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What Do Fecal Worm Egg Counts Tell Us?

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What is a worm egg count?

A fecal worm egg count (FEC) is done on manure to look for worm eggs. It is quantitative versus qualitative in that the result is expressed as the number of eggs per gram (epg) of manure as opposed to "positive" or "negative" or "+, ++, or +++" results that are often given from simple flotation procedures. A quantitative result gives us a means to quantify changes over time or in response to a treatment.

What are we measuring?

The worms we are most concerned about are in the roundworm family and live in the stomach and intestines of sheep and goats. The ones of most importance in our region are in the trichostrongylid family and all have similar life cycles. Adult worms expel eggs that pass outward in feces (manure). These eggs hatch into larvae on pastures under favorable conditions of moisture and temperature. When the worm larvae are ingested by sheep and goats, they develop into adult worms in the gastrointestinal tract and begin the cycle all over again.

What can we use worm egg counts for?

There are three main uses for worm egg counts. They are to detect dewormer resistance, to monitor pasture contamination, and to select animals for their genetic ability to resist worms. This fact sheet will discuss the first use.

What are some limitations of worm egg counts?

First off, FECs are *estimates*. There is some day-to-day variability in counts even in stable worm populations. The count can also be influenced by the type of forage the animals are eating with respect to its digestibility and its water content. Very loose stools are somewhat diluted, which causes some underestimation of the actual egg count. In addition, there are different ways of performing quantitative egg counts, and the method can influence the magnitude of the result. Therefore, people should be very careful in comparing their results with those from other laboratories.

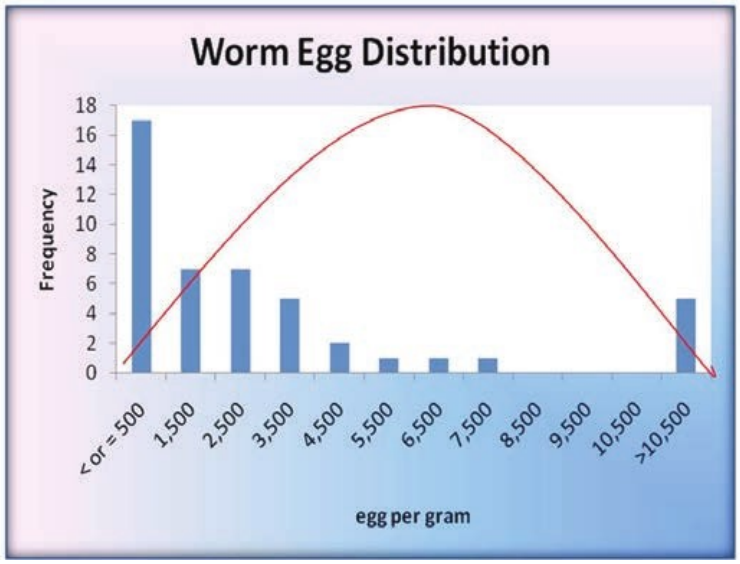
Secondly, FECs are a snapshot in time. They reflect the degree of egg shedding and pasture contamination at that point in time, but without other information, they are not very predictive. Mature *Haemonchus contortus* females may produce (depending on the author) 3,000–10,000 eggs per day. However, immature worms may not produce many eggs, but they still suck blood. As the immature worms age over a couple of weeks, their egg production increases. Depending on the level of pasture contamination, FECs can rise considerably in a short time. For example, during the course of an NCR SARE-funded, on-farm project in 2010, two groups of lambs comprised of 20 lambs each were removed

from a light to moderately contaminated pasture after grazing it for three weeks and then were placed on experimental forage plots (worm larvae-free) on August 30. Their average FECs were about 600 epg. On September 14, the FECs for both groups averaged about 2,800 epg. Over those two weeks of grazing, both groups gained acceptably well, and their red blood cell counts were still in the normal range. Moreover, the September 14 FECs still weren't reflecting eggs from worms lambs acquired in the last week of grazing the contaminated pasture because it takes about 19–21 days from the time infective larvae are ingested until they produce eggs in the manure. On heavily contaminated pastures, FECs can rise explosively such that lambs can initially appear normal and then begin dying within a two-week period. Average FECs in such situations can reach 10,000 epg of manure (two to three lamb-sized manure pellets weigh about one gram).

In another project, weekly FECs were obtained on a group of March-born lambs maintained continuously on pasture from birth. FECs averaged 0; 42; 89; 1,050; and 1,950 epg from May 14 through June 11. This dramatic change was the result of the following: 1) the lambs' gradual increase in consumption of infective larvae as they got older and consumed more forage; 2) the buildup of worm larvae on the pasture as a result of the prolific egg producer, *Haemonchus,* becoming the predominant worm species; and 3) the time needed for ingested larvae to become egg-laying adults. It is also characteristic of what happens on many Ohio pastures in a typical summer. Therefore, a single egg count for a group of lambs or ewes taken out of context with other information is not predictive of what ***is going*** to happen nor is it always a good measure of the worm burden the animals are carrying. Nevertheless, FECs do give us some information about what is happening at the time the samples are taken and can be generally reflective of worm burdens.

How many animals do I need to sample?

If FECs are to be useful, they have to reasonably reflect the makeup of the group of interest, whether that be lambs/kids or ewes/does. Research and observations over the last 50–60 years consistently show that egg counts from a group of individuals, as well as worm numbers, are not "normally" distributed across a typical bell-shaped curve. Usually only a few individuals have very high counts, and even when severe parasitic disease is present in a group, there frequently are animals in that group with very low FECs. For example, in an investigation of ivermectin resistance, where lambs were actually dying from parasitism, the average FEC for a group of 46 lambs was 3,800 epg of feces. However, two animals had FECs of zero, and the lowest 21 were each less than 1,000 epg. The top five animals had counts of 13,800; 20,050; 23,950; 25,000; and 29,250 epg. If you wanted a reliable estimate of the average count for the group, would samples from three animals, or even five, be enough? Not likely. Generally speaking, you need samples from at least 15 animals to get a reliable estimate of the group average. For those of you reading this that are statistically inclined, you are probably thinking "Yes, but using a simple numeric average for populations like this is flawed!" You are correct; however, for reasons beyond the scope of this discussion, a simple group average is an accepted measure used by parasitologists the world over.

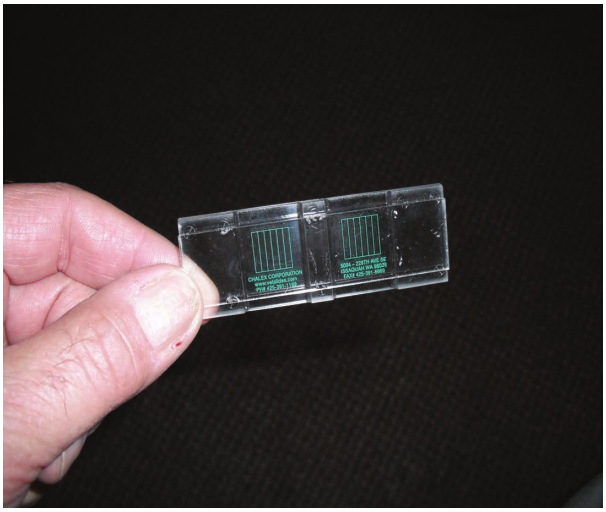


FEC distribution of samples collected from 46 lambs experiencing severe parasitism. The red curve illustrates a hypothetical "normal" distribution of data results.

How are worm egg counts performed?

The most common method of determining FECs for sheep and goats is the McMaster technique. Although there are several variations of how this is done, the basic method uses a weighed sample of

manure, a known dilution of it in a solution that causes the eggs to float, and a specialized counting slide to count the eggs. After the slide's chambers have been filled with the manure suspension in flotation solution, it is placed under a microscope, and the eggs are counted under a grid that defines a known volume of the suspension. Usually the area under two grids is counted and the results averaged and multiplied by a dilution factor. Because the number of grams of feces and their dilution is known, the result gives an estimate of the number of eggs in a specific amount of manure—expressed as eggs per gram (epg) of feces. McMaster counts are not harder to do than simple flotations, and the equipment is relatively inexpensive and reusable. Most methods require at least two grams of manure, and usually four grams are used as this amount provides a more accurate estimate. This means you need to provide your veterinarian with about a tablespoonful of manure from each animal sampled for a proper exam. One pellet is not enough.



A two-chambered slide for counting worm eggs using the McMaster technique.

How are worm egg counts used to detect dewormer resistance?

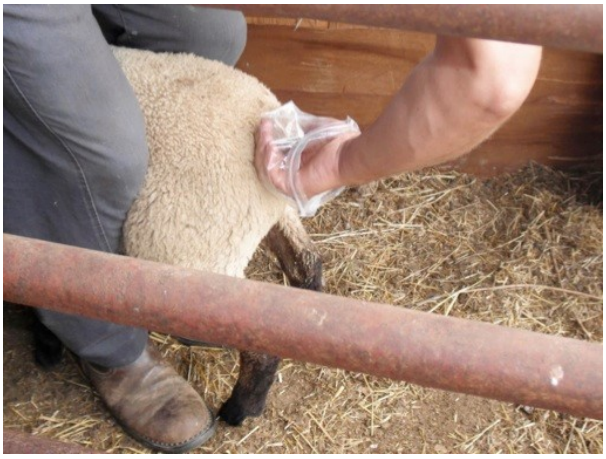
Perhaps the most common approach used for sheep and goats is to 1) collect about 15 samples from animals at the time they are treated; 2) perform the individual egg counts and determine the average FEC for the group; and 3) then collect samples again from those animals at a later time and again determine the average FEC for the group. If the dewormer is working as we would like it to, there should be at least a 95% reduction in the average FEC for the post-treatment samples as compared to the pre-treatment samples. It is often best to sample the same animals both times, but if 15–20 animals that accurately represent the group are used, it is not necessary.

Because dewormers in the benzimidazole class (Valbazen®, Safeguard®) and the macrocyclic lactone class (Ivermectin Sheep Drench®) can temporarily lower egg production in worms, manure samples should be collected about 8–10 days or 14–17 days, respectively, after using them. Manure samples can be collected as quickly as 5–7 days post-treatment if levamisole is used (Prohibit®). If more than one dewormer class is being examined at a time, a 14–17 day post-treatment interval should be used with the caution that occasionally levamisole does not kill all immature stages of worms, which could result in seeing eggs from them at 14–17 days.[1]

An alternative approach, and certainly the best one if enough animals are available, uses an untreated control group of animals. In this approach, the test group of 15 or more animals is treated with a dewormer, and then after the appropriate post-treatment period has passed (see above), FECs are determined on samples from the animals in the treated group and from a group of untreated animals of similar age, weight, and parasite exposure. As in the other method, we are looking for at least a 95% reduction in average FEC in the treated group compared with the untreated animals. This method accounts for reductions in egg counts in the groups during the post-treatment interval that might not be attributable to the dewormer as well as increases in egg counts resulting from maturing worms. It has the additional advantage of requiring considerably fewer total samples to be examined if several drugs are being tested at the same time. Pre-treatment egg counts are not required, and several test groups can be compared to the same control group. You do have to know, or expect, that average egg counts will be above at least 250 epg in the control group for valid comparisons.[2] An electronic spreadsheet-based calculator is available; it will do the calculations and will also indicate "suspected resistance" to a dewormer based on statistical analysis of the input data.[3] Lambs or ewes can be used with either approach but don't mix the two in a test.

When is the best time to perform resistance testing?

This is a good question, and there is no single correct answer. However, testing should probably be done from early to mid-summer. The rationale for this is, at the present time, *Haemonchus contortus* is the most important worm we have to deal with here in our region. Although there are several common species of worms in sheep and goats that produce similar-looking eggs that can't be readily distinguished from *Haemonchus* under the microscope, it is usually safe to assume that by July at least 80–95% of the eggs of this type will be *Haemonchus*. Therefore, resistance testing in mid-summer will give one a good idea of what dewormers will do against this very important worm species. *Haemonchus* season begins earlier in the year in the South, and it may not be the most important worm in the more arid regions of the West, so producers in these areas have to adjust their approach to their conditions.



Manure samples for dewormer resistance testing should preferably be collected from the rectum. A resealable plastic bag inverted over the hand is a convenient way to collect the sample. The sample should be kept at refrigerator temperature, not frozen, until it is examined.

Most parasitologists today recommend conducting resistance testing at least every two years. Testing for resistance does require significant work or expense; however, not knowing whether the dewormer you are using is effective can be more expensive. It can be disastrous.

Notes

1. G.C. Coles et al. The detection of anthelmintic resistance in nematodes of veterinary importance. *Veterinary Parasitology* 136, no. 3–4 (2006): 167–185.
2. G.C. Coles et al. World Association for the Advancement of Veterinary Parasitology (W.A.A.V.P.) methods for the detection of anthelmintic resistance in nematodes of veterinary importance. *Veterinary Parasitology* 44, no. 1–2 (1992) 35–44.
3. S. Love. "Drench Efficacy Calculators—UPDATE!," *Wormmail in the Cloud, August 16, 2011,* wormmailinthecloud.wordpress.com/2011/03/.