<u>Title</u>
Savanna dynamics: linkages between predator-prey-fire-parasite interactions and vegetation dynamics

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<u>Introduction</u>
Since first established by Dr. Anderson in 2010, the overarching goal of the savanna dynamics project has been to understand how a wide range of abiotic and biotic drivers shape the vegetation and animal community dynamics of the Serengeti ecosystem. A key focus is to understand the long-term dynamics of key ecosystem properties, such as tree cover, herbaceous biomass, fire, and herbivore spatial distributions.

<u>Problem Statement</u>
A core question of the Savanna Dynamics project is: what are the major drivers of savanna structure and function? Much of the work to date has focused on top-down or bottom-up determinants, with less examination of reciprocal influences and interactions between multiple factors. A major obstacle in predicting the vegetation dynamics of ecosystems to future perturbations is our limited understanding of how complex suites of connected and covarying traits change from the past conditions under which they evolved to their current role in stability and resilience of ecosystems. We argue that to understand the present ecological responses of ecosystems we must focus on the historical context of the past evolutionary drivers that have brought them to the present time.

<u>General Objective</u>
To understand how migratory herbivore movements and fire impacts parasite dynamics in resident herbivores.

<u>Specific Objectives</u>
(i) We will continue to quantify nematode and tick density in the environment as well as nematode intensity and prevalence in wildebeest and four focal resident species before/after wildebeest migration within our camera grid. For fecal sampling, we will visit camera trap sites following a rotation that maximizes our ability to sample sites varying in terms of expected wildebeest intensity and duration during the migration. Our target sample size is n = 40 fecal samples species-1 month-1 during migratory periods. We will sample intensively over a two-month period centered on the expected time of the wildebeest passage through the camera grid (approx. May-Jun). For pasture L3 sampling, we will use a subset of 20 camera sites per migration event to assess changes in GIN density in the environment. Fecal samples will be placed on ice in the field immediately after collection and then stored at 4oC under anaerobic conditions in the field laboratory until processing. GIN fecal egg counts will be performed using

a modified McMaster method following protocols established in our previous research (Ezenwa 2003). We will also culture a subset of fecal samples collected per species per month for infective larvae (L3) isolation by incubating samples for ~10 days followed by isolation of L3s using a modified Baermann method (Archie and Ezenwa 2011, Budischak et al. 2015). Infective larvae will be isolated and enumerated from pasture clippings (as L3 kg-1 dry herbage) using standard protocols for pasture larval sampling (Hansen and Perry 1994, Ezenwa 2004). Because environmental conditions strongly influence GIN larval development and survival in pasture (Khadijah et al. 2013, Leathwick 2013, Rose et al. 2015), we will also collect continuous data on a number of key environmental covariates at a subset of 20 camera trap sites. At every camera trap, we are using buried Arduino-based wireless dataloggers with GS-1 soil moisture sensors and thermocouples to monitor hourly soil moisture and ground surface temperature. We will supplement these ground-based data with remote sensing data on pasture greenness (NDVI) and fire occurrence based on MODIS and Sentinel satellite imagery.

(ii) Finally, we will continue to track herbivore abundance across space to: i) generate maps of wildebeest occupancy (intensity and duration of use) during the pre- and post-migration phases; and ii) map the abundance of our target resident species and other ungulates. For i, we will first construct time series of wildebeest camera captures. We consider uninterrupted daily sequences containing at least one wildebeest capture as individual events, with the numbers of days in event being defined as 'duration' and the mean number of daily capture events defined as the 'intensity' of the event. For ii, we will use camera-trap data to model occupancy over time, based on established methods developed for our grid (Anderson et al. 2016, Hepler et al. 2018).

(iii) Beginning in March 2024, we will conduct an experiment to simulate wildebeest transient grazing patches through mowing (Cromsigt and Olff 2008), and will monitor subsequent resident herbivore feeding rates, grass regrowth, and pasture L3 density. We will use stratified randomization to select 15 camera sites (blocks) that capture those representing the distributions of our four target resident host species, where we will set up a pair of 400-m2 plots: mowed and control. We will conduct the mowing and subsequent data collection in 5 blocks at a time, over a 1-month period. The mowed plots will simulate grazing resulting from the arrival of WB. Immediately following mowing, we will add freshly-collected wildebeest dung (collected from the migratory WB herds) to each plot. In addition to pasture sampling, we will measure grass height using a disc pasture meter (February et al. 2013) and quantify herbivore visitation rates using dung counts and camera trap data. We will also quantify recent feeding activity by counting grazed stems along transects. We will collect environmental data through buried dataloggers (Holdo et al. 2020) in both experiments.

Expected Output
Our herbivore-parasite-vegetation work will help disentangle the relative role of transport vs. trophic effects in shaping the impacts of migration on parasite distributions in resident hosts. Because of significant spatial overlap between migratory wildebeest and resident livestock, the impact of the migration on parasite burden and species composition in livestock forms an important aspect of this project.

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