic coverglass is suitable. The plastic covers are more economical and require no cleaning before they are used, after which they are discarded. Coverglasses should be stored in a covered container such as a small glass dish.

10. *Flotation solution.* The preparation of this fluid has been previously described (page 5).

11. *Tongue depressors.* These are a standard item of wood supplied to the medical professions. They measure 15 cm. long by 2 cm. wide by 2 mm. thick (6 in. by 3/4 in. by 1/16 in.). They are disposable and may be stored for use in a covered glass jar.
12. *Centrifuge.* This instrument may be equipped to hold two or more tubes. The tube holders should accommodate the test tubes (Item 8) as well as the conventional 15 ml. centrifuge tubes. The centrifuge should be provided with a speed regulating switch so that approximately 1,500 revolutions per minute may be maintained. The motor should be of the specifications suitable to the electric current that is available. An electric timer switch, attached to the centrifuge line, may be added in order to shut off the current automatically at the end of the centrifuging period. Angle-type centrifuges are not suitable for the preparation of feces for parasite diagnosis.
13, 14, and 15. *Large paper cups, small paper cups, and aluminum beakers.* Any of these, or similar containers may be used in preparing the fecal samples. The large paper cups have a capacity of 225 ml. (8 oz.). The small paper cups are 90 ml. (3 oz.) capacity; being more economical but less durable than the larger size. It is advisable to discard used paper cups. The aluminum beakers hold 300 ml. (10 oz.). They are comparatively economical because they should last for several years and are easily cleaned.
16. *Rubber test tube closure.* This item may be made from a discarded automobile inner tube, pieces from which are cut approximately 5 cm. (2 in.) square. A hole is punched in one corner so that it may be hung up to dry after it is rinsed clean.
17. *Sieve.* The most suitable sieve is a tea-strainer made of metal wire, approximately 30 mesh to the inch (25 mm.).
18. *Test tube brush.* The brush should be approximately 8 cm. (3 in.) long by 12 mm. (1/2 in.) in diameter. The bristles should be stiff.
19. *Jar for waste.* Any convenient receptacle having a lid closure may be used. It may contain a disinfectant solution.
20. *Towel.* Smooth cotton or linen towels are used to dry the utensils. Paper towels are a convenience for drying the hands.

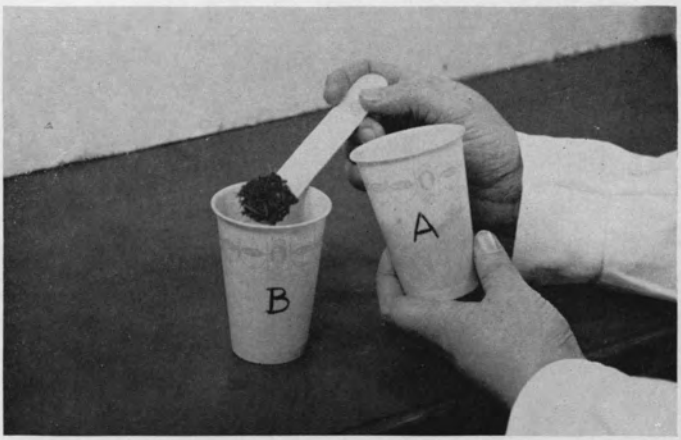

FIG. 3—Transferring approximately 1 gm. of feces from the collecting container **A** to the mixing container **B**.

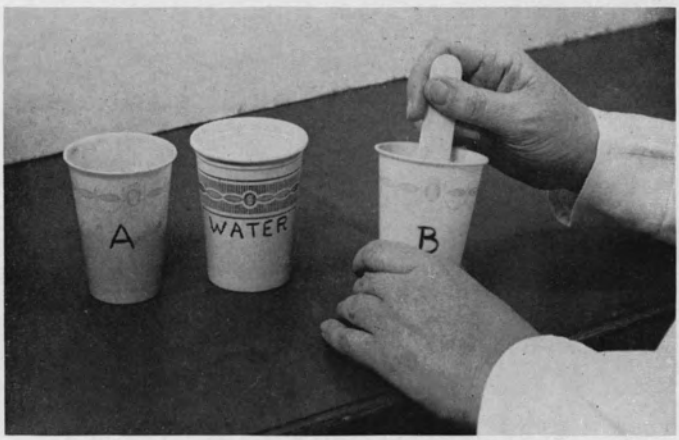

FIG. 4—Suspending the fecal sample in cold water in container **B**.

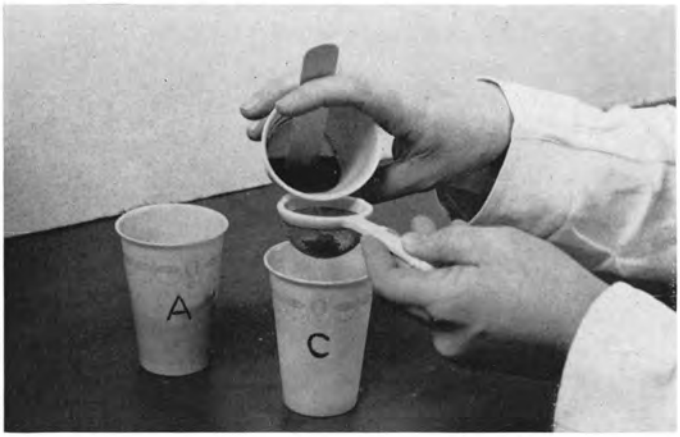

FIG. 5—The watery portion of container **B** is passed through the sieve into container **C.**

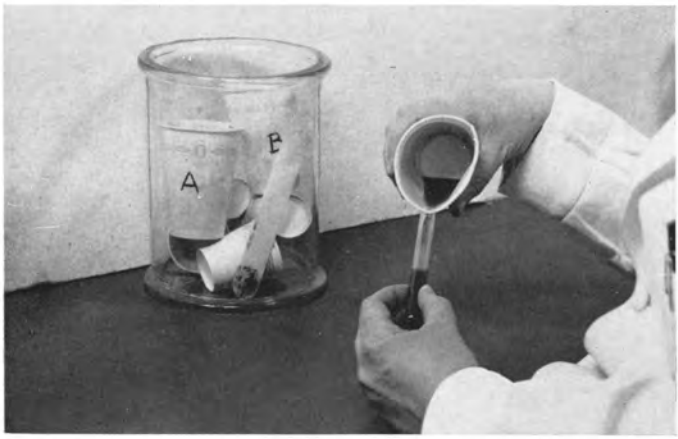

FIG. 6—Transferring sieved feces from container **C** to the test tube. The tube should be slightly less than half filled.

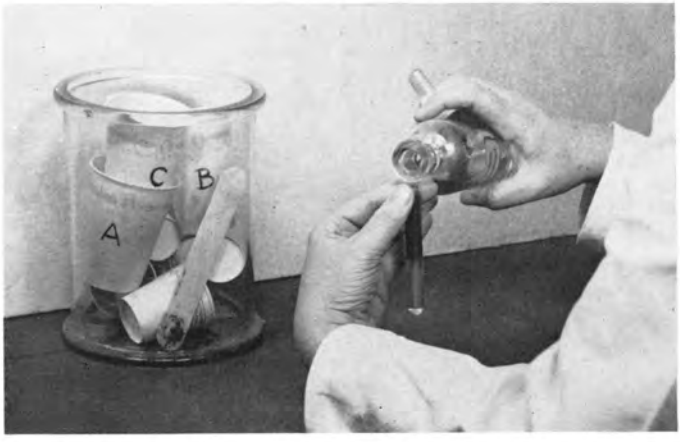

FIG. 7—Adding the flotation solution to the fecal sample in the test tube, leaving about one-fourth inch space at the top of the tube.

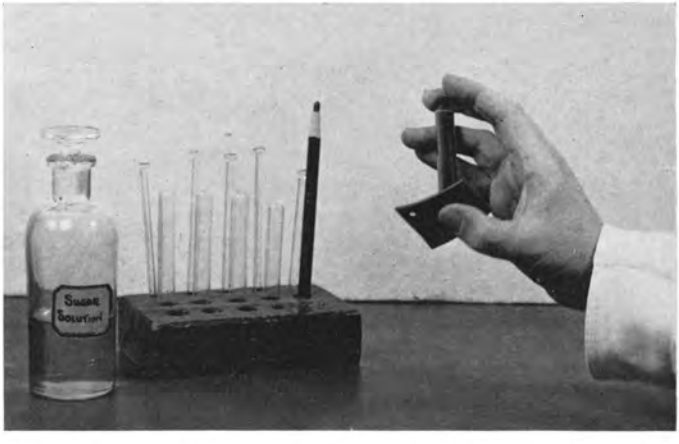

FIG. 8—Mixing the contents of the test tube with the rubber closure applied.

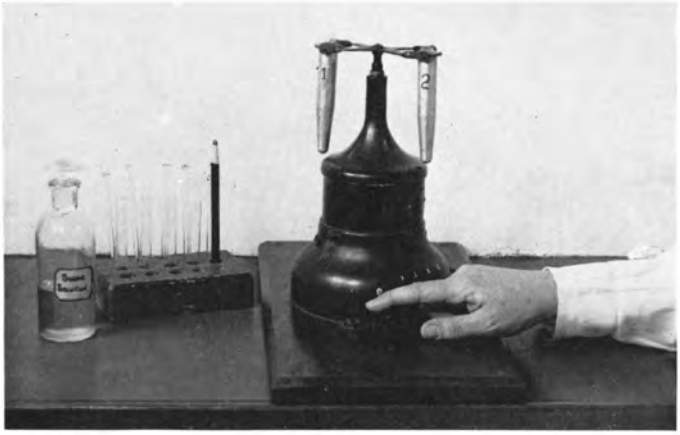

FIG. 9—Centrifuge the sample at approximately 1,500 revolutions for 3 minutes.

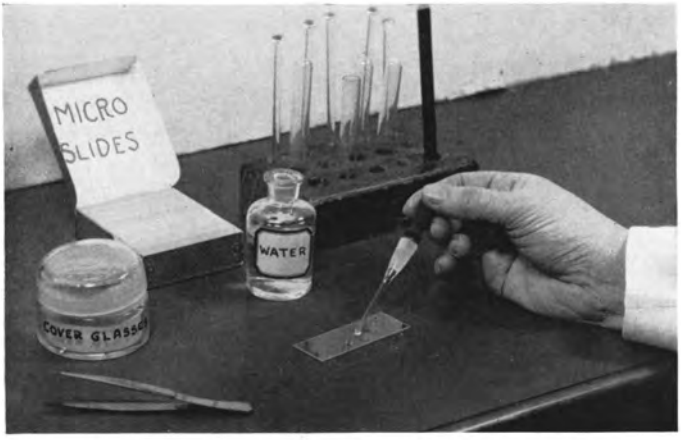

FIG. 10—Placing a drop of water on a microslide.

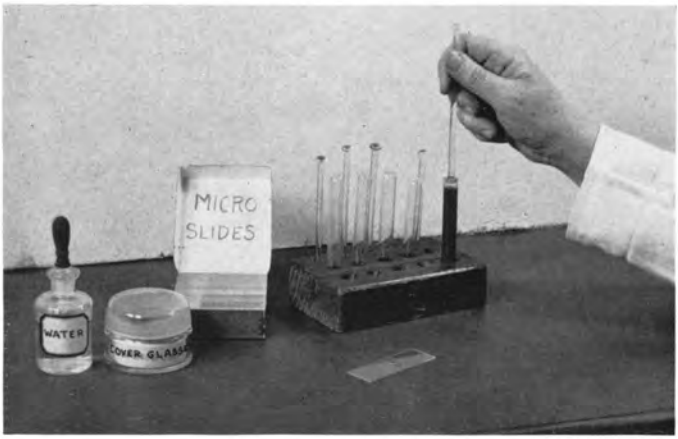

FIG. 11—Removing a drop of fluid, by means of a headed glass rod, from the surface of the centrifuged specimen.

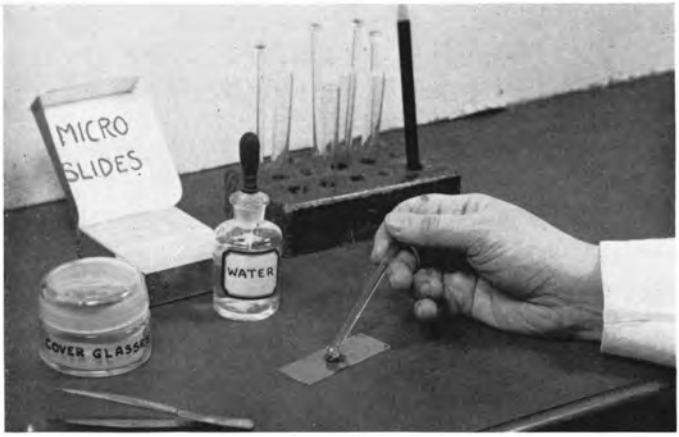

FIG. 12—Transferring the material from the headed glass rod to the drop of water on the microslide.

FIG. 13—Applying the coverglass, using forceps. DO NOT PRESS DOWN!

TECHNIQUE FOR A QUALITATIVE CONCENTRATION METHOD OF FECAL EXAMINATION

1. Transfer approximately 1 gram of feces from the collection container to a mixing cup (Fig. 3).
2. Add small quantities of cold water until stirring results in a watery suspension thin enough to pour (Fig. 4). Too much water will decrease the chance of finding parasite forms.
3. The watery suspension of feces is poured through the sieve into a second container (Fig. 5). The debris left in the sieve is discarded and the sieve is immediately cleaned in running water (preferably hot) before the contents have a chance to dry.
4. The sieved sample is briefly agitated to mix it thoroughly before pouring it into a test tube. The tube should be filled to slightly below the halfway mark (Fig. 6).
5. To the sample in the tube there is added sugar solution to fill the tube to within one-fourth inch (6 mm.) of the top (Fig. 7). Avoid contaminating the opening of the sugar solution bottle.

6. Mix the contents by closing the tube with the rubber thumb protector, then invert the tube some five or six times (Fig. 8). The rubber closure is immediately rinsed off and hung up to dry.
7. Place the tube in the centrifuge. If necessary, place balancing tubes containing water in the centrifuge carrier. Centrifuge the specimen or specimens for three minutes at approximately 1,500 revolutions per minute (Fig. 9). An automatic electric timer switch is very convenient in carrying out this step in the technique.
8. While the centrifuge is in operation, a microslide is placed on the table and a drop of water is centered on it (Fig. 10). Also a clean, headed glass rod, coverglass forceps, and a coverglass are made available. The test tube is transferred from the centrifuge to the test tube holder, care being taken not to agitate the contents.
9. Transfer a drop of sample from the test tube to the drop of water on the microslide (Fig. 11). To do this properly, hold the headed glass rod vertically over the tube, resting the elbow on the table. Slowly lower the head of the glass rod onto the surface of the sample; then quickly withdraw the rod without making contact with the inside of the tube. This operation may require some practice. Then hold the glass rod at about a 45 degree angle and rotate the headed end in the drop of water on the microslide (Fig. 12), thus washing off any parasite eggs or oocysts adhering to the rod. Replace the rod in the test tube block. It should be rinsed and dried before further use.
10. Pick up a coverglass by means of the coverglass forceps. Lower one edge of the coverglass onto the slide near the drop of suspension; then release the forceps as the coverglass is gently lowered onto the drop. The fluid should spread out evenly under the coverglass (Fig. 13). Too rapid an application of the coverglass will probably result in the formation of air bubbles, which may interfere with the microscopic examination of the specimen. *Avoid pressure on the coverglass.*

11. Place the slide on the stage of the microscope so that the near right-hand corner of the coverglass is centered under the low power (16 mm.) objective. Focus on this corner. Adjust the substage condenser and diaphragm of the microscope so as to see a distinct image of the suspension under the coverglass. Using the low power magnification (x 100), systematically move the microslide back and forth until the entire area of the coverglass has been scanned (Fig. 14). Objects having a resemblance to parasite forms may be

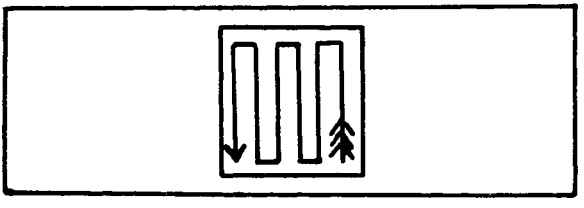

FIG. 14—The material under the coverglass should be systematically examined at 100 magnification according to this diagram.

centered and examined under the high power (x 400) dry lens (4 mm.). Always return to the low power (x 100) lens for further search of the specimen.

If worm eggs and coccidial oocysts are present in the same specimen, the coccidia, being the smaller, tend to float upward until they rest directly beneath the coverglass. Therefore, when the worm eggs are in focus under high power (x 400), the coccidia may be out of focus and vice versa. Both types of parasitic forms may be brought clearly into focus by turning the fine-adjustment knob of the microscope.

MODIFIED FLUKE EGG TECHNIQUE

From the early reference of Cobb (1904) to the latest work of Dennis, Stone, and Swanson (1954), workers have attempted to find a simple, rapid method for demonstrating fluke ova in feces.

Nearly all the investigators have tried some type of flotation technique but were unable to obtain consistent results because of the collapsibility of the ova in solutions of high specific gravity. In the limited number of times we have demonstrated canine lung

fluke (*Paragonimus westermanni*) ova in fecal samples, we have used the modification of Sheather's sugar solution technique and have experienced little or no difficulty with the ova collapsing.

The technique of Dennis, Stone, and Swanson (1954) appears to be a relatively simple quantitative method for demonstrating fluke ova. It requires about one-half hour to perform. The following modification of this *quantitative* method is useful for *qualitative* clinical diagnosis.

Reagents for Fluke Egg Technique

1. Detergent solution:
   Liquid detergent ("Joy," or "Glim," or similar) .. 5 cc.
   Tap water .....995 cc.
   1% alum (aluminum potassium sulfate U.S.P.) ... 8 drops
2. Tincture of iodine U.S.P.

Apparatus for Fluke Egg Technique

1. Fecal containers. Samples up to 500 gm. (1 lb.) may be used.
2. Wooden tongue blades for stirring the sample.
3. A tin-coated or zinc-coated funnel, 9 cm. (3½ in.) in diameter with 80 mesh copper screen soldered 25 mm. (1 in.) from the top.
4. Test tubes of 30 cc. (1 oz.) capacity, dimensions 150 x 18 mm. (6 x ¾ in.).
5. Test tube rack or block for holding tubes.
6. Stirring rod (glass or metal), 20 cm. (8 in.) long.
7. Centrifuge tubes, capacity 50 cc. (1.7 oz.).
8. Centrifuge tube rack or block.
9. Wash bottle.
10. Pipette, 2 cc. capacity.
11. Microslides, 75 x 25 mm. (3 x 1 in.).
12. Coverglasses, 22 mm. (¾ in.) diameter.
13. Filter pump (using faucet water pressure, such as the Richards filter pump); or a decanting bottle (using mouth suction); or a bulb syringe of about 30 cc. (1 oz.) capacity.
14. Clinical microscope.

Procedure for Fluke Egg Technique

1. Using a tongue blade, mix the fecal sample thoroughly; and, if it is very dry, add cold tap water to form a pasty mass.

2. Place about 1 gm. of the mixed feces in a 30-cc. (1-oz.) test tube.
3. Add 15 cc. (½ oz.) detergent solution. Mix well with a stirring rod. To avoid sudsing, do not shake.
4. Strain the mixture through the funnel-strainer into a 50-cc. (1.7-oz.) centrifuge tube.
5. Rinse the test tube with more detergent solution and strain.
6. Pour enough detergent solution in a flooding, swirling motion through the feces in the funnel-strainer to fill the centrifuge tube.
7. Allow the tubed mixture to stand for 5 to 15 minutes.
8. Decant three-fourths of the liquid portion from the centrifuge tube.
9. Rewash the fecal material in the funnel-strainer to refill again the centrifuge tube, in order to obtain any ova trapped previously. Discard the funnel contents.
10. Again allow the tubed mixture to stand for 5 to 15 minutes.
11. Again decant all liquid down to about 2 to 3 cc. Do not disturb the sediment.
12. Add 1 to 3 drops tincture of iodine to the sediment, allowing the tube to stand for 2 to 5 minutes.
13. Using a pipette, transfer the sediment to one or more microslides and apply coverglasses.
    (Note: Dennis, Stone, and Swanson recommend placing all of the sediment in a standard Petri dish, adding tap water to make 15 to 20 cc. and searching for ova with a binocular dissecting microscope magnifying 18 x or higher.)
14. Search the sediment on the slide or slides, using a clinical microscope magnifying 100 x.

C. *Quantitative methods of fecal examination.* Various techniques have been proposed for the determination of the *number* of parasite eggs or coccidial oocysts per gram of feces. Such methods are of value in the study of parasite life cycles, or in determining the effects of experimental therapy for the removal of gastro-intestinal parasites. Quantitative fecal techniques are of little value in clinical diagnosis; therefore, such methods are not included in this publication.

References for Section One will be found on pages 169 to 190.

HORSE

FIG. 15—Ova of **Paranoplocephala mamillana,** the small tapeworm of the horse. x 100.

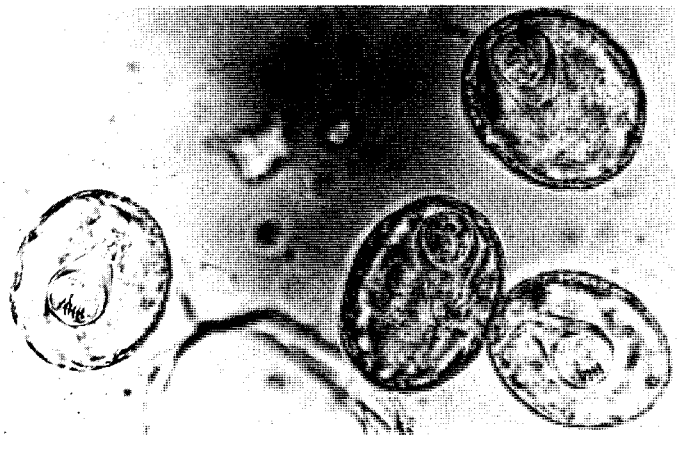

FIG. 16—Ova of **Paranoplocephala mamillana.** The eggs enclose a pear-shaped embryo having six hooklets. x 410.

HORSE

FIG. 17—Ova of **Parascaris equorum,** the ascarid of the horse. The egg shells are rough and thick, and are yellow to brown in color. Also included are three strongyle ova. x 100.

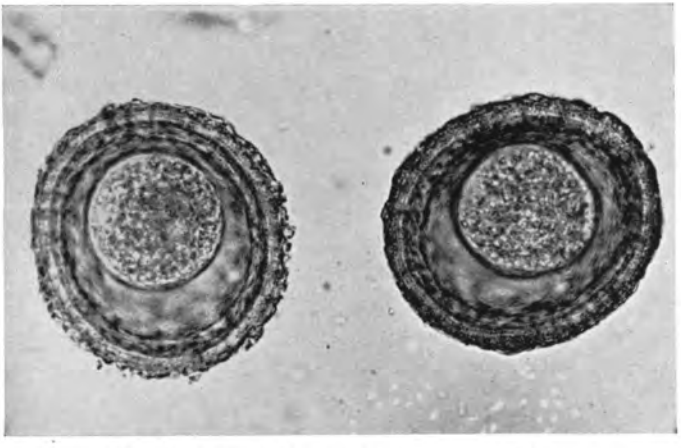

FIG. 18—Ova of **Parascaris equorum.** x 410.

HORSE

FIG. 19—Ova from several species of strongyles of equines. Thirty-nine species of these nematodes have been reported from the large intestine of horses, asses, and mules in North America. The eggs of all species are similar. x 100.

FIG. 20—Ova from two species of strongyles of equines. x 410.

HORSE

FIG. 21—Ova of **Draschia megastoma,** one of the three larger gastric nematodes of the horse. These eggs are elongated; embryonated when laid and are surrounded by a very thin membranous shell. x 100.

FIG. 22—Ova of **Draschia megastoma.** x 410.

HORSE

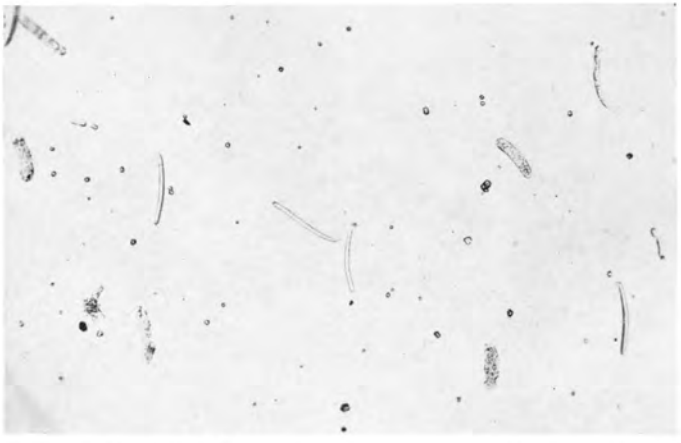

FIG. 23—Ova of **Habronema muscae,** one of the three larger gastric nematodes of the horse. These ova are elongated; embryonated when laid and surrounded by a very thin membranous shell. x 100.

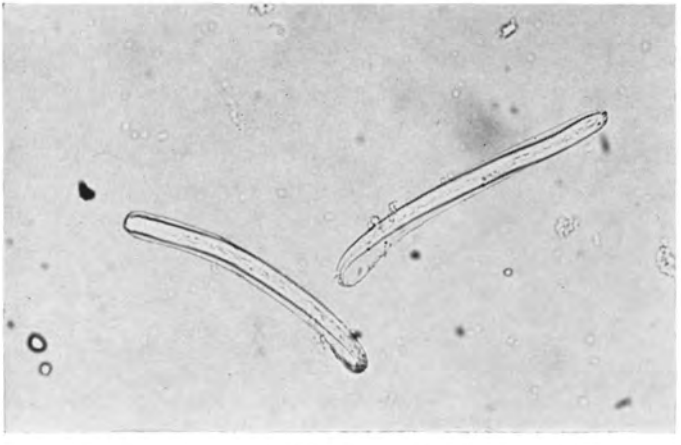

FIG. 24—Ova of **Habronema muscae.** x 410.

HORSE

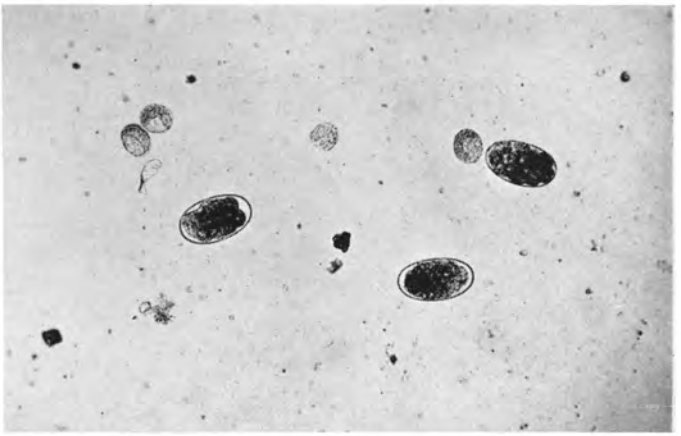

FIG. 25—Ova of **Strongyloides westeri,** the intestinal threadworm of the horse. The three larger eggs are those of strongyles. x 100.

FIG. 26—Ova of **Strongyloides westeri.** These eggs are embryonated when laid. x 410.

HORSE

FIG. 27—Ova of **Oxyuris equi,** the rectal worm of the horse. These eggs may be found in the feces but the examination of anal scrapings is a more accurate method of diagnosis. x 100.

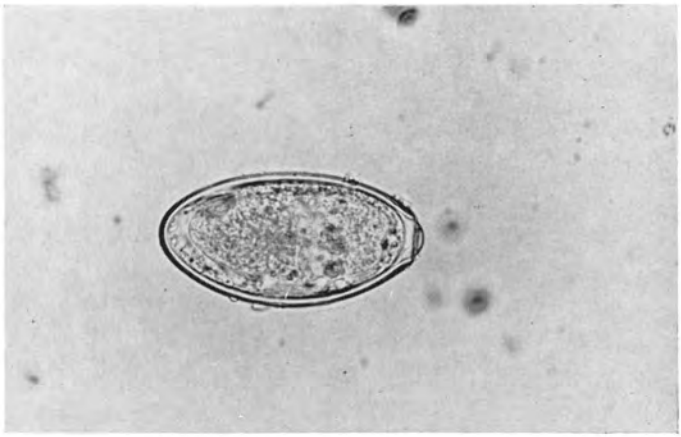

FIG. 28—Ovum of **Oxyuris equi.** Note the operculum (cap) at one end. x 410.

HORSE

FIG. 29—Ova and larvae of **Dictyocaulus arnfieldi,** the lungworm of horses. These were taken from bronchial exudate but they may also be found in feces. The eggs are embryonated when laid. x 100.

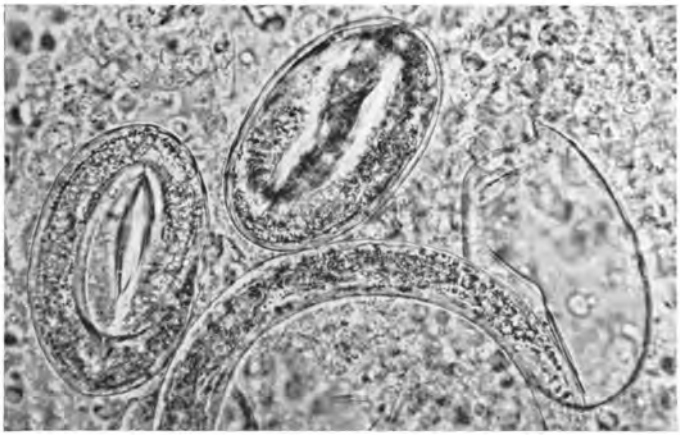

FIG. 30—Ova, part of a larva and an empty egg shell of **Dictyocaulus arnfieldi.** x 410.

CATTLE

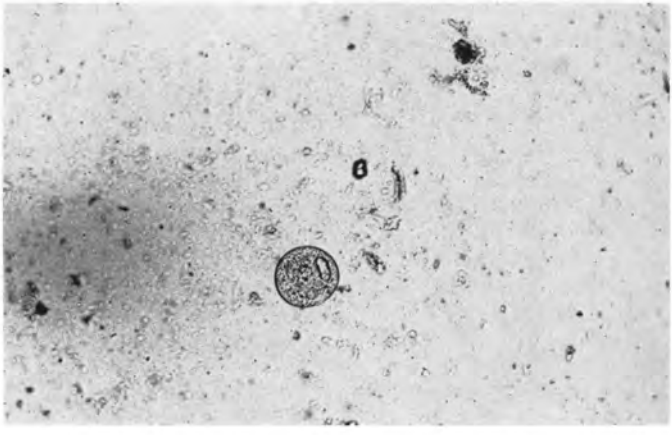

FIG. 31—A cyst of **Buxtonella sulcata** of cattle. This is the resting stage of a large ciliated protozoon of the caecum of cattle. Nothing is known regarding its possible pathogenicity. It is commonly found in cattle feces. x 100.

FIG. 32—**Buxtonella sulcata** cyst. x 410.

CATTLE

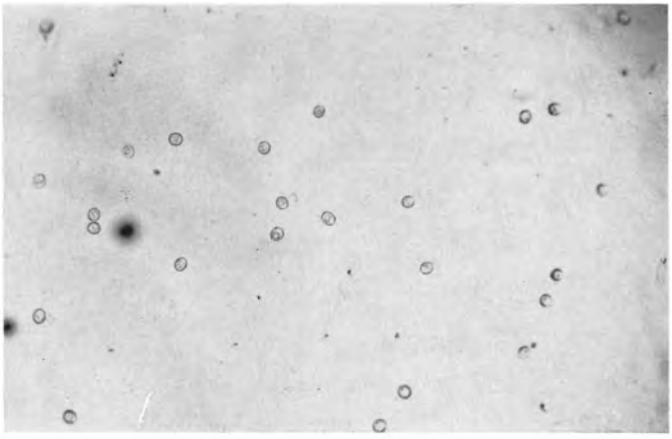

FIG. 33—Oocysts of **Eimeria zurnii,** one of the more pathogenic of the eleven species of coccidia of cattle in North America. x 100.

FIG. 34—Oocysts of **Eimeria zurnii.** x 410.

CATTLE

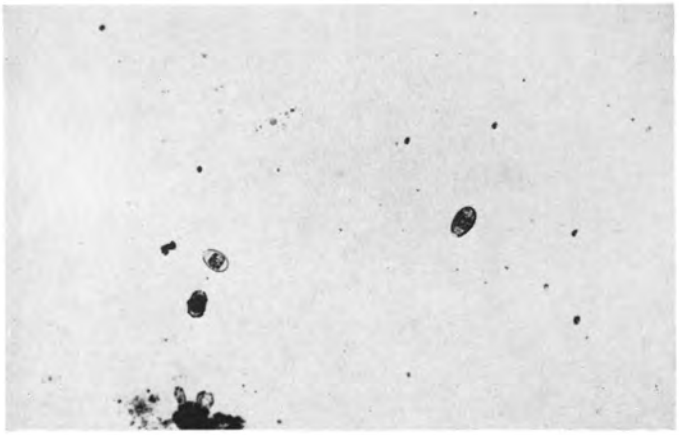

FIG. 35—Oocysts of **Eimeria auburnensis,** a coccidium of cattle. The color is yellowish-brown. One smooth-walled and two rough-walled cysts are shown. x 100.

FIG. 36—Oocysts of **Eimeria auburnensis.** Smooth-walled form at the left; rough-walled form at the right. x 410.

SHEEP, GOAT

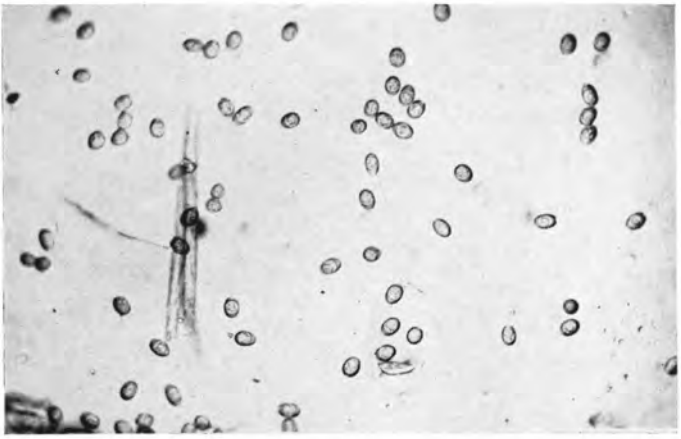

FIG. 37—Oocysts of **Eimeria arloingi,** one of the more pathogenic of the eight species of coccidia of sheep and goats in North America. The color varies from pale yellow to yellowish-green. x 100.

FIG. 38—Oocysts of **Eimeria arloingi.** A polar cap is present at one end of the cyst. x 410.

SHEEP, GOAT

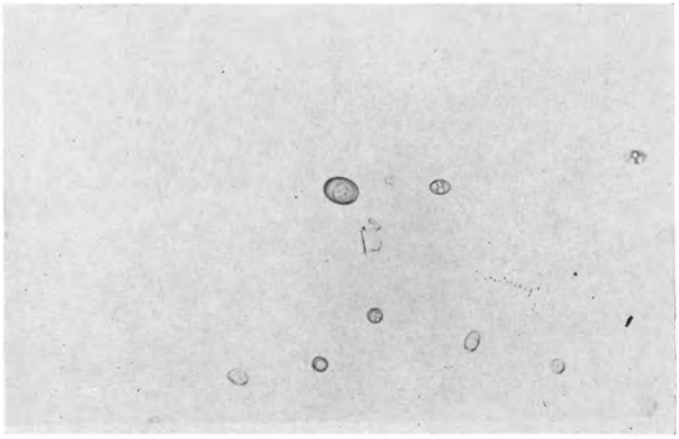

FIG. 39—Oocysts of **Eimeria intricata** and **Eimeria arloingi,** coccidia of sheep and goats. The large oocyst is that of **E. intricata,** the color of which is dark brown. x 100.

FIG. 40—Oocysts of **Eimeria intricata (right)** and of **Eimeria arloingi (left).** x 410.

CATTLE

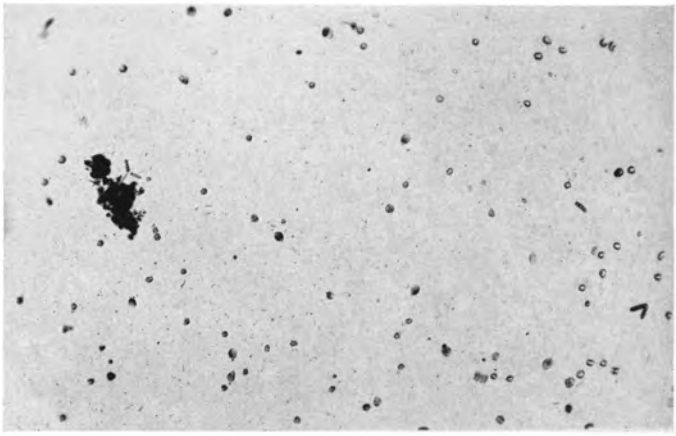

FIG. 41—**Giardia bovis,** a flagellate protozoan of cattle. It is motile. Similar species are found in sheep, goats, dogs, and cats. x 100.

FIG. 42—**Giardia bovis** showing the two posterior flagella. x 410.

CATTLE

FIG. 43—**Giardia bovis** showing the ventral sucking disc and the two nuclei. x 410.

FIG. 44—**Giardia bovis,** oblique view to show the ventral concavity and the posterior flagella. x 410.

CATTLE, SHEEP, GOAT

FIG. 45—Ova of **Fasciola hepatica,** the common liver fluke of cattle, sheep, and goats. x 100.

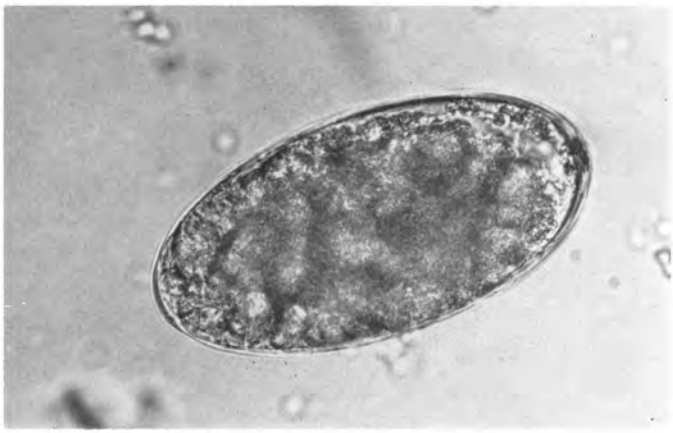

FIG. 46—Ovum of **Fasciola hepatica.** x 410.

CATTLE

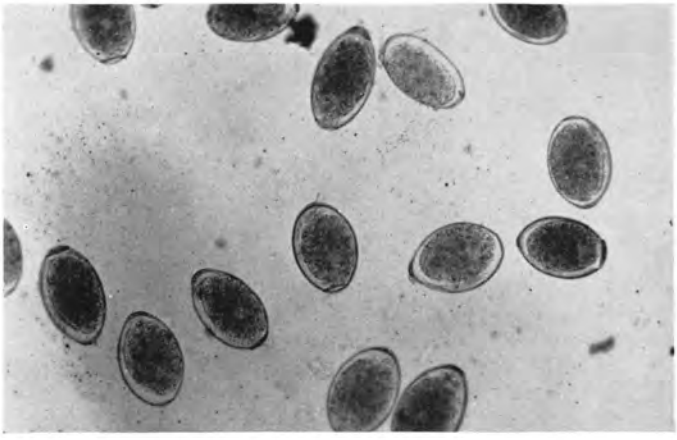

FIG. 47—Ova of **Fascioloides magna,** the large American liver fluke of cattle. The eggs are heavy and sink in sugar solution. x 100.

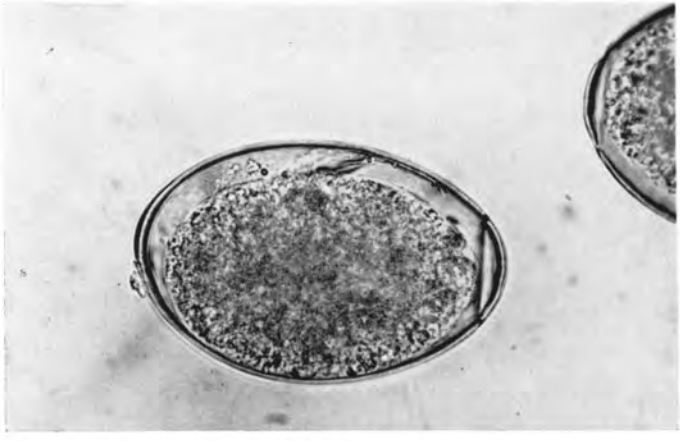

FIG. 48—Ovum of **Fascioloides magna.** Note the operculum at one end. x 410.

CATTLE, SHEEP

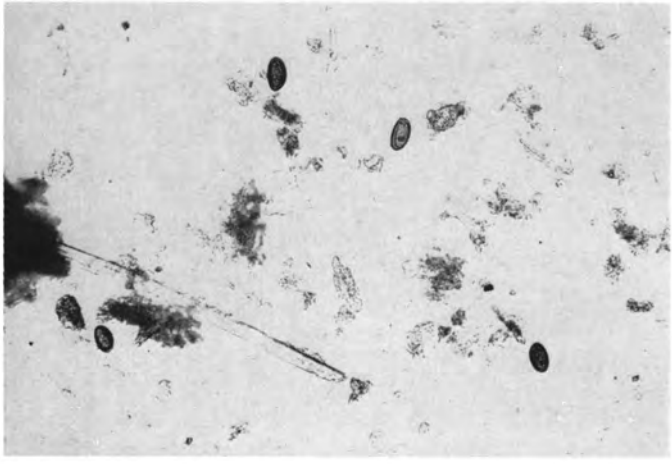

FIG. 49—Ova of **Dicrocoelium dendriticum,** the lancet liver fluke of cattle, sheep, deer, and woodchuck. x 100.

FIG. 50—Ovum of **Dicrocoelium dendriticum.** x 410.

CATTLE, SHEEP, GOAT

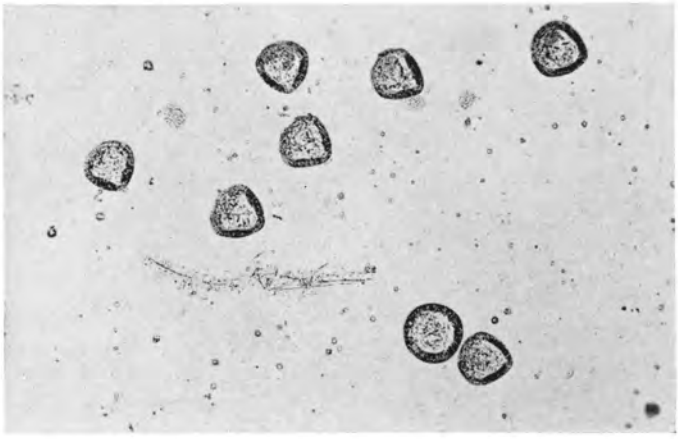

FIG. 51—Ova of **Moniezia expansa,** a tapeworm of cattle, sheep, and goats. x 100.

FIG. 52—Ovum of **Moniezia expansa.** Note the pear-shaped embryo which contains six hooklets. x 410.

SHEEP, GOAT

FIG. 53—A packet containing ova of **Thysanosoma actinioides,** the fringed tapeworm of sheep and goats. These usually leave the host within the tapeworm segments, hence are seldom found on routine fecal examination. x 100.

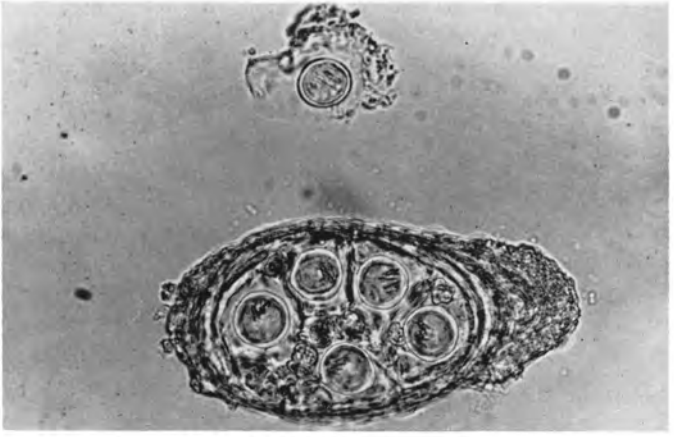

FIG. 54—A packet containing ova of **Thysanosoma actinioides.** Five ova are visible within the packet and one ovum is free. x 410.

CATTLE, SHEEP, GOAT

FIG. 55—Ovum of **Haemonchus contortus,** the common or "twisted" stomach worm of cattle, sheep, and goats. x 100. (See footnote)

FIG. 56—Ovum of **Haemonchus contortus.** x 400.

Note: Cattle, sheep, and goats of North America are reported to harbor 39 species of nematode worms in the alimentary canal. The eggs of the following 24 species are very similar to those seen in Figs. 55 and 56: Common stomach worms (2 species); trichostrongylid worms (4 species); cooperid worms (5 species); nodule worms (3 species); hookworms (2 species); ostertagid stomach worms (7 species); large-mouthed bowel worm (1 species).

CATTLE, SHEEP, GOAT

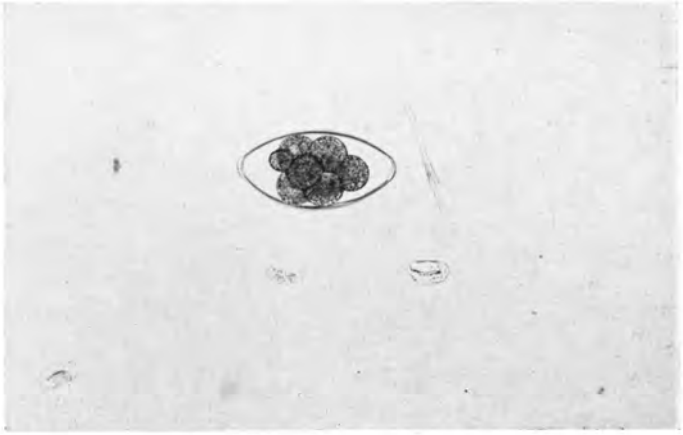

FIG. 57—Ovum of **Nematodirus spathiger,** an intestinal nematode of cattle, sheep, and goats. The two small, embryonated ova are those of **Strongyloides papillosus.** x 100.

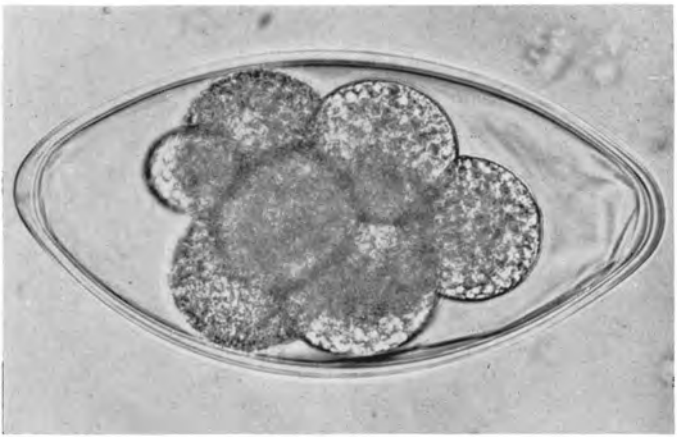

FIG. 58—Ovum of **Nematodirus spathiger.** The embryonic mass is in the eight-celled stage. Note the thickened shell at the poles. x 400.

SHEEP, GOAT

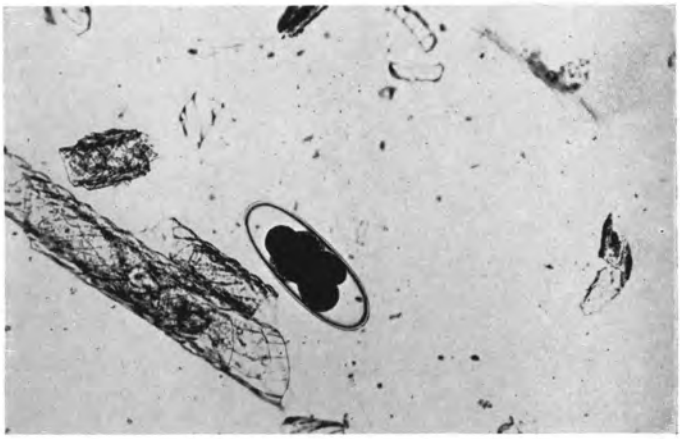

FIG. 59—Ovum of **Marshallagia marshalli**, a stomach worm of sheep and goats. x 100.

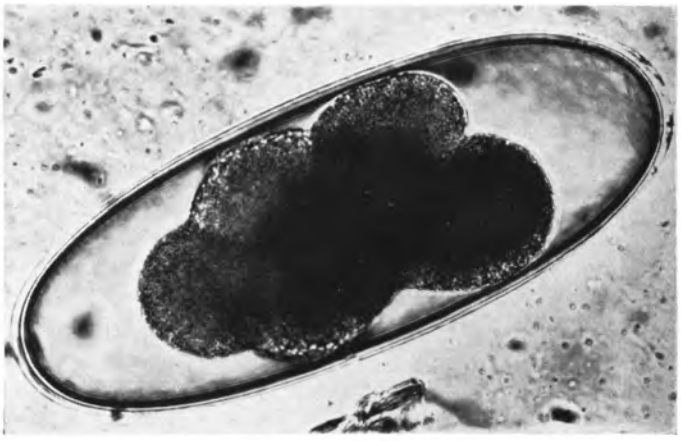

FIG. 60—Ovum of **Marshallagia marshalli.** x 410.

CATTLE, SHEEP, GOAT

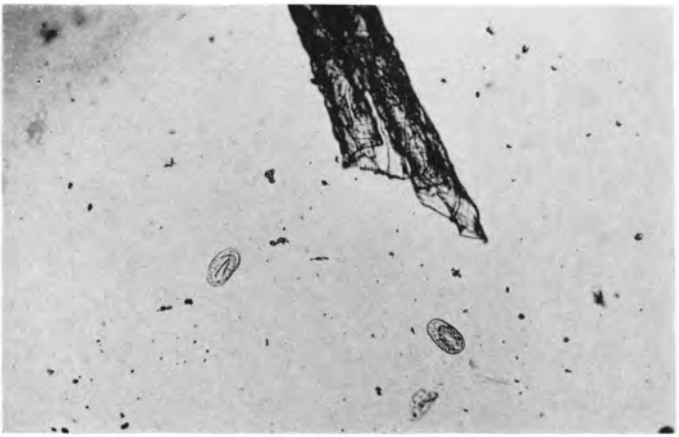

FIG. 61—Ova of **Strongyloides papillosus,** a threadworm of the small intestine of cattle, sheep, and goats. x 100.

FIG. 62—Ovum of **Strongyloides papillosus.** The eggs of this nematode are embryonated when laid. x 410.

CATTLE

FIG. 63—Ova of **Neoascaris vitulorum,** the ascarid of cattle. x 100.

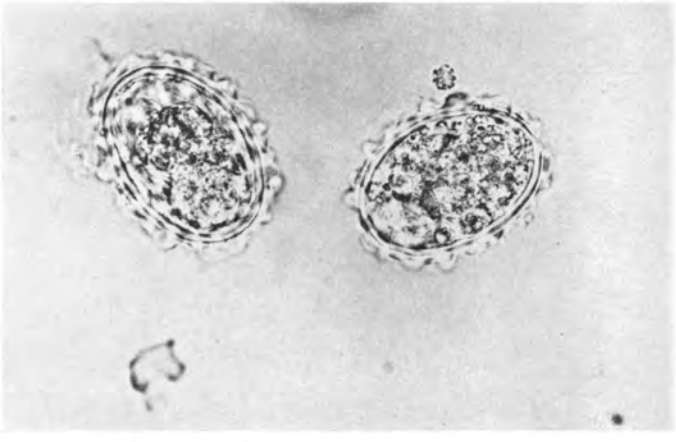

FIG. 64—Ova of **Neoascaris vitulorum.** x 410.

SHEEP, GOAT

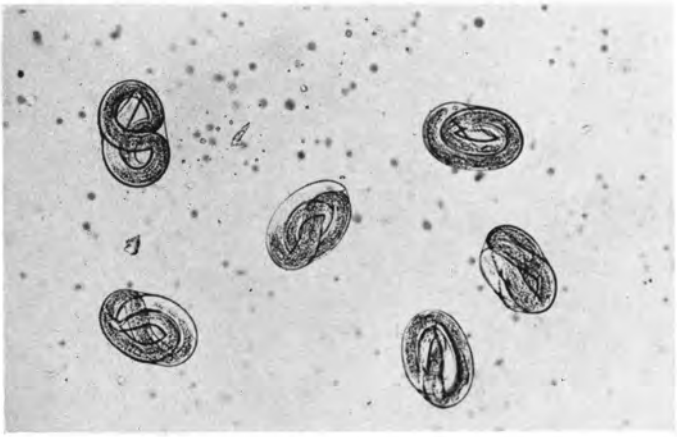

FIG. 65—Ova of **Dictyocaulus filaria,** a lungworm of sheep and goats. These were taken from bronchial exudate, but they may also be found in feces. The eggs are embryonated when laid. x 100.

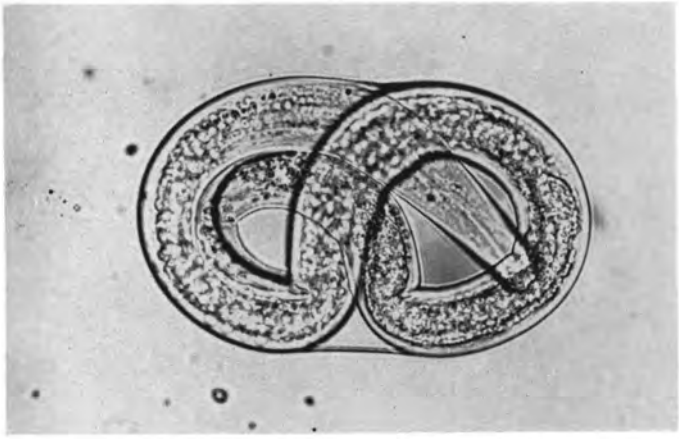

FIG. 66—Ovum of **Dictyocaulus filaria.** x 410.

CATTLE

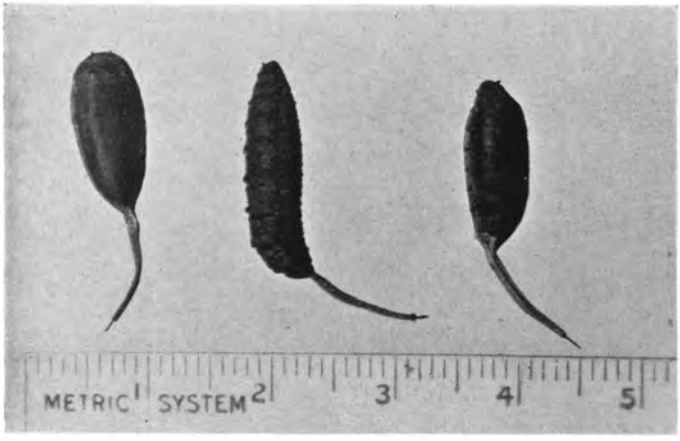

FIG. 67—Pseudoparasite. Rat-tailed maggots from cattle feces. These are the larvae of harmless flies commonly known as drone flies. They belong in the dipterous family Syrphidae. x 1.7.

SWINE

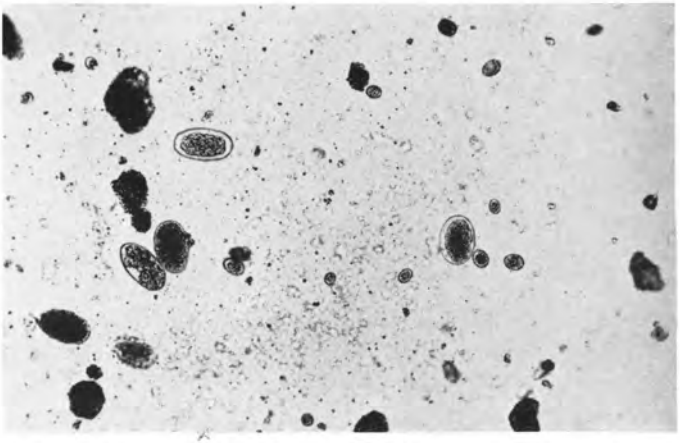

FIG. 68—Oocysts of **Eimeria** sp., coccida of swine. Several species are shown. The ova are those of **Oesophagostomum** sp., one of the nodule worms. x 100.

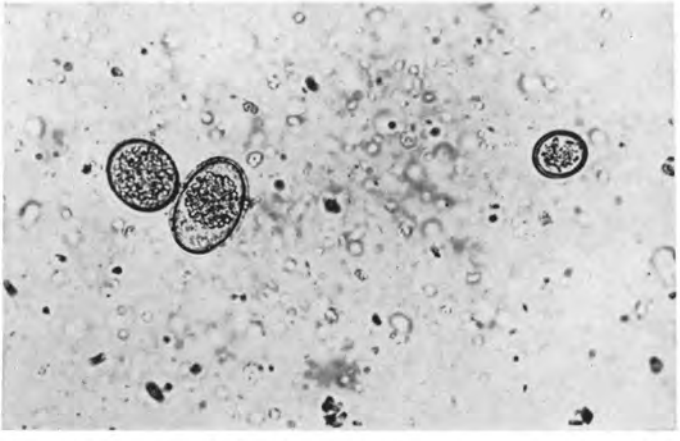

FIG. 69—Oocysts of **Eimeria** sp. Three species are shown. x 410.

SWINE

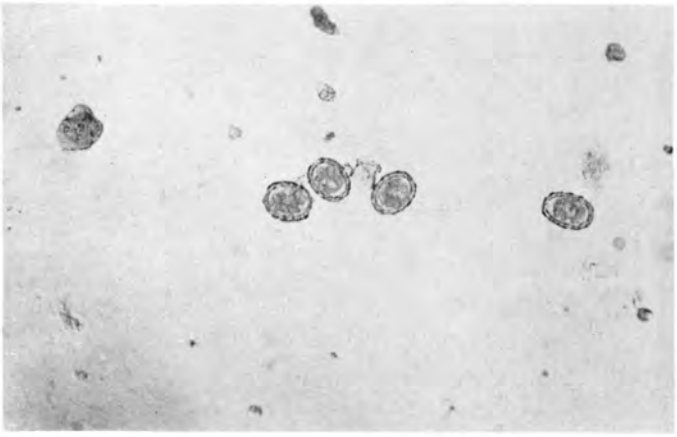

FIG. 70—Ova of **Ascaris lumbricoides,** the ascarid of swine. x 100.

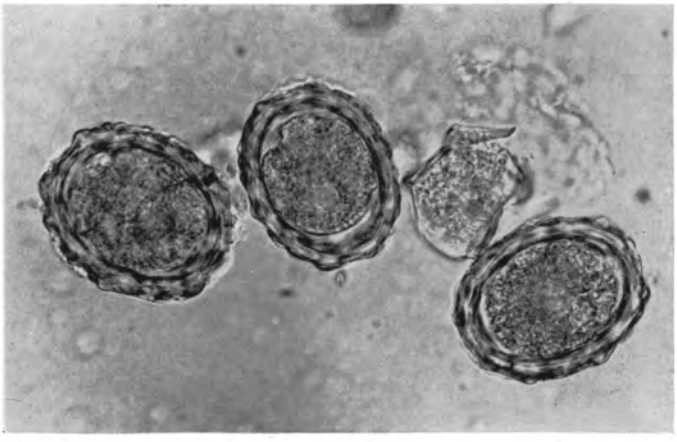

FIG. 71—Ova of **Ascaris lumbricoides.** Note the rough shell. The color is yellow. x 400.

SWINE

FIG. 72—Ova of **Macracanthorhynchus hirudinaceus,** the thorny-headed worm of swine. x 100.

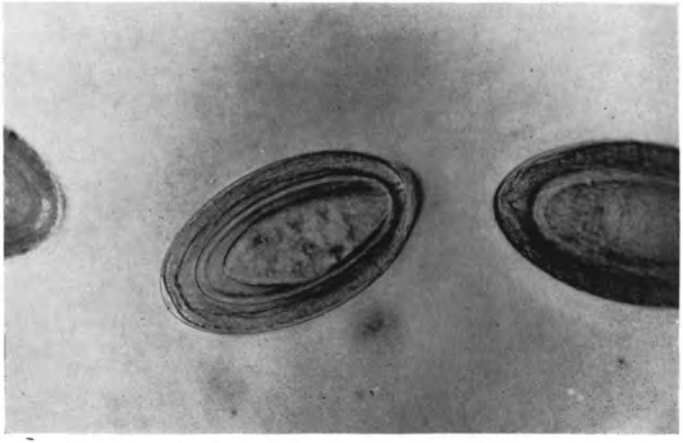

FIG. 73—Ova of **Macracanthorhynchus hirudinaceus.** The embryo is surrounded by three shells. The outer shell is dark brown. x 400.

SWINE

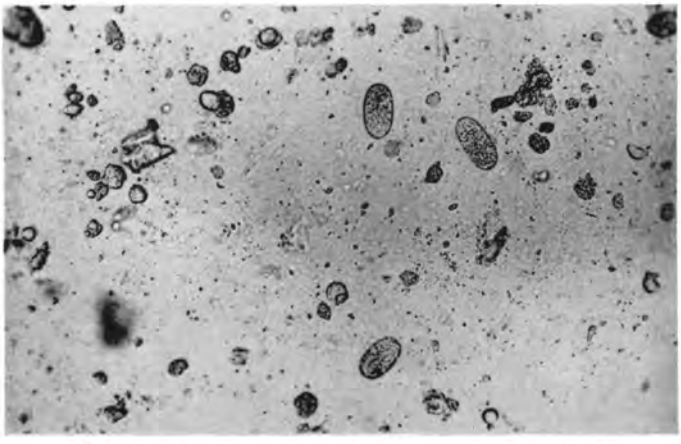

FIG. 74—Ova of **Oesophagostomum** sp., one of the four species of nodule worms of swine. x 100.

FIG. 75—Ova of **Oesophagostomum** sp. x 410.

SWINE

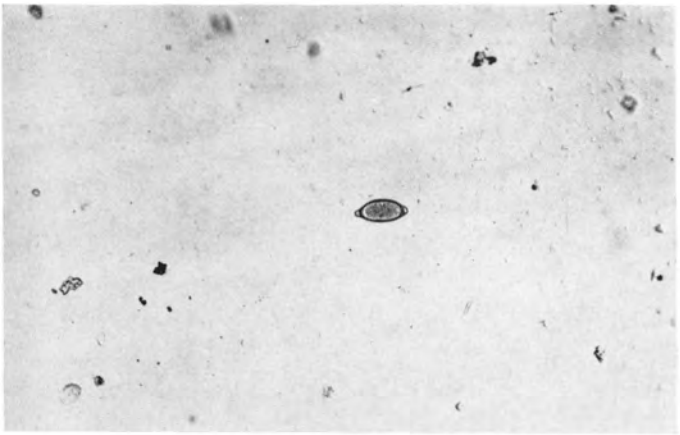

FIG. 76—Ovum of **Trichuris suis**, the whipworm of swine. x 100.

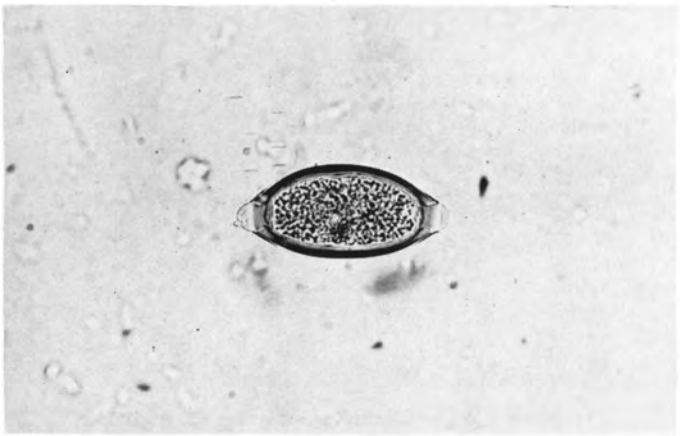

FIG. 77—Ovum of **Trichuris suis**. x 400.

SWINE

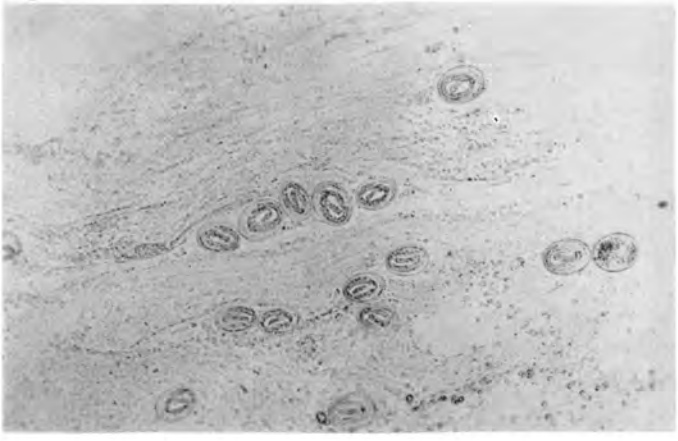

FIG. 78—Ova of **Metastrongylus apri,** one of the lungworms of swine. These were removed from the bronchial exudate but they may also be found in the feces. The eggs are embryonated when laid. x 100.

FIG. 79—Ova of **Metastrongylus apri.** x 400.

SWINE

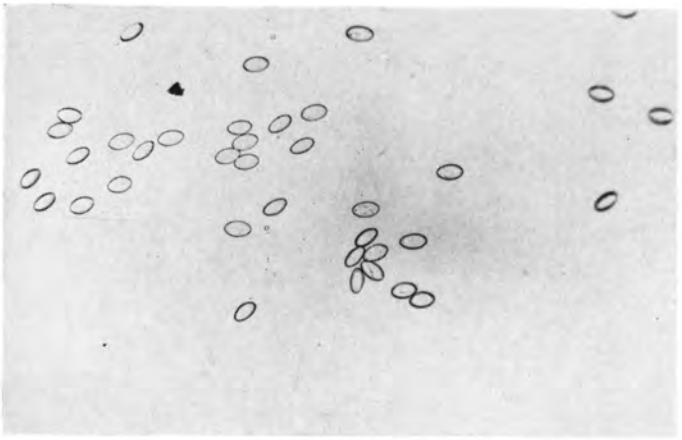

FIG. 80—Ova of **Ascarops strongylina,** one of the stomach worms of swine. x 100.

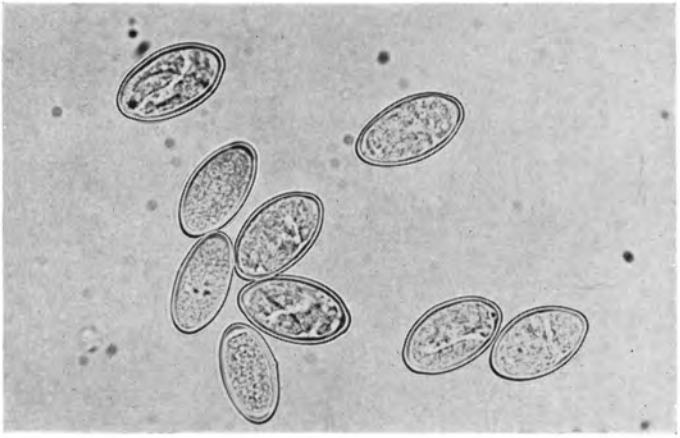

FIG. 81—Ova of **Ascarops strongylina.** x 410.

SWINE

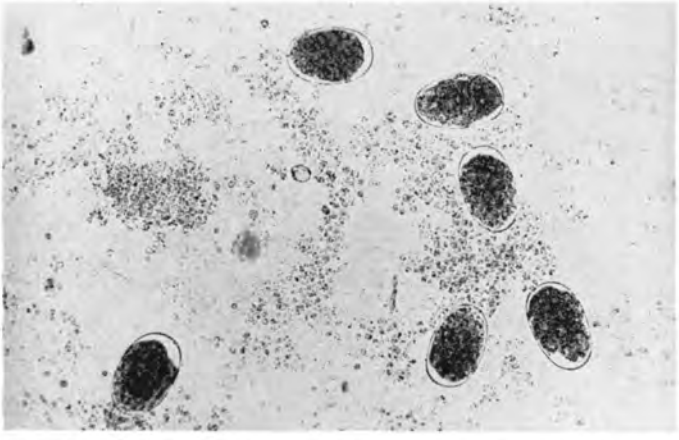

FIG. 82—Ova of **Stephanurus dentatus,** the kidney worm of swine. These eggs are found in urinary sediment and occasionally in the feces. x 100.

FIG. 83—Ovum of **Stephanurus dentatus.** x 410.

DOG, CAT, FOX

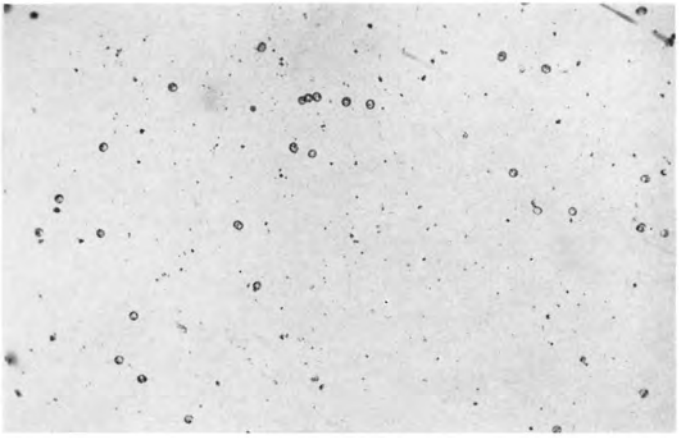

FIG. 84—Oocysts of **Isospora** sp., one of the coccidia of dogs, cats, and foxes. This is often referred to as the smaller form of **Isospora bigemina.** The oocysts are **not** sporulated when found in the feces. x 100.

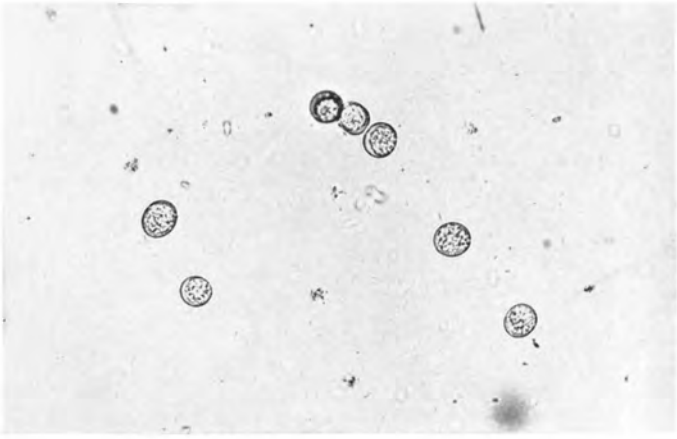

FIG. 85—Oocysts of **Isospora** sp. x 410.

DOG, CAT, FOX

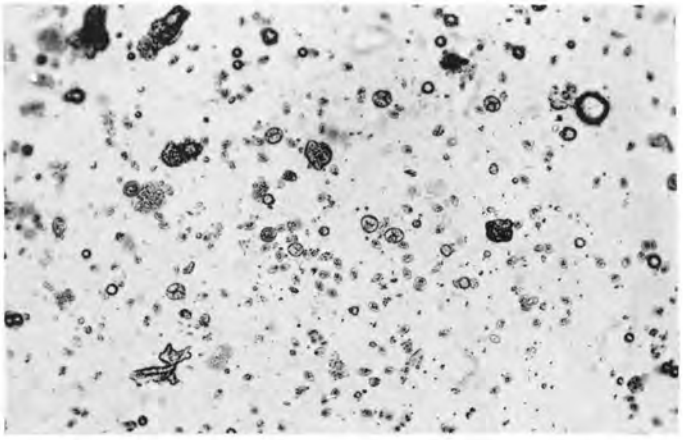

FIG. 86—Sporulated oocysts and sporocysts of **Isospora bigemina**, a coccidium of dogs, cats, and foxes. The larger oocysts seen are those of **Isospora rivolta**. x 100.

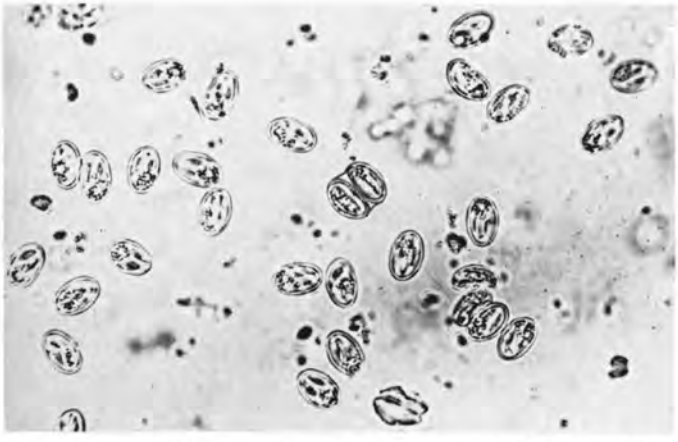

FIG. 87—Sporulated oocysts and sporocysts of **Isospora bigemina**. This coccidium is often referred to as the larger form of this species. The oocysts sporulate before leaving the body of the host and the delicate oocyst wall frequently ruptures, liberating the two sporocysts, each of which contains four sporozoites. x 410.

DOG, CAT

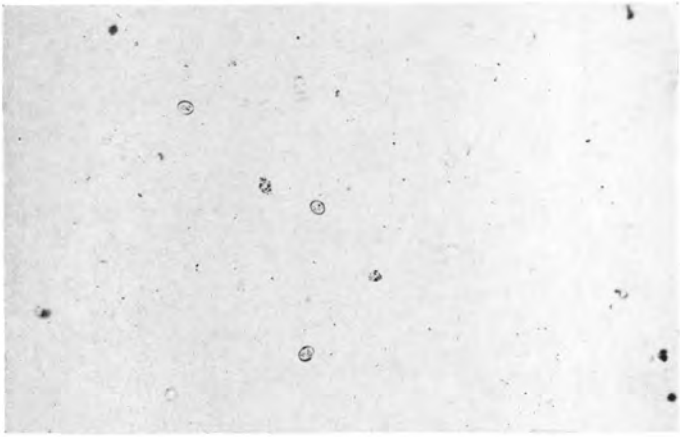

FIG. 88—Oocysts of **Isospora rivolta,** one of the coccidia of dogs and cats. These oocysts are intermediate in size between those of **I. bigemina** and **I. felis.** x 100.

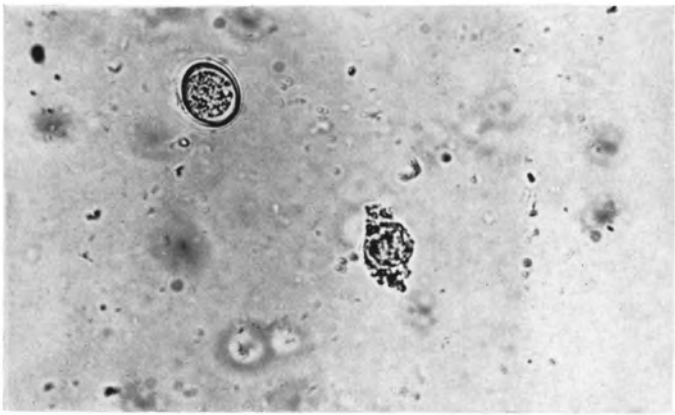

FIG. 89—Oocyst of **Isospora rivolta.** x 410.

DOG, CAT

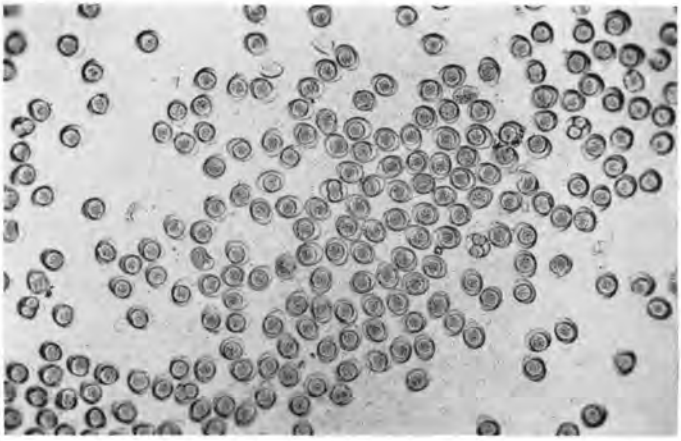

FIG. 90—Oocysts of **Isospora felis,** the largest species of the coccidia of dogs and cats. x 100.

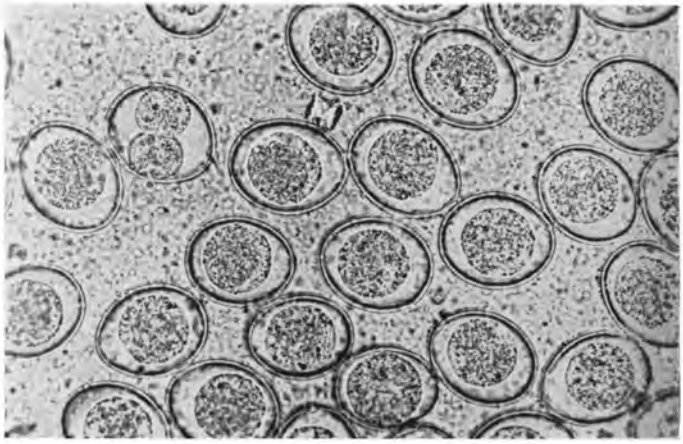

FIG. 91—Oocysts of **Isospora felis.** One shows beginning sporulation. x 410.

Note: A flagellate protozoon, **Giardia canis,** has been reported from the small intestine of dogs, and the same or a similar species from cats. Their morphology is similar to that of **Giardia bovis,** shown in Figs. 41, 42, 43, 44.

DOG, CAT, FOX, GOAT, SWINE, MINK, MUSKRAT, MAN

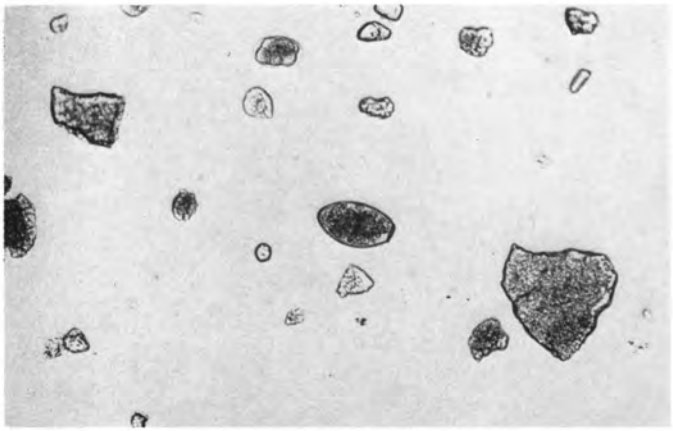

FIG. 92—Ovum of **Paragonimus westermanni,** the lung fluke of dogs, cats, foxes, goats, swine, mink, muskrat, and man. x 100.

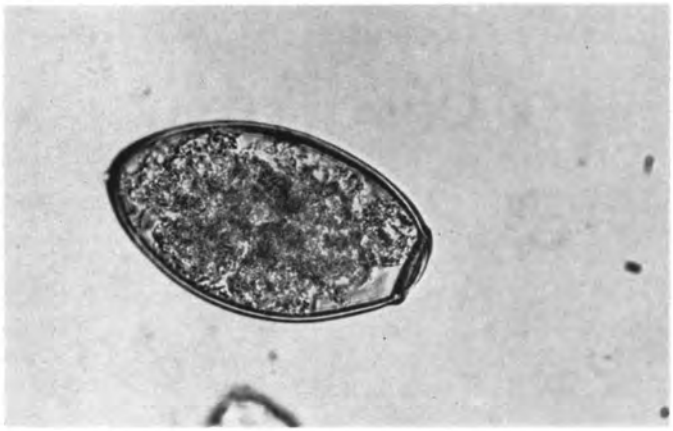

FIG. 93—Ovum of **Paragonimus westermanni.** Note the prominent lid (operculum) at the right. x 410.

DOG, CAT, FOX, MAN

FIG. 94—Ova packets of **Dipylidium caninum,** the double-pored tapeworm of dogs, cats, foxes, and man. The smaller packets may be detected by flotation; the heavier packets sink in the centrifuge tube. x 100.

FIG. 95—An ova packet of **Dipylidium caninum.** Each egg in the packet is provided with six hooklets. x 400.

DOG, CAT, FOX

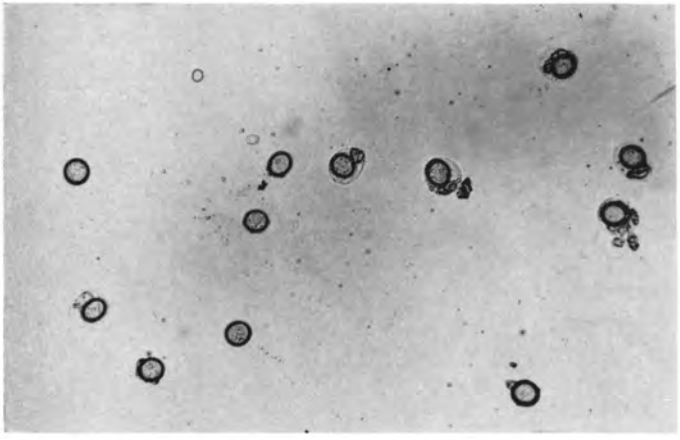

FIG. 96—Ova of **Taenia pisiformis,** one of the rabbit-cyst tapeworms of dogs, cats, and foxes. In general, tapeworm eggs leave the host in ripe tapeworm segments. However, eggs may be found by microscopic fecal examination. x 100.

FIG. 97—Ova of **Taenia pisiformis.** Note the radially striated shell and the embryonic hooklets. The egg at the right is contained within an embryonic membrane. x 400.

CAT, FOX

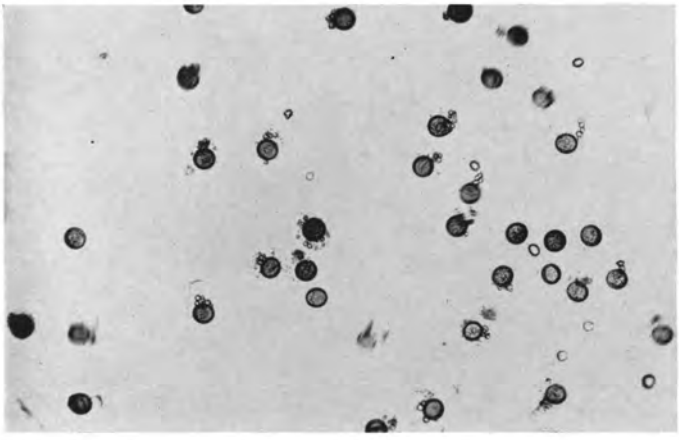

FIG. 98—Ova of **Taenia taeniaeformis,** a common tapeworm of cats and foxes. x 100.

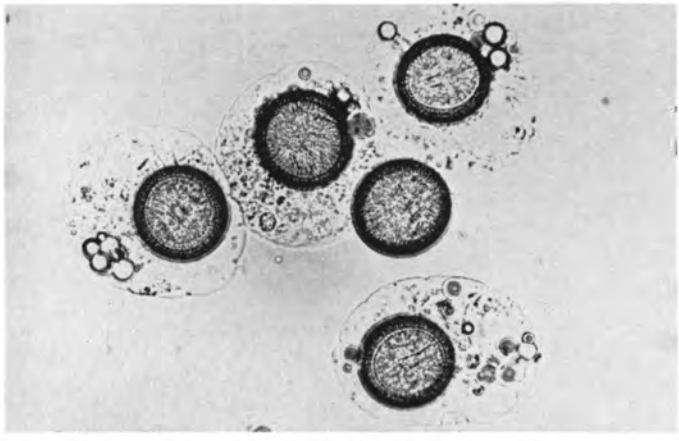

FIG. 99—Ova of **Taenia taeniaeformis.** Four of these are enclosed in embryonic membranes. x 410.

DOG, CAT, FOX, BEAR, MAN, OTHER FISH-EATING MAMMALS

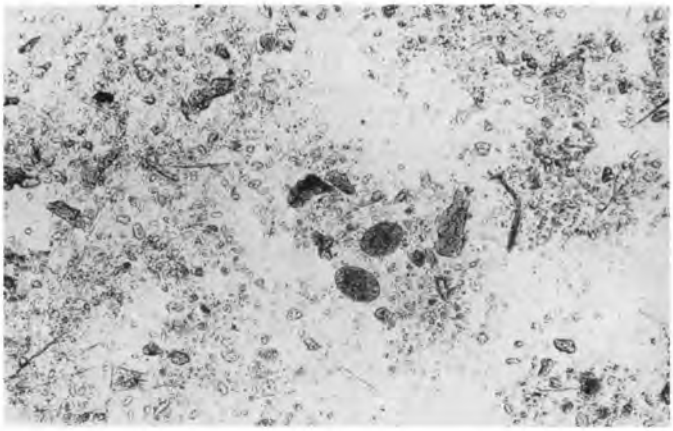

FIG. 100—Ova of **Diphyllobothrium latum,** the broad fish tapeworm of dogs, cats, foxes, bears, man, and other fish-eating mammals. x 100.

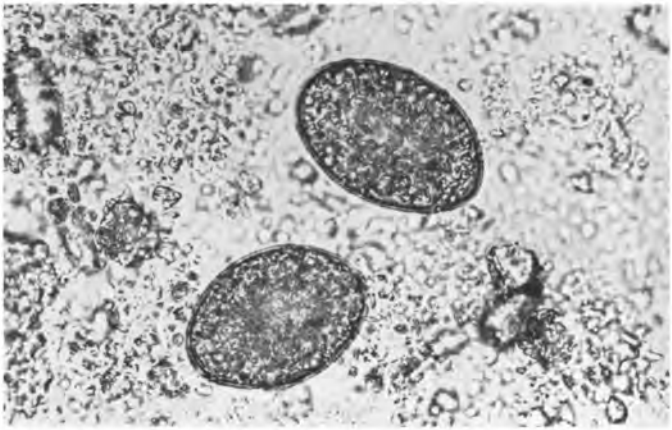

FIG. 101—Ova of **Diphyllobothrium latum.** x 410.

DOG, CAT, FOX

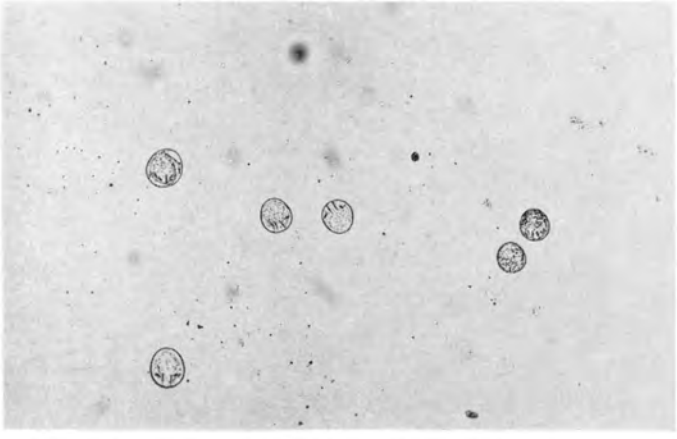

FIG. 102—Ova of **Mesocestoides variabilis,** a seldom-reported tapeworm of dogs, cats, and foxes. These eggs were removed from the egg-sac of a ripe segment. x 100.

FIG. 103—Ova of **Mesocestoides variabilis.** x 410.

DOG, CAT, FOX

FIG. 104—Ova of **Ancylostoma caninum,** the commoner hookworm of dogs, cats, and foxes. x 100.

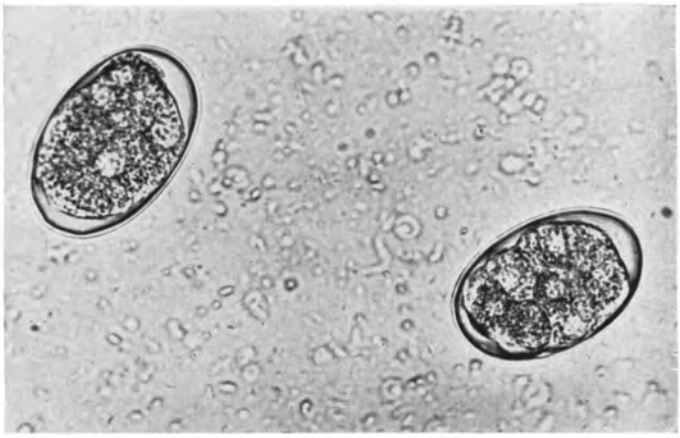

FIG. 105—Ova of **Ancylostoma caninum.** x 410.

DOG, CAT, FOX

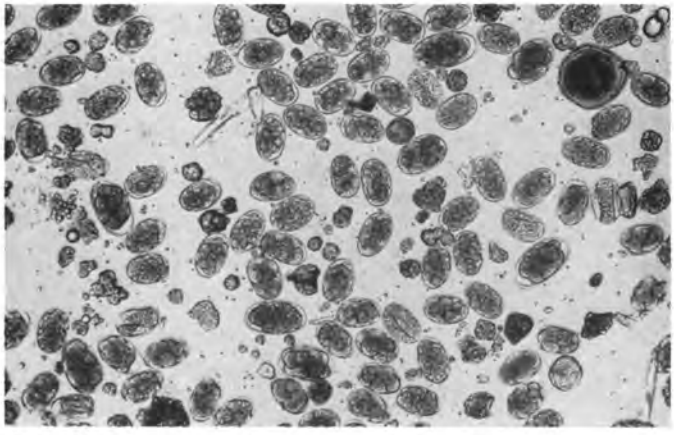

FIG. 106—Ova of **Uncinaria stenocephala** (larger ova) and **Ancylostoma caninum,** (smaller ova) hookworms of dogs, cats, and foxes. At the upper right is an ovum of **Toxocara canis,** one of the ascarids (see Figs. 108, 109). x 100.

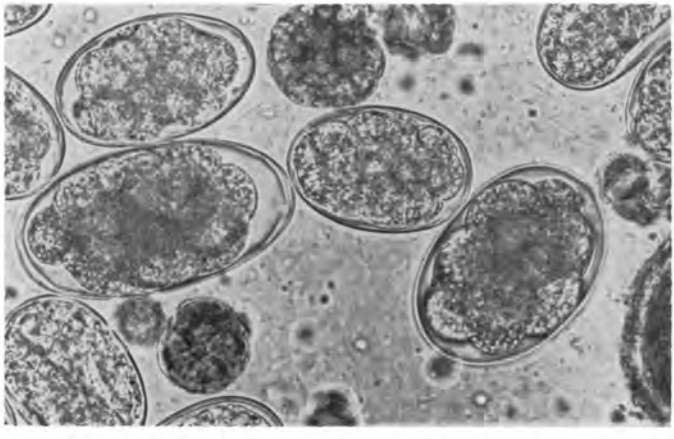

FIG. 107—Ova of **Uncinaria stenocephala** (larger ova) and **Ancylostoma caninum** (smaller ova). x 410.

DOG, CAT, FOX

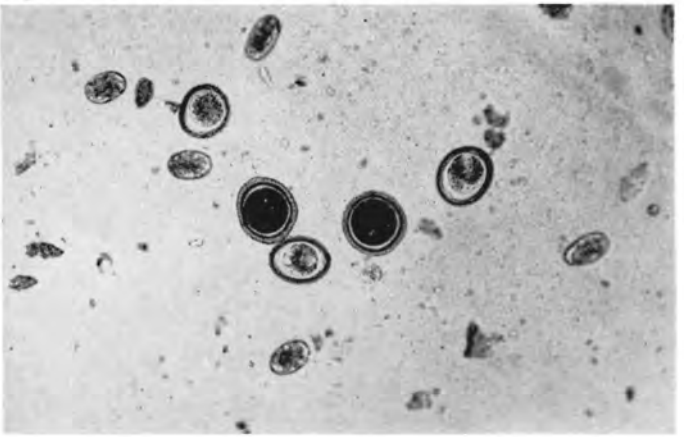

FIG. 108—Ova of **Toxocara canis** and **Toxascaris leonina,** both species of ascarids of dogs and foxes. The latter species also occurs in cats. Included are five ova of **Ancylostoma caninum,** the hookworm of dogs, cats, and foxes. x 100.

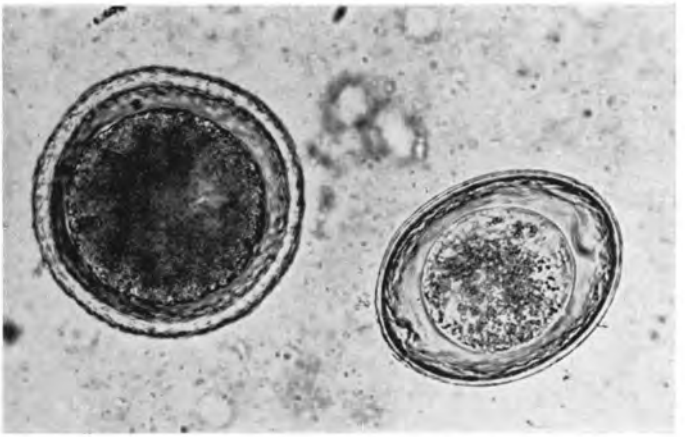

FIG. 109—Ova of **Toxocara canis (left)** and of **Toxascaris leonina (right).** The eggs of **Toxocara canis** are yellow. x 410.

CAT

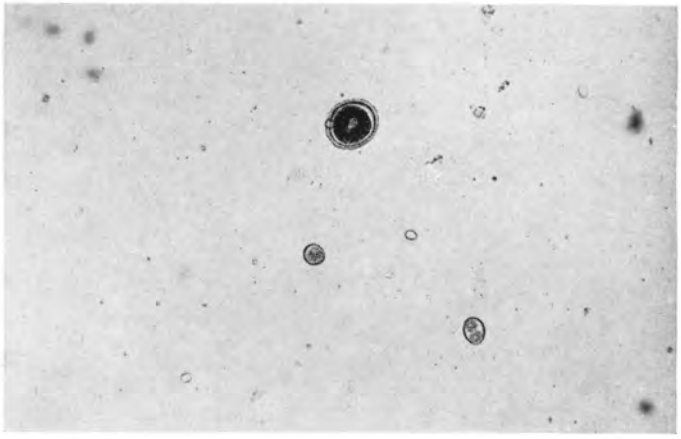

FIG. 110—Ovum of **Toxocara mystax,** an ascarid of cats. Also included are two oocysts of **Isospora felis.** x 100.

FIG. 111—Ovum of **Toxocara mystax** and an oocyst of **Isospora felis.** x 410.

DOG, CAT, FOX

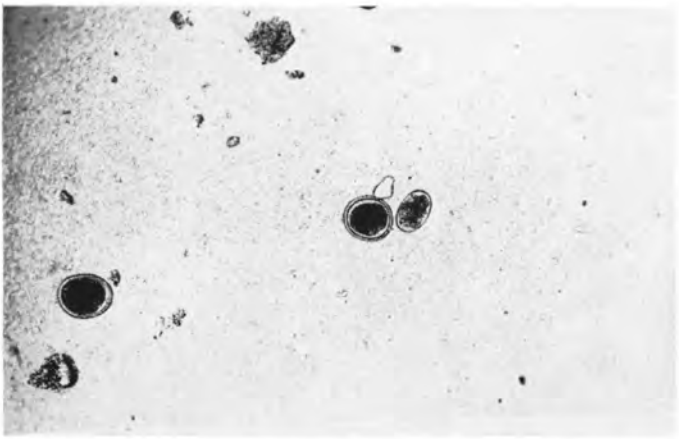

FIG. 112—Ova of **Toxocara mystax,** an ascarid of cats; and an ovum of **Ancylostoma caninum,** a hookworm of cats, dogs, and foxes. x 100.

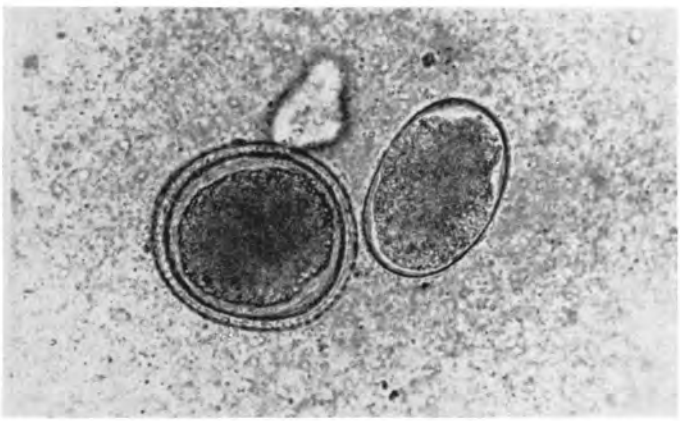

FIG. 113—Ovum of **Toxocara mystax** and an ovum of **Ancylostoma caninum.** x 410.

DOG, FOX

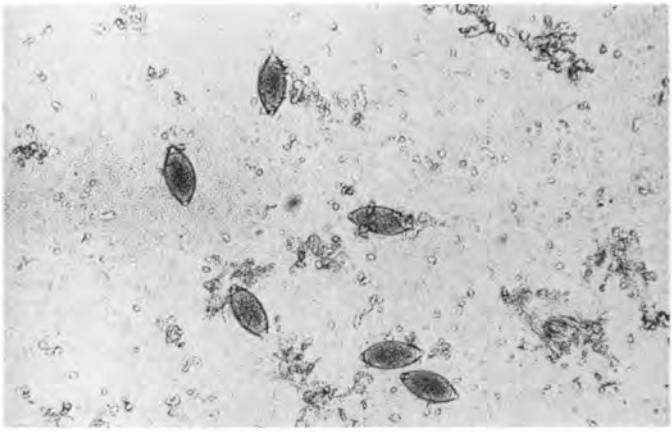

FIG. 114—Ova of **Trichuris vulpis,** the whipworm of dogs and foxes. x 100.

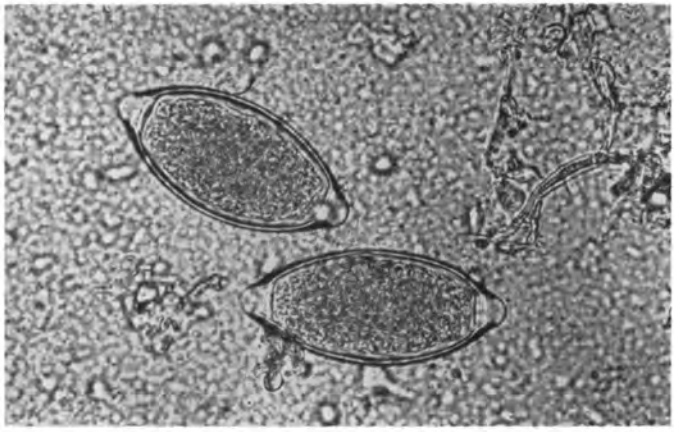

FIG. 115—Ova of **Trichuris vulpis.** Note the larger size and the smooth shell compared with lungworm ova (see Fig. 119). x 410.

DOG, CAT

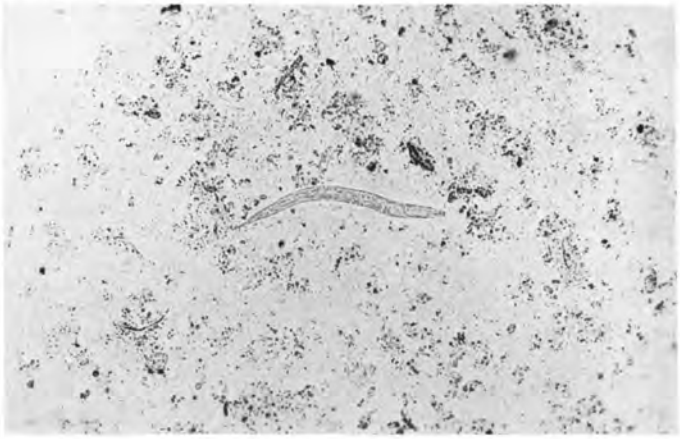

FIG. 116—Rhabditiform larva of **Strongyloides stercoralis,** the threadworm of dogs and cats. The ova hatch in the intestinal mucosa. x 100.

FIG. 117—Rhabditiform larva of **Strongyloides stercoralis.** x 410.

DOG, CAT, FOX

FIG. 118—Ova of **Capillaria aerophila,** the more common lungworm of dogs, cats, and foxes. x 100.

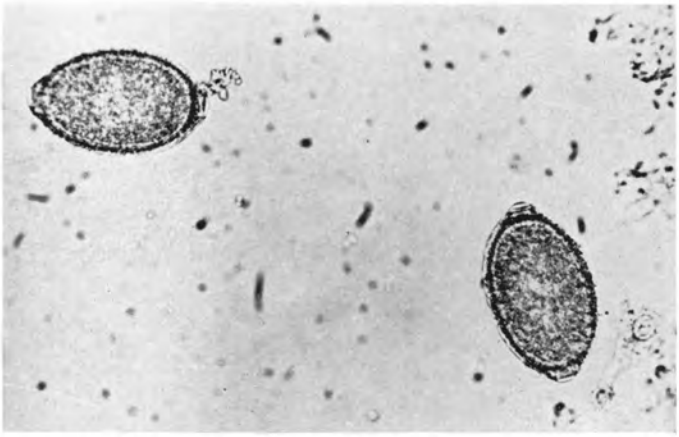

FIG. 119—Ova of **Capillaria aerophila.** The color is yellowish. The shells are finely granular and there is an operculum at each end. The size and the granular shell differentiate them from ova of **Trichuris vulpis,** the whipworm (Fig. 115). x 410.

DOG, FOX

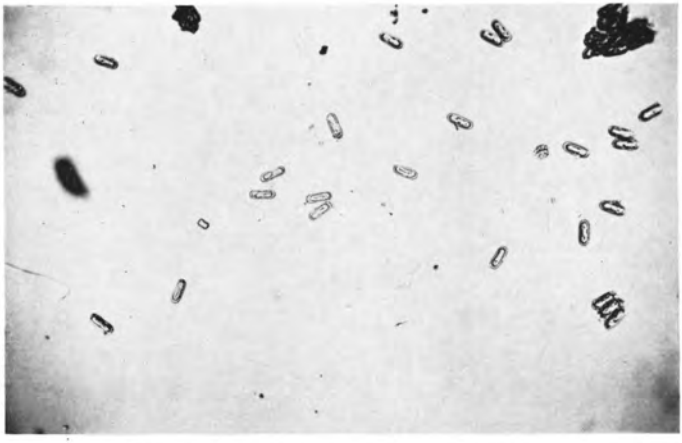

FIG. 120—Ova of **Spirocerca lupi,** the esophageal worm of dogs and foxes. x 100.

FIG. 121—Ova of **Spirocerca lupi.** These eggs are embryonated when laid. x 410.

DOG, CAT, FOX

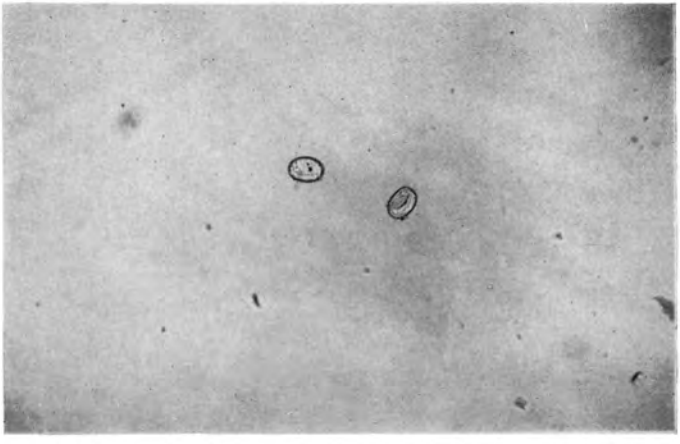

FIG. 122—Ova of **Physaloptera rara**, a stomach worm of dogs, cats, and foxes. x 100.

FIG. 123—Ova of **Physaloptera rara**. These eggs are embryonated when laid. x 410.

DOG, CAT, FOX

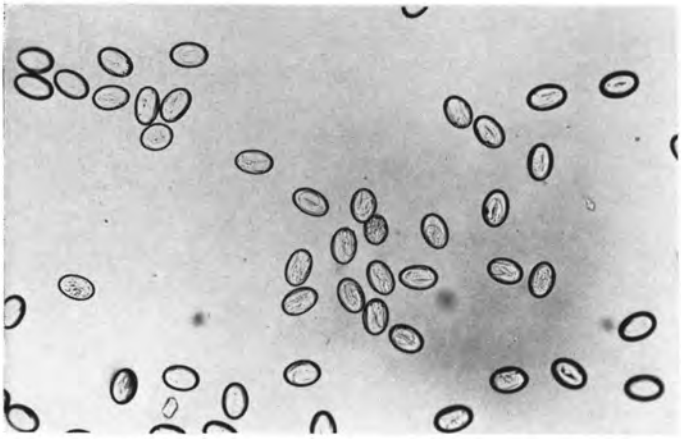

FIG. 124—Ova of **Physaloptera praeputialis,** a stomach worm of dogs, cats, and foxes. x 100.

FIG. 125—Ova of **Physaloptera praeputialis.** These eggs are embryonated when laid. x 410.

DOG, FOX

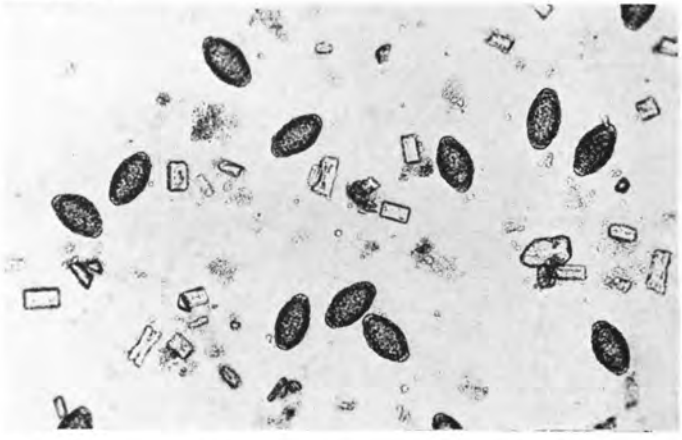

FIG. 126—Ova of **Dioctophyma renale,** the giant kidney worm of dogs and foxes. These eggs are usually found in urinary sediment (note triple phosphate crystals). x 100.

FIG. 127—Ova of **Dioctophyma renale.** The shells are thick and rough. The color is yellowish-brown. x 410.

DOG

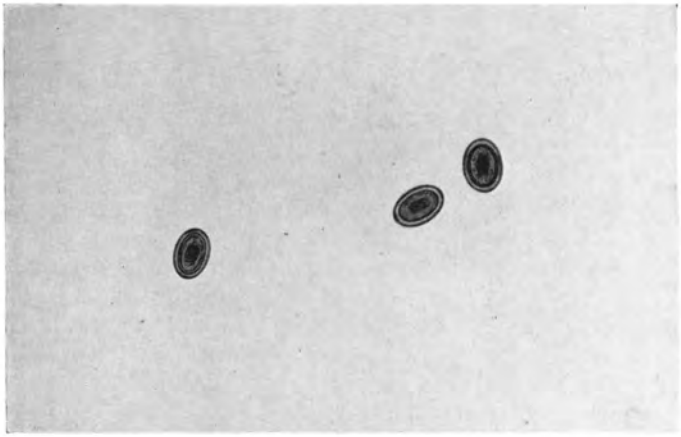

FIG. 128—Ova of **Oncicola canis,** the thorny-headed worm of dogs. x 100.

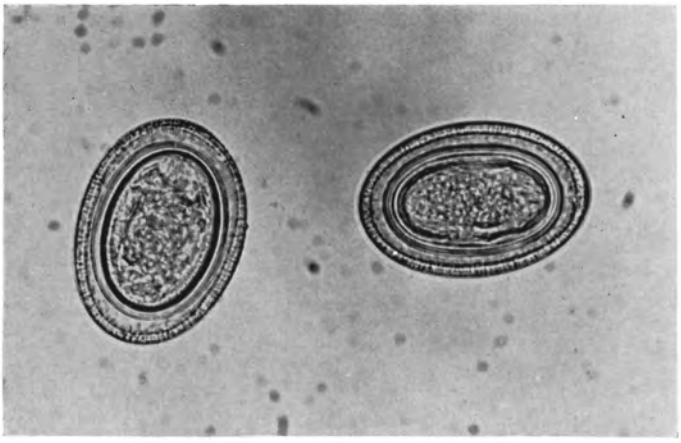

FIG. 129—Ova of **Oncicola canis.** Note the three shells enclosing the embryo. x 410.

DOG

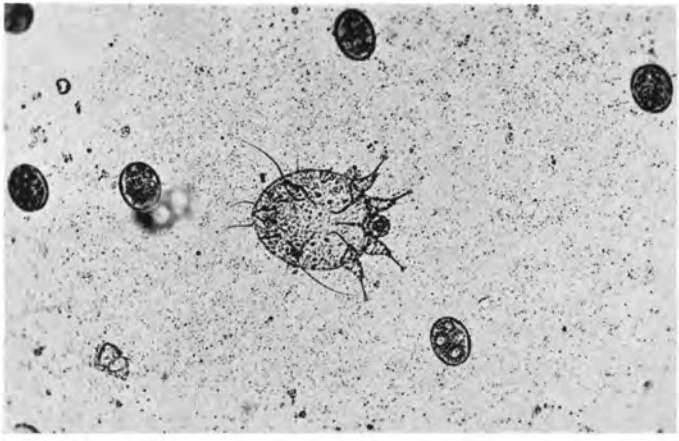

FIG. 130—A larva of **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs; also several ova of **Ancylostoma caninum,** a hookworm, in dog feces. Mange, especially in dogs and cats, may be diagnosed by fecal examination if the host happens to ingest mites when biting the skin lesions. x 100.

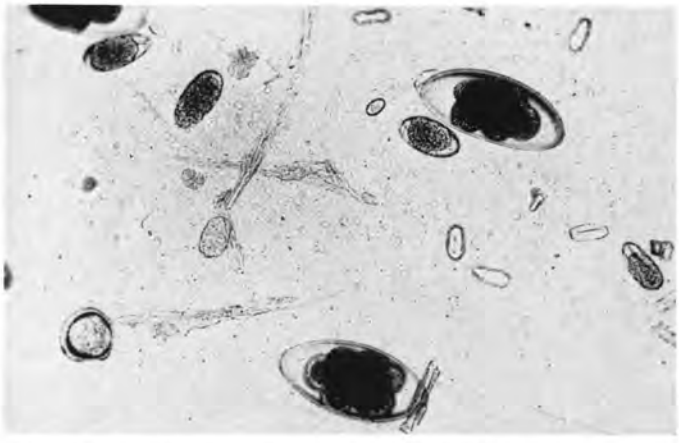

FIG. 131—Spurious parasites. The feces of this dog contains ova and oocysts of sheep parasites. The dog's food was contaminated by sheep feces. The field contains ova of **Nematodirus spathiger, Moniezia expansa, Strongyloides papillosus,** also an unidentified nematode ovum and a coccidial oocyst. x 100.

DOG

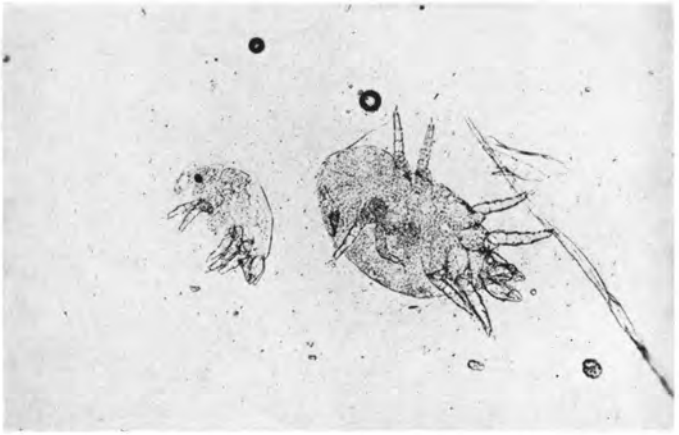

FIG. 132—Pseudoparasite. An adult and a larval "grain" mite in the feces of a dog. x 100.

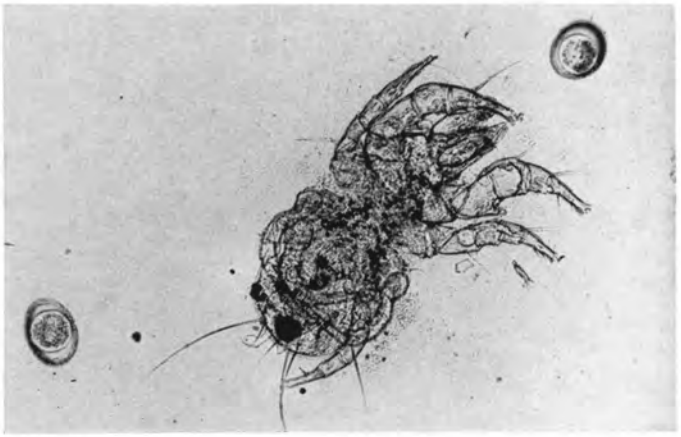

FIG. 133—Pseudoparasite. An adult "grain" mite and two ova of **Toxocara canis** appear in this sample of dog feces. x 100.

DOG

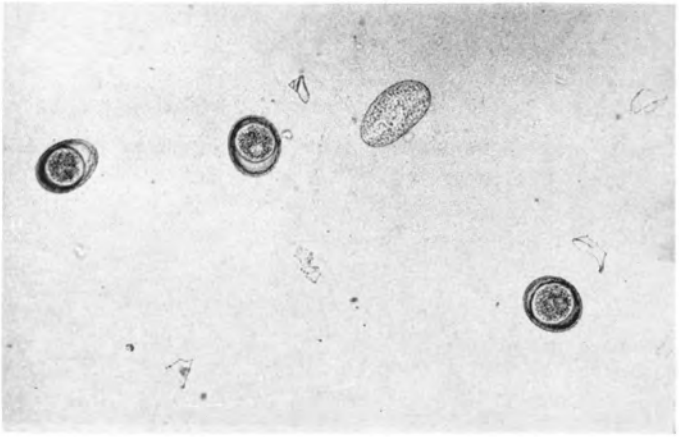

FIG. 134—Pseudoparasite. An ovum of a "grain" mite and three ova of **Toxocara canis** appear in this sample of dog feces. x 100.

DOG

FIG. 135—Pseudoparasite. Pine pollen in dog feces. The color is pale brown. x 100.

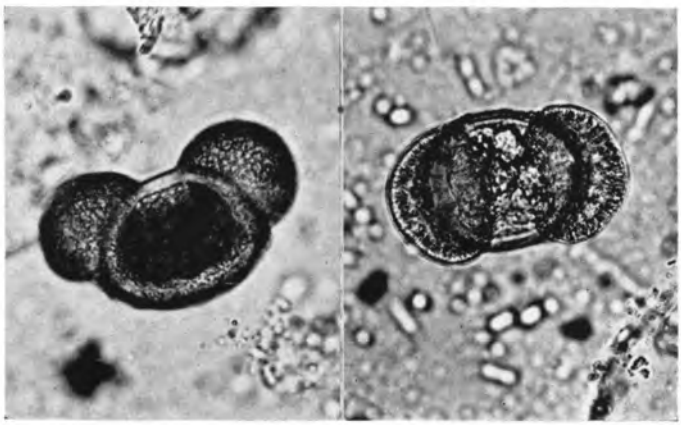

FIG. 136—Pine pollen in the feces of a dog. Side view of a pollen grain **(left)**, showing the two wing-like floats. View from above at the right. x 410.

DOG

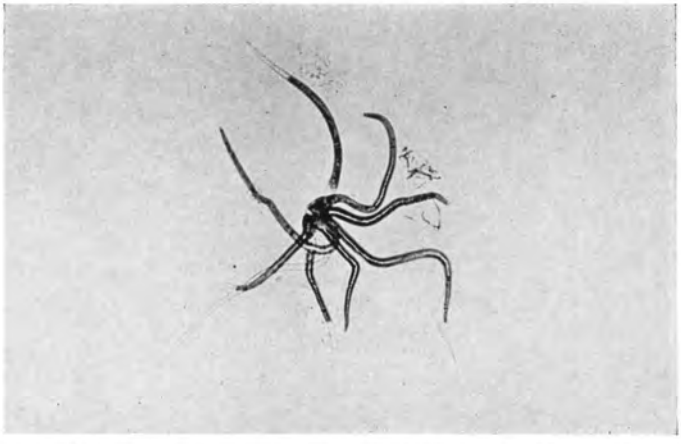

FIG. 137—Pseudoparasite. Plant hairs from dog feces. These resemble the groups of hair-like projections seen on the under surface of oak leaves. x 100.

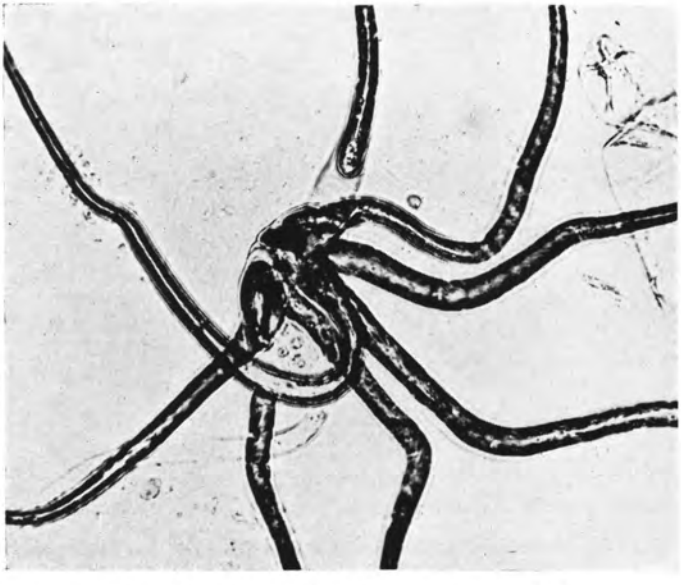

FIG. 138—Plant hairs from dog feces. x 338.

DOG

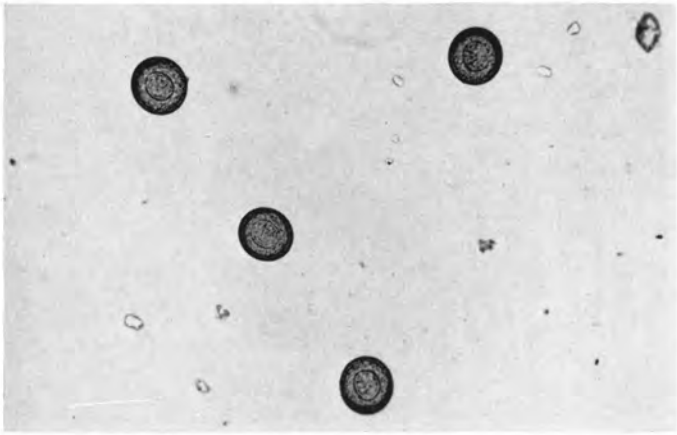

FIG. 139—Spurious parasite. The feces of this dog contains ova of **Hymenolepis diminuta**, a tapeworm of rats, mice, and man. Presumably the dog ingested the small intestine of an infected rodent. These eggs are yellow in color. x 100.

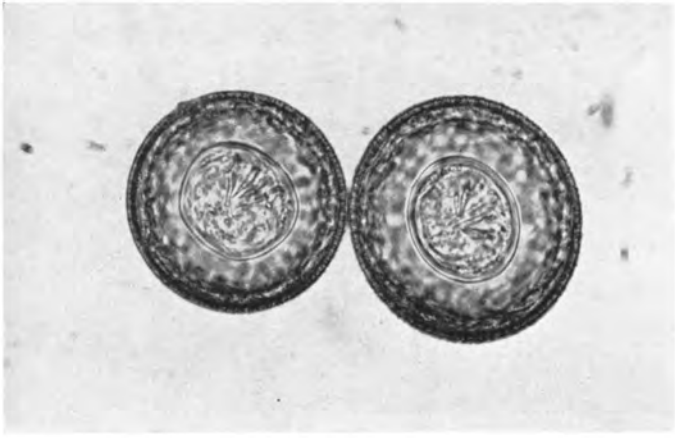

FIG. 140—**Hymenolepis diminuta** ova in dog feces. Note the six hooklets in each embryo. x 410.

DOG

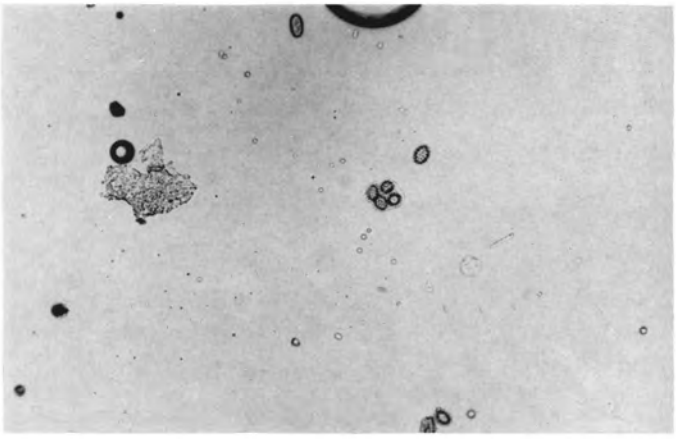

FIG. 141—Pseudoparasite. Corn smut spores in the feces of a dog. These resemble certain tapeworm ova under low power. x 100.

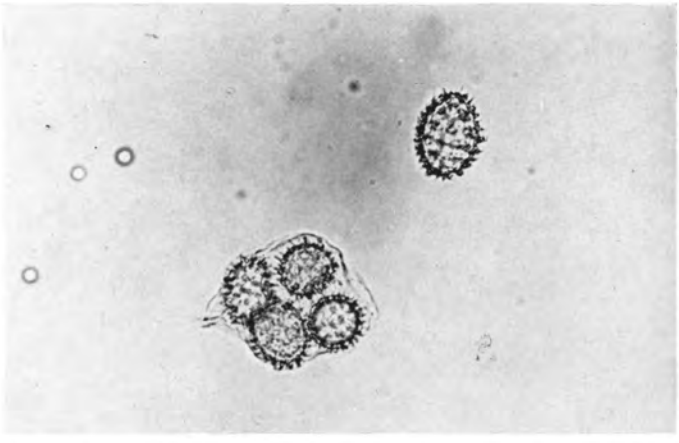

FIG. 142—Corn smut spores in feces. Note the spiny covering. x 410.

DOG

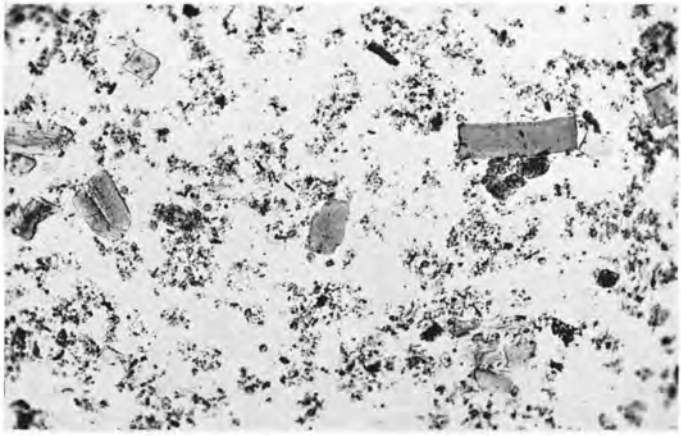

FIG. 143—Undigested muscle in a dog's feces. x 100.

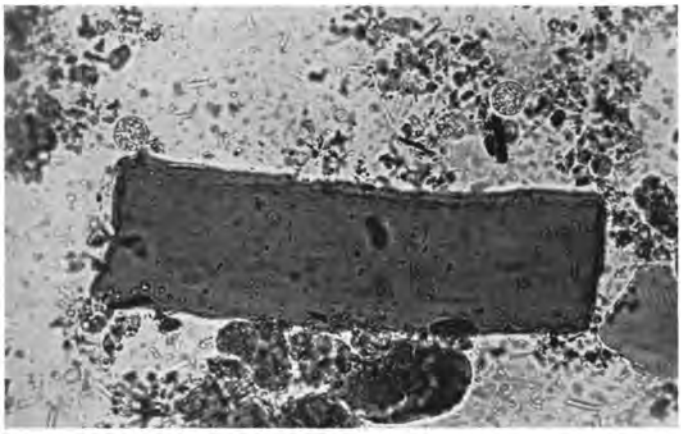

FIG. 144—Undigested muscle in a dog's feces. x 410.

CHICKEN

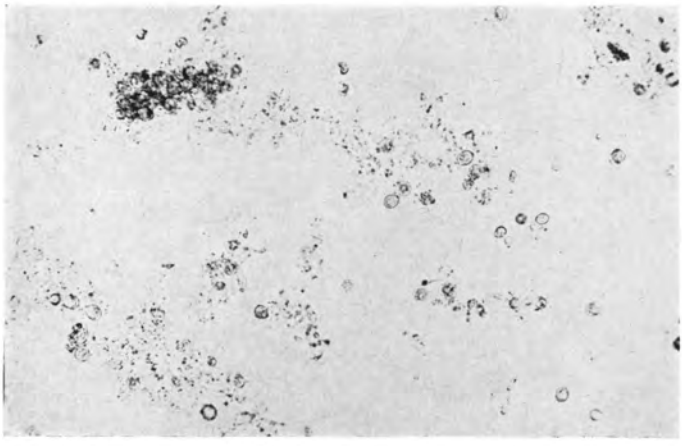

FIG. 145—Oocysts of **Eimeria tenella,** the cecal coccidium of chickens. x 100.

FIG. 146—Oocysts of **Eimeria tenella.** x 410.

TURKEY

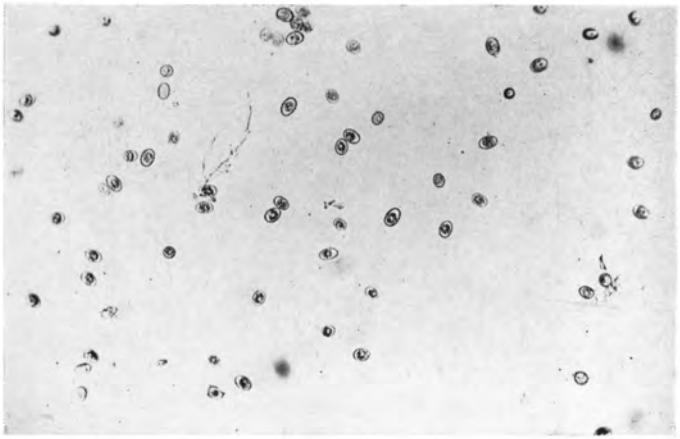

FIG. 147—Oocysts of **Eimeria meleagridis** and **Eimeria meleagrimitis,** two species of coccidia of turkeys. x 100.

FIG. 148—Two oocysts of **Eimeria meleagridis** and four oocysts of **Eimeria meleagrimitis.** x 410.

PHEASANT, TURKEY

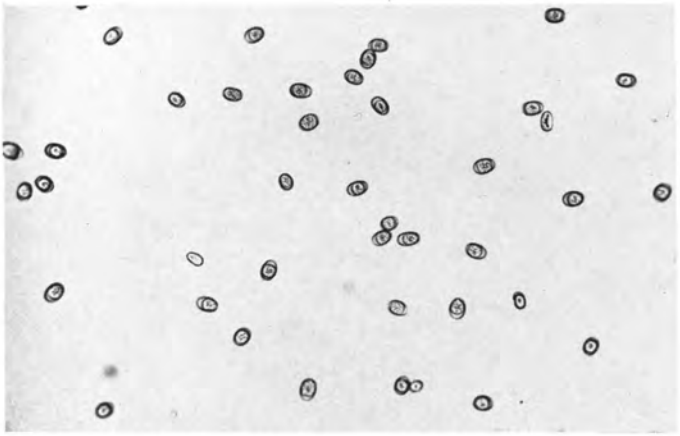

FIG. 149—Oocysts of **Eimeria dispersa** and **Eimeria phasiani,** coccidia of pheasants. **Eimeria dispersa** is also a coccidium of turkeys. x 100.

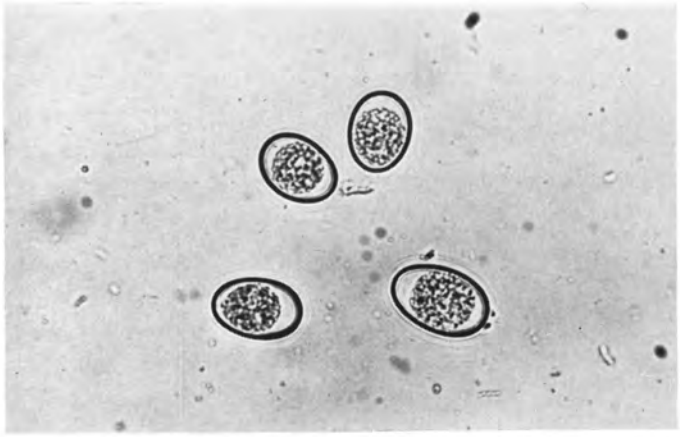

FIG. 150—Oocysts of **Eimeria dispersa** and **Eimeria phasiani.** The latter species is slightly the larger. x 410.

PIGEON

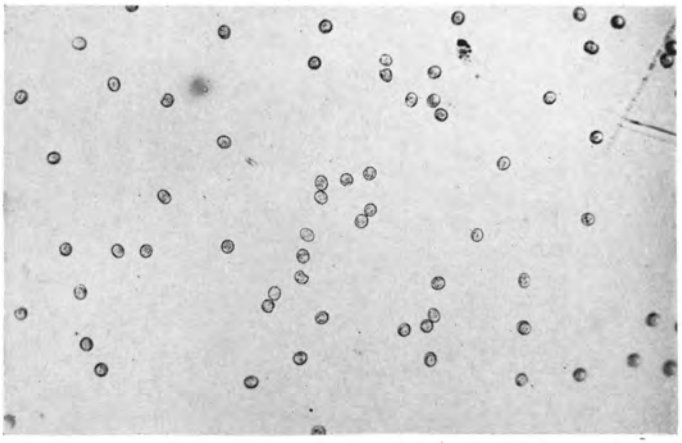

FIG. 151—Oocysts of **Eimeria labbeana,** the coccidium of pigeons. x 100.

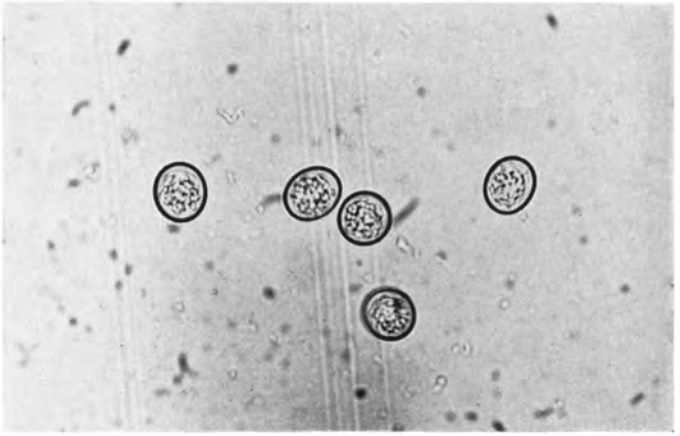

FIG. 152—Oocysts of **Eimeria labbeana.** x 410.

CHICKEN, TURKEY

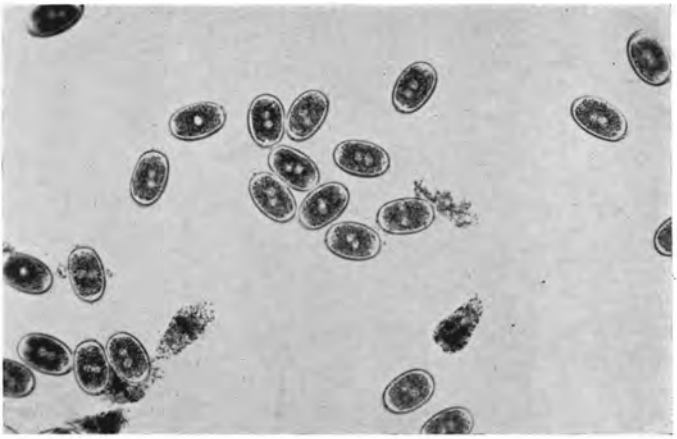

FIG. 153—Ova of **Ascaridia galli,** the ascarid of the chicken and rarely of the turkey. x 100.

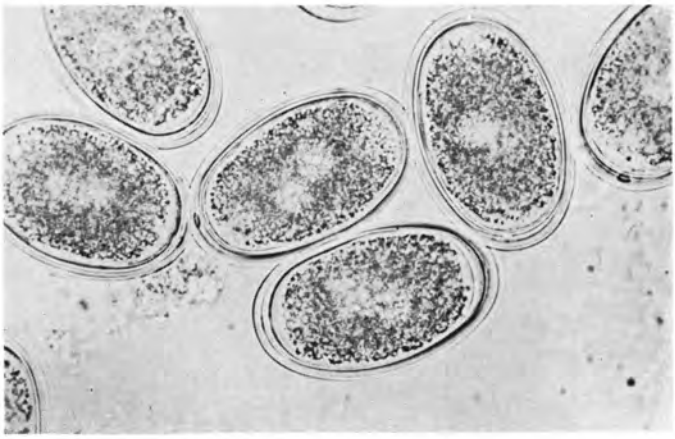

FIG. 154—Ova of **Ascaridia galli.** x 400.

CHICKEN, TURKEY, GUINEA FOWL, QUAIL, PHEASANT

FIG. 155—Ova of **Heterakis gallinae,** the cecal worm of chickens, turkeys, guinea fowl, quail, and pheasants. x 100.

FIG. 156—Ova of **Heterakis gallinae.** x 410.

TURKEY, DUCK, QUAIL, PHEASANT

FIG. 157—Ova of **Capillaria contorta,** the crop capillarid of turkeys, ducks, quail, and pheasants. x 100.

FIG. 158—Ova of **Capillaria contorta.** There is an operculum at each pole. x 410.

CHICKEN, TURKEY, PHEASANT

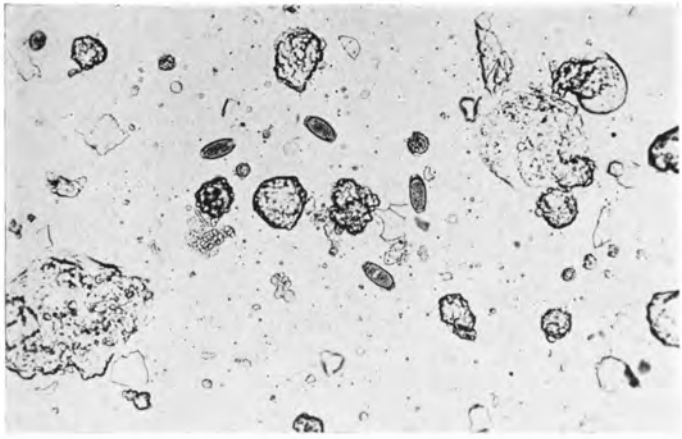

FIG. 159—Ova of **Capillaria caudinflata,** a capillarid worm of the small intestine of chickens, turkeys, and pheasants. x 100.

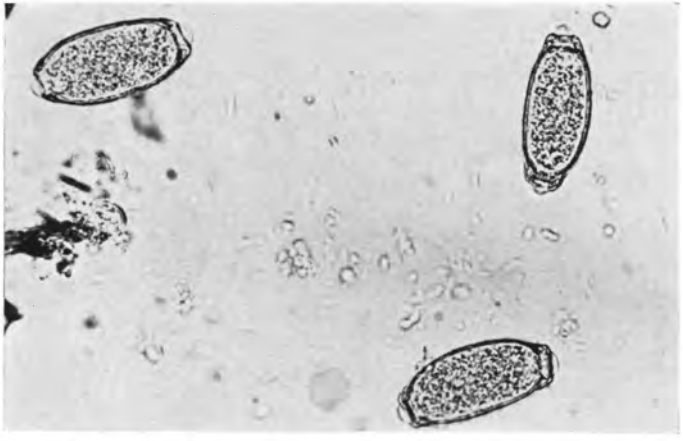

FIG. 160—Ova of **Capillaria caudinflata.** Note the operculum at each pole. x 410.

POULTRY

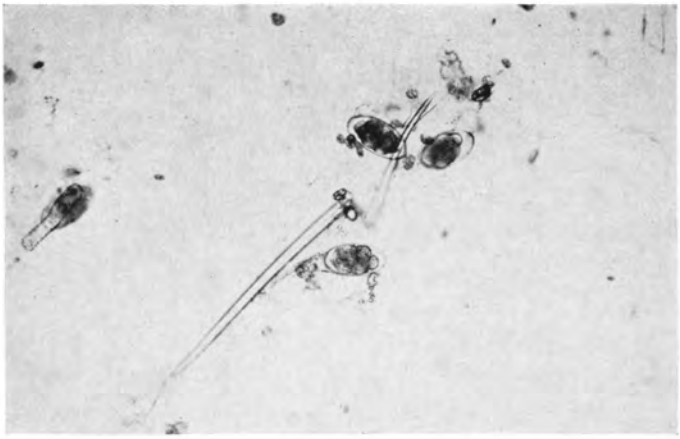

FIG. 161—Ova of **Syngamus trachea,** the gapeworm of poultry. x 100.

FIG. 162—Ovum of **Syngamus trachea.** There is an operculum at both of the poles. x 410.

CHICKEN

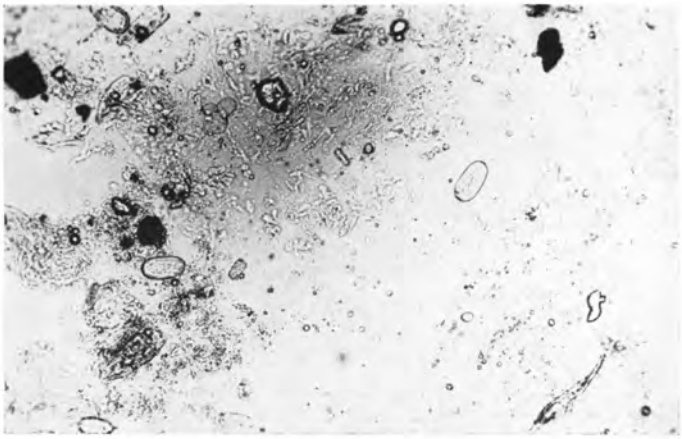

FIG. 163—Ova of **Tetrameres americana**, the globular stomach worm of chickens. x 100.

FIG. 164—Ovum of **Tetrameres americana**. The eggs of this nematode are embryonated when laid. x 410.

CHICKEN, TURKEY, GUINEA FOWL, PIGEON

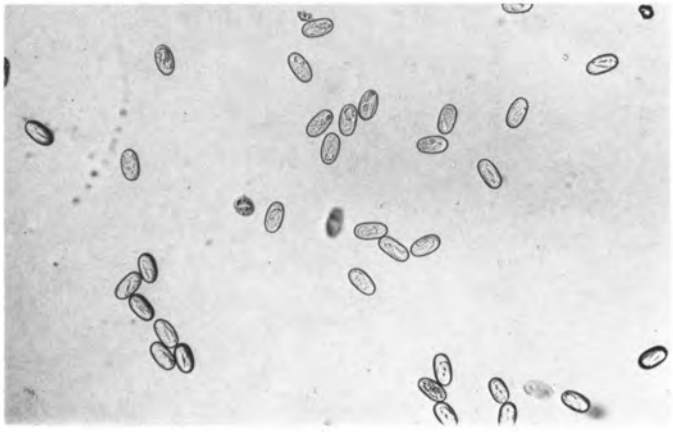

FIG. 165—Ova of **Dispharynx nasuta,** the spiral stomach worm of chickens, turkeys, guinea fowl, and pigeons. x 100.

FIG. 166—Ova of **Dispharynx nasuta.** The ova are embryonated when laid. x 410.

RABBIT, HARE

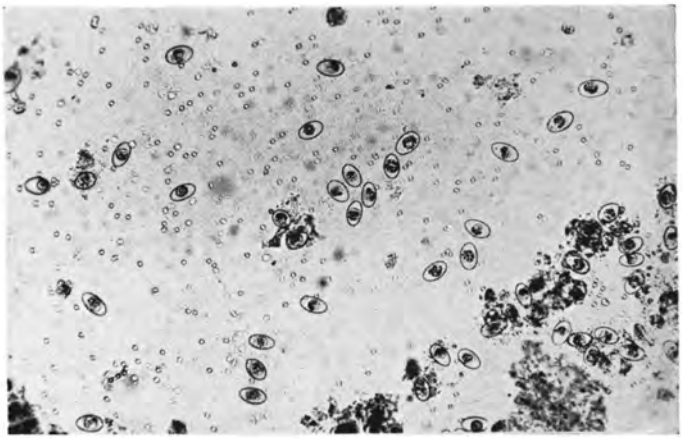

FIG. 167—Oocysts of **Eimeria stiedae,** the hepatic coccidium of rabbits and hares. These were removed from the bile duct. They may also be found in the feces. x 100.

FIG. 168—Oocysts of **Eimeria stiedae.** x 410.

RABBIT

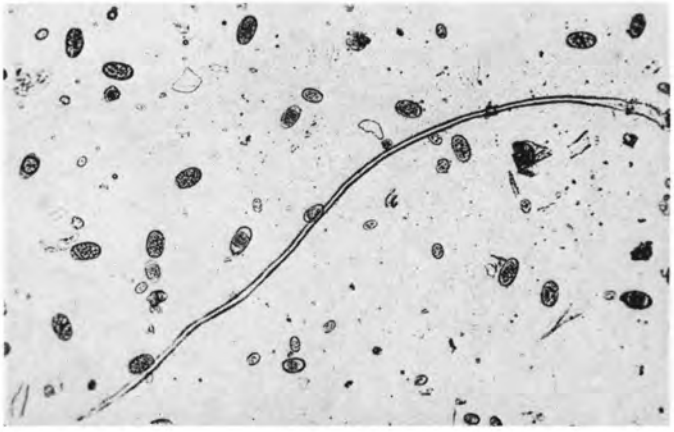

FIG. 169—Oocysts of several **Eimeria** species, intestinal coccidia of rabbits. A long plant hair is present. x 100.

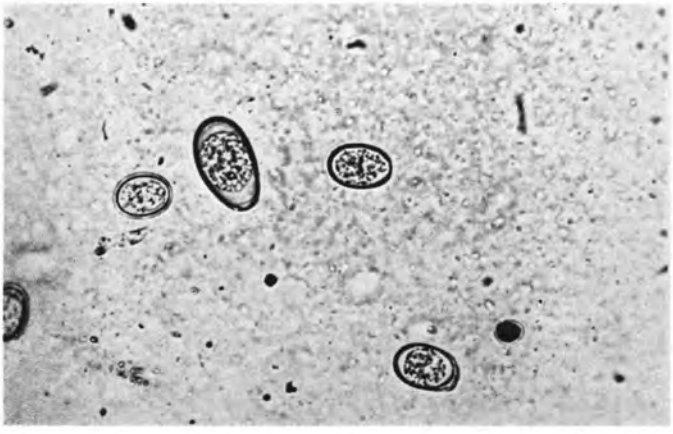

FIG. 170—Oocysts of three **Eimeria** species, coccidia of rabbits. x 410.

RABBIT, GUINEA PIG

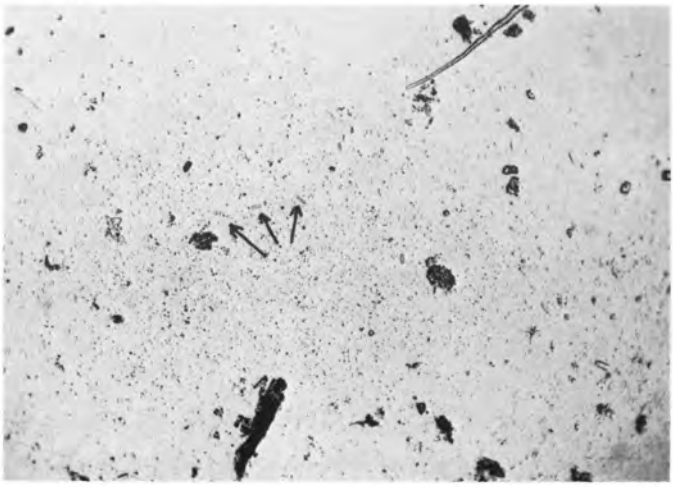

FIG. 171—Pseudoparasite. **Saccharomycopsis guttulatus,** a yeast commonly found in the feces of rabbits and guinea pigs. It is not believed to be pathogenic. Arrows point to the yeasts. x 100.

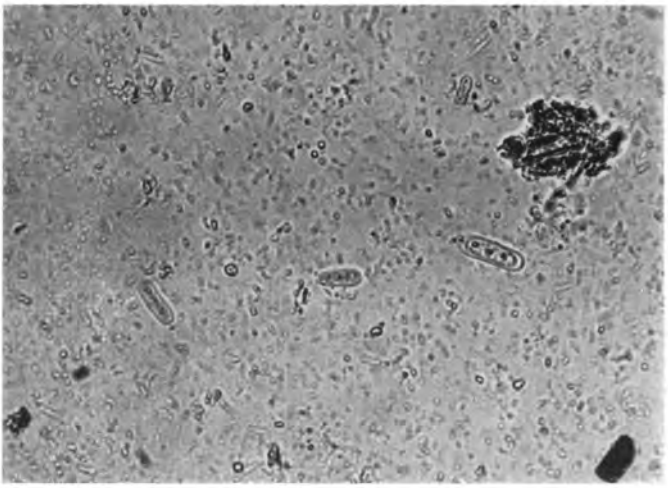

FIG. 172—Pseudoparasite. **Saccharomycopsis guttulatus.** x 410.

MAN

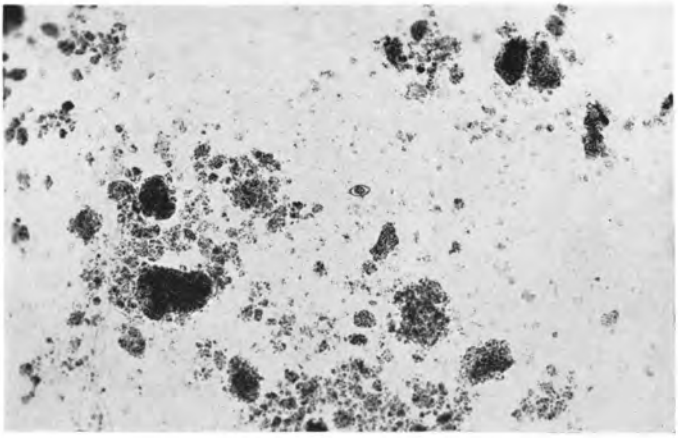

FIG. 173—Oocyst of **Isospora hominis,** the coccidium reported as occurring in man. From human feces. x 100.

FIG. 174—Oocyst of **Isospora hominis.** x 410.

MAN

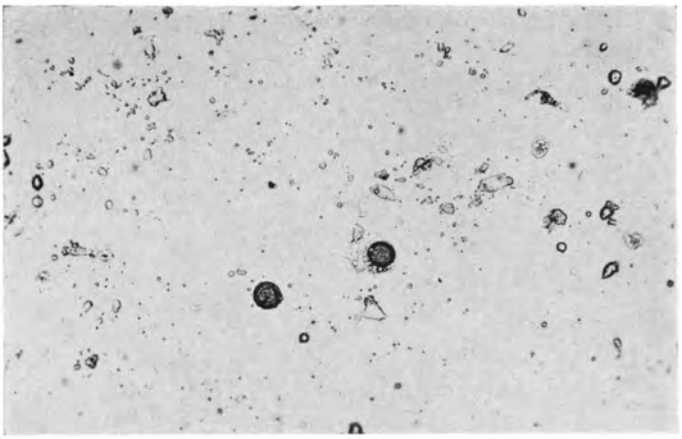

FIG. 175—Ova of **Taenia saginata,** the beef tapeworm of man. x 100.

FIG. 176—Ova of **Taenia saginata.** The egg at the right is contained within an embryonic membrane. x 410.

MAN, RAT, MOUSE

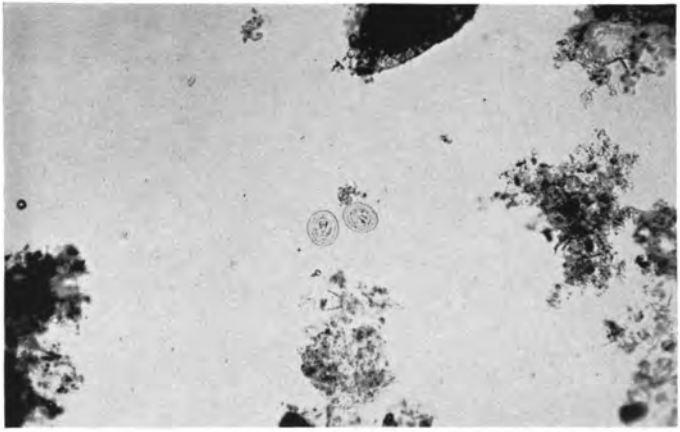

FIG. 177—Ova of **Hymenolepis nana,** the dwarf tapeworm of man, rats, and mice. x 100.

FIG. 178—Ova of **Hymenolepis nana.** There are from four to eight slender filaments on each polar thickening of the inner shell membrane. x 385.

MAN

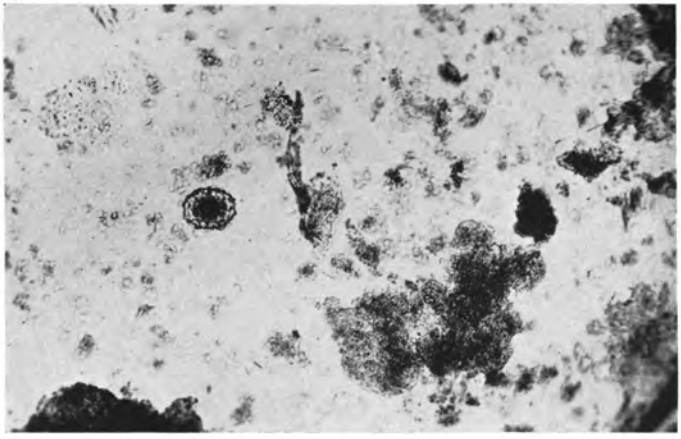

FIG. 179—Ovum of **Ascaris lumbricoides,** the ascarid of man. x 100.

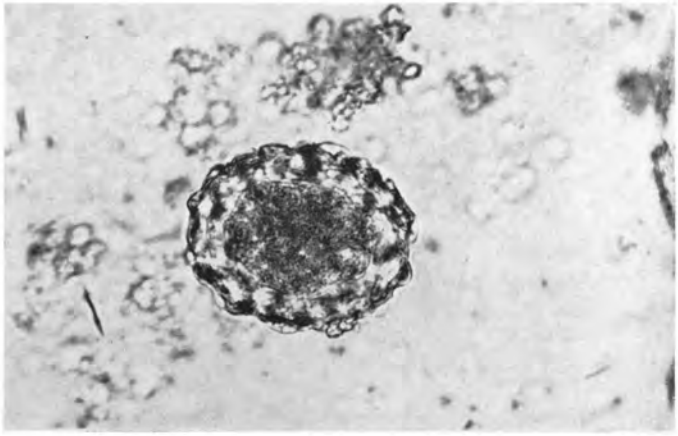

FIG. 180—Ovum of **Ascaris lumbricoides.** x 410.

MAN

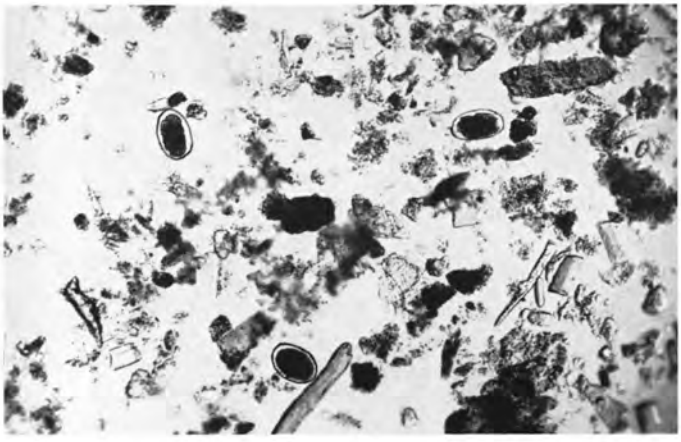

FIG. 181—Ova of **Necator americanus,** the new-world hookworm of man. Simple smear. x 100.

FIG. 182—Ovum of **Necator americanus.** x 410.

MAN

FIG. 183—Ova of **Enterobius vermicularis,** the pinworm or rectal worm of man. x 100.

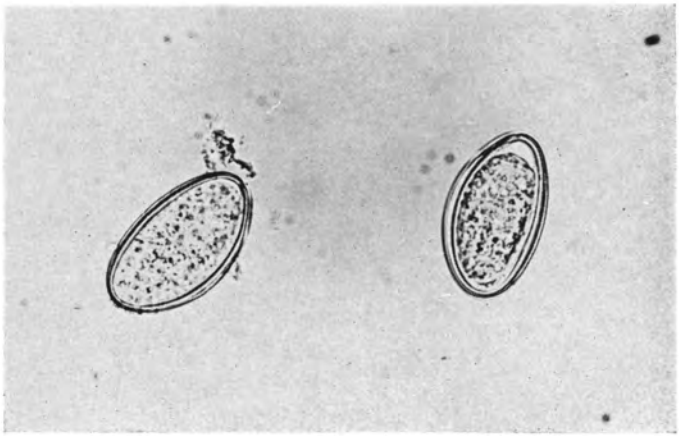

FIG. 184—Ova of **Enterobius vermicularis.** x 410.

MAN

FIG. 185—Larvae of **Strongyloides stercoralis,** the threadworm of man. x 100.

FIG. 186—Larva of **Strongyloides stercoralis.** x 410.

MAN

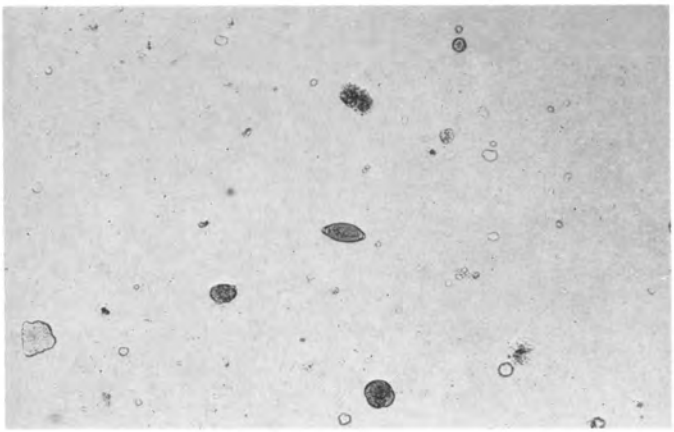

FIG. 187—Ovum of **Trichuris trichiura,** the whipworm of man. x 100.

FIG. 188—Ovum of **Trichuris trichiura** of man. Note the resemblance to the ova of the swine whipworm (Fig. 77). x 410.

MAN

FIG. 189—Pseudoparasite. Banana seeds in human feces. Grossly these resemble small brownish tapeworm segments. x 3.

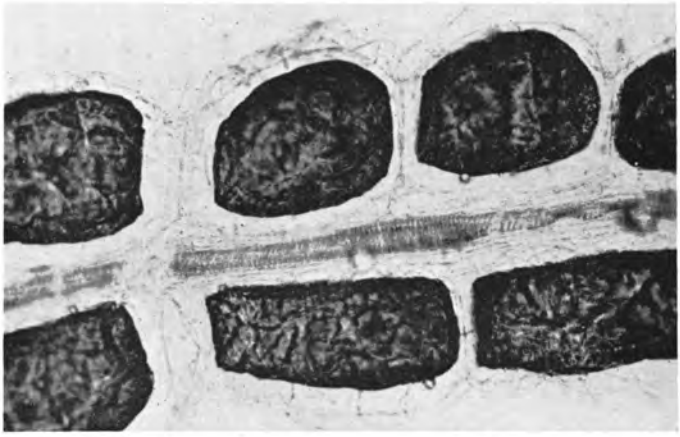

FIG. 190—Banana seeds in human feces. x 100.