Comparative analysis of the metabolically active microbial communities in the rumen of dromedary camels under different feeding systems using total rRNA sequencing

Alaa Rabee [Corresp., 1], **Robert Forster** [2], **Chijioke Elekwachi** [2], **Ebrahim Sabra** [3], **Mebarek Lamara** [4]

[1] Animal and Poultry nutrition department, Desert Research Center, Cairo, Cairo, Egypt

[2] Lethbridge Research and Development Centre, Agriculture and Agrifood Canada, Lethbridge, Alberta, Canada

[3] Genetic Engineering and Biotechnology Research Institute, University of Sadat City, Sadat City, Menoufia, Egypt

[4] Institut de Recherche sur les Forêts, Université du Québec en Abitibi-Témiscamingue, Rouyn-Noranda, Québec, Canada

Corresponding Author: Alaa Rabee
Email address: alaa.bakr.stu@gebri.usc.edu.eg

The breakdown of plant biomass in rumen depends on interactions between bacteria, archaea, fungi, and protozoa; however, the majority of studies of the microbiome of ruminants, including the few studies of the rumen of camels, only studied one of these microbial groups. In this study, we applied total rRNA sequencing to identify active microbial communities in twenty-two solid and liquid rumen samples from eleven camels reared under three feeding systems. These camels were separated in three groups, G1 (n=3), G2 (n=6) and G3 (n=2) and fed Egyptian clover hay and wheat straw and concentrates feed mixture, fresh Egyptian clover, and wheat straw, respectively. Bacteria dominated the libraries of reads generated from all rumen samples, followed by protozoa, archaea, and fungi respectively. Firmicutes, Thermoplasmatales, *Diplodinium,* and *Neocallimastix* dominated bacterial, archaeal, protozoal and fungal communities, respectively in all samples. Feeding systems influenced the microbial diversity and relative abundance of microbial groups; libraries generated from camels fed fresh clover showed the highest alpha diversity. Principal co-ordinate analysis and linear discriminate analysis showed clusters associated with feeding system and the relative abundance of microbes varied between liquid and solid fractions. In addition, the analysis showed positive and negative correlations between the microbial groups. This provides preliminary evidence that bacteria dominate the microbial communities of the camel rumen and that feed changes that microbiome.

Comparative analysis of the metabolically active microbial communities in the rumen of dromedary camels under different feeding systems using total rRNA sequencing

Alaa. M.E. Rabee[1], Robert. J. Forster[2], Chijioke. O. Elekwachi[2], Ebrahim.Sabra[3], Mebarek. Lamara[4]

[1]Animal and Poultry Nutrition Department, Desert Research Center, Cairo, Egypt.
[2]Lethbridg Research and Development Center, Agriculture and Agrifood Canada, Lethbridge, Alberta, Canada .
[3] Genetic Engineering and Biotechnology Research Institute, University of Sadat City, Sadat City, Menoufia, Egypt.
[4]Institut de Recherche sur les Forêts, Université du Québec en Abitibi-Témiscamingue, Rouyn-Noranda, Canada.

Corresponding Author:
Alaa Rabee
Animal and Poultry Nutrition Department, Desert Research Center
1Mathaf El Matariya St.B.O.P.11753
Matariya, Cairo, Egypt
Email: rabee_a_m@yahoo.com
Phone: +20226332846; fax: +20226357858

Abstract

The breakdown of plant biomass in rumen depends on interactions between bacteria, archaea, fungi, and protozoa; however, the majority of studies of the microbiome of ruminants, including the few studies of the rumen of camels, only studied one of these microbial groups. In this study, we applied total rRNA sequencing to identify active microbial communities in twenty-two solid and liquid rumen samples from eleven camels reared under three feeding systems. These camels were separated in three groups, G1 (n=3), G2 (n=6) and G3 (n=2) and fed Egyptian clover hay and wheat straw and concentrates feed mixture, fresh Egyptian clover, and wheat straw, respectively. Bacteria dominated the libraries of sequence reads generated from all rumen samples, followed by protozoa, archaea, and fungi respectively. Firmicutes, Thermoplasmatales, *Diplodinium,* and *Neocallimastix* dominated bacterial, archaeal, protozoal and fungal communities, respectively in all samples. Feeding systems influenced the microbial diversity and relative abundance of microbial groups; libraries generated from camels fed fresh clover showed the highest alpha diversity. Principal co-ordinate analysis and linear discriminate analysis showed clusters associated with feeding system and the relative abundance of microbes varied between liquid and solid fractions. In addition, the analysis showed positive and negative correlations between the microbial groups. This provides preliminary evidence that bacteria dominate the microbial communities of the camel rumen and that feed changes that microbiome.

Introduction

Camels (*Camelus dromedaries*) can produce milk and meat in hot, arid and semi-arid regions and can provide food security as the climate warms (Samsudin et al., 2011; Faye, 2013). Camels also provide textiles (fiber and hair) and are commonly used for transportation, agriculture, tourism, race and riding (Rabee et al., 2019). The unique feeding behavior and the functional structure of digestive tract of these pseudo-ruminants is well adapted to deserts (Kay et al., 1989). The retention time of feed particles in the camel forestomach is longer than other true ruminants, which prolongs the exposure of plant biomasses to the symbiotic microorganisms, which helps the efficient digestion (Lechner-Dolland and Engelhardt, 1989).

Based on feeding type, camel production lies under three systems: camels in traditional extensive system depend on low quality feeds; while, camels in semi-intensive system depend on high-quality forage and camels in intensive system depend on high-quality forage and concentrated supplements (Faye, 2013). Diet and feeding plan, determine the diversity of rumen microbial communities but age, animal breed can also influence the structure of this microbiome (Henderson et al., 2015).

The chemical composition of the diet shapes fermentation in the rumen. For instance, cellulolytic and hemicellulytic diets favor the fibrolytic microorganisms; while, starch and sugars favor the amylolytic (Carberry et al., 2012). Also, the microbial composition and diversity varies between liquid and solid rumen fractions, which might indicate different roles in rumen fermentation; for instance, plant-adherent microbiota might have a major role in fiber degradation (Ren et al., 2020). Digestion in the camel depends on microbial fermentation in the rumen (Samsudin et al., 2011)

and the efficiency of this microbial fermentations is based on the interactions between a wide variety of microbial groups, including bacteria, archaea, fungi and protozoa (Yanagita et al., 2000; Kamra, 2005). Analysis of these microbial communities could lead to increases in animal productivity and reduction of greenhouse gas emissions (Henderson et al., 2015). Unlike other ruminants, camels can utilize thorny and low quality plants like shrubs with high lignocelulolytic content (Samsudin et al., 2011). Consequently, camel rumen microbes must have the capacity to degrade such poor-quality feeds (Gharechahi et al., 2015). However, the microbial community in the rumen of dromedary camel received less attention than other domesticated ruminants.

Most of PCR-based assessments of microbial groups in the rumen have relied on amplicon sequencing, which target a specific variable region on 16S rRNA gene (Li et al. 2016). This approach needs a wide range of primers to study different microbial communities (Kittelmann et al., 2013). Therefore, the output could be biased due to the primer selection and amplification cycling conditions (Guo et al., 2015; Li et al., 2016; Elekwachi et al., 2017). The recent development of next generation sequencing technologies provide a rapid method of microbial identification in rumen and overcome the intrinsic constraints of traditional culture-based methods (Samsudin et al., 2011; Ishaq and Wright, 2014).

Total RNA sequencing (RNA-Seq) offers the advantage of specifically targeting active microbes and avoids biases associated with primer selection and chimera generation in PCR (Gaidos et al., 2011; Guo et al., 2015; Li et al., 2016). In addition, RNA-Seq approach is capable of identifying novel microbes as it is not reliant on primers for known microbes (Li et al., 2016). High-throughput metatranscriptome sequencing provides a comprehensive understanding of the biological systems by characterization of different groups of organisms in the same environment based on the sequencing of coding and noncoding RNA (Elekwachi et al., 2017). Total RNA-Seq was applied to investigate microbial communities in many different systems including, for example, human gut (Qin et al., 2012), and cow rumen (Li et al., 2016; Elekwachi et al., 2017).

Previous microbiome studies on the camel rumen have characterized one or two microbial groups using classical or molecular approaches. For example, the protozoal community in camel rumen was studied heavily by conventional microscopic methods (Ghali et al., 2005; Baraka, 2012). Regarding the anaerobic fungi, a new fungal genus, *Oontomyces* was isolated from the rumen of Indian camel (Dagar et al., 2015), and only one study investigated the whole fungal community in the gut of the camel (Rabee et al., 2019). Only three molecular-based studies are available on the bacterial community (Samsudin et al., 2011; Bhatt et al., 2013; Gharechahi et al., 2015). Furthermore, only one study classified the rumen archaea (Gharechahi et al., 2015).

In the present study, total rRNA sequencing was applied to 1) get insight into the composition of active microbiota in the rumen of camels reared under different feeding systems; 2) describe the distribution of microbial groups among the solid and liquid rumen fractions; 3) investigate the correlations between all the microbial groups.

Materials and Methods

Rumen samples

Rumen samples were collected from eleven adult dromedary camels under three different feeding systems. Camels in group G1 (n=3) were housed in the Maryout Research Station, Alexendria, Egypt and were fed on Egyptian clover hay (*Trifolium alexandrinum*), wheat straw and concentrates feed mixture. Camels in group G2 (n=6) were fed on fresh Egyptian clover (100 % high-quality forage diet) then slaughtered in the Kom Hammada slaughterhouse, Elbehera, Egypt. Camels of group G3 (n=2) were fed on wheat straw (100 % low-quality forage diet) then were slaughtered in Pasateen slaughterhouse, Cairo, Egypt. All the animals kept on the diet for at least one month before the sampling time. The proximate analysis of feeds is illustrated in Supplementary table S1. Details regarding the camel rumen samples in this study presented in Supplementary table S2. The rumen contents were strained immediately by two layers cheesecloth to separate the liquid and solid to form twenty-two samples, frozen using liquid nitrogen and stored at -80^{o}C before further processing (Elekwachi et al., 2017). The project was approved and all samples were collected according to the Institutional Animal Care and Use Committee, Faculty of Veterinary Medicine, University of Sadat City, Egypt (Approval number: VUSC00003).

RNA isolation, quality and quantity estimation and sequencing

The frozen rumen samples were ground using liquid nitrogen. About 0.5 gram of frozen fine powder was used for total RNA isolation using Trizol-Reagent protocol (Invitrogen, Carlsbad, CA), followed by RNA clean up using MEGA clear Kit (Invitrogen). Total RNA quality and quantity were estimated using an Agilent 2100 bioanalyzer (Agilent Technologies, USA) and RNA 6000 Nano kit (Agilent Technologies, USA). One hundred nanogram of total RNA was reverse-transcribed into first strand cDNA and sequenced using Illumina rRNA MiSeq preparation kit (Illumina, USA) by Illumina MiSeq platform.

Bioinformatic data analysis

The generated RNA sequence reads were analysed using pipeline developed by Elekwachi et al. (2017). Briefly, the sequence quality was checked using the FastQC program v. 0.11.4 (Andrews, 2010), then Trimmomatic program v. 0.35 (Bolger et al., 2014) was used to trim adaptors, barcodes, ambiguous and low quality reads. PEAR program v. 0.9.6 (Zhang et al., 2014) was used to merge read 1 and read 2 using default options. Then after, the hidden Markov models rRNA-HMM tool of the rapid analysis of multiple metagenomes with a clustering and annotation pipeline (RAMMCAP) (Li, 2009) was used to sort the reads into archaea and bacteria (16S, 23S), and eukaryote (18S, 23S) rRNA sequences. Merged sequence files were then sub-sampled as needed using MEME program v. 4.10.2 (Bailey et al., 2009). For each sample, 70,000 reads were run through the pipeline. For subsequent analysis steps, 20 000, 10 000, and 2000 sequences were used for bacteria, eukaryote and archaea, respectively. Taxonomy binning for eukaryote and archaeal SSU rRNA sequences was performed using BLASTN. The sub-sampled query sequences were searched against the SILVA SSURef-111 database using an *e*-value of 1e^{-5}. Bacterial SSU sequences were binned into operational taxonomic units (OTUs) using the "classify. seqs" command of Mothur v. 1.33.1 program (Schloss et al., 2009). The SSURef -108 gene and the SSURef-108b taxonomy databases were used. Principal co-ordinate analysis (PCoA) using Bray Curtis dissimilarity and alpha diversity indices (Chao1, Shannon and Inverse Simpson) were evaluated by Mothur (Schloss et al., 2009) based on sub-sampling of 70,000 reads per sample

according the protocol "Community Structure Analysis Based on OTU Clustering" outlined in Elekwachi et al. (2017).

Statistical analyses

Data of relative abundance of bacterial phyla, protozoal genera, fungal genera and archaea genera and order Thermoplasmatales were tested for normality and homogeneity using Shapiro-Wilk test and variables that were deemed non-normal were then arcsine transformed. Linear Discriminate Analysis (LDA) and Bray Curtis Permutational Multivariate Analysis of Variance (PERMANOVA) tests depended on the relative abundance of bacterial phyla. All the protozoal, fungal and archaeal genera and the order Thermoplasmatales were used to show the differences in community structure and to compare the clustering of samples. Pearson correlation analysis was used to identify correlation within and between microbial communities and the correlation scores were visualized as a heatmap. The statistical analyses were performed using the SPSS v. 20.0 software package (SPSS, 1999) and PAST (Hammer et al., 2001). Sequences were deposited to the sequence read archive (SRA) under the accession number: SRP107370.

Results

The composition and diversity of active microbial community

Total rRNA sequencing in twenty-two solid and liquid rumen samples from eleven camels resulted in a total of 3958591 reads with average of 359872 ± 85366 (mean ± standard error (SE)) reads per animal in the solid fraction (SF) and 3386392 reads with an average of 307854 ± 60989 reads per animal in the liquid fraction (LF). The sequence reads of bacteria dominated the active microbial community, followed by protozoa, archaea and fungi (Table 1). The relative abundance of protozoa was higher in LF-G1 (liquid fraction of G1), while the relative abundance of bacteria was higher in SF-G1 (solid fraction of G1). The highest population of archaea was observed in G2 camels. Additionally, G3 camels showed the highest relative abundance of fungi (Table 1; Supplementary Figure 1). Number of OTUs and Alpha-diversity indices, Chao1, Shannon and Inverse Simpson, were higher in the rumen of LF-G2 samples (Table 1).

Bacterial community

The composition of bacterial community varied little between treatments and consisted of 12 phyla. The five most predominant phyla were Firmicutes, Bacteroidetes, Proteobacteria, Spirochaetes and Fibrobacteres, respectively (Table 2). Phylum Firmicutes dominated the bacterial community in all groups and was higher in G2 followed by G1 and G3 camels, respectively, and was also higher in SF compared to LF (Table 2). On the family level, the Firmicutes phylum was dominated by Lachnospiraceae and Ruminococcuceae. In addition, six genera dominated this phylum, including *Butyrivibrio*, RFN8-YE57*, Ruminococcus,* vadinHA42*, Acetitomaculum* and *Blautia* (Fig. 1a and Supplementary Table S3). The second largest phylum, Bacteroidetes, showed the highest relative abundance in G3 followed by G1 and G2 camels and was higher in LF than SF (Fig. 1a and Supplementary Table S3). On the family and genus levels, Bacteroidete*s* was dominated by three families (Prevotellaceae, BS11_ gut_ group, Rikenellaceae) and two genera (*Prevotella,* RC9_gut_group) higher relative abundance in LF-G1 samples and was dominated by Succinivibrionaceae family and *Desulfovibrio* genus (Table 2, Fig. 1a, Supplementary Table S3). The Spirochaetes phylum was higher in the SF-G3 and it was classified into two families including Spirochaetaceae and PL-11B10 and was dominated by *Treponema* genus. The Fibrobacteres phylum was higher in SF-G3 (Table 2, Fig. 1a, Supplementary Table S3). The other

phyla, including Actinobacteria, that was higher in SF-G2 samples, Tenricutes phylum was higher in the LF-G1 samples and Lentisphaerae phylum, was about 3-fold higher in the LF as relative to SF and accounted for a large population in the camels of G3 (Table 2). Additionally, several minor bacterial phyla were also observed in the rumen of camels such as Verrucomicrobia, Elusimicrobia, Cyanobacteria and Chloroflexi (Table 2).

All Bacterial genera were observed in all groups except seven genera, including uncultured *Marinilabiaceae* (Bacteroidetes)*, Quinella* (Firmicutes) and *Streptococcus* (Firmicutes) that were observed only in G2 and G3 camels. *Ruminobacter* (Proteobacteria) was observed only in G1 and G2 camels. On the other hand, *Arcobacter* and *Succinivibrio* within phylum Proteobacteria were observed only in G1 camels and *Betaproteobacteria* (Proteobacteria) was observed only in G3 camels. Moreover, many unclassified bacteria were observed across samples and accounted for 39% of total bacterial reads. Most of these unclassified bacterial reads were observed in phylum Firmicutes and Bacteroidetes.

Archaeal community

Reads that classified as archaea were further classified to three orders within the phylum Euryacheota: Thermoplasmatales, Methanobacteriale and Methanomicrobial. Thermoplasmatales dominated the archaeal community and showed the highest population in LF-G3 camels, this order was not classified out of order level (Table 3, Fig. 1b). Reads that classified in the Methanobacteriale were further classified to family Methanobacteriacea that includes three genera: *Methanobrevibacter*, *Methanophera* and *Methanobacterium. Methanobrevibacter* is the second largest contributor in archaeal population and was higher in SF-G1 camels. *Methanosphaera* exhibited higher relative abundance in SF-G2 camels. *Methanobacterium* was absent in G3 camels; however, a small proportion of this genus was found in the camels of G1 and G2. *Methanomicrobium* genus*,* which belongs to order Methanomicrobiales and family Methanomicrobiaceae was the least contributor in archaeal population and was more prevalent in LF-G3 camels (Table 3, Fig. 1b).

Protozoal community

Reads that classified as protozoa were further classified to two families: Ophryoscolecidae and Isotrichidae (Table 4). Reads that classified in the Ophryoscolecidae were further classified to seven genera, *Diplodinium, Ophryoscolex, Entodinium, Polyplastron, Eudiplodinium, Epidinium* and *Trichostomatia*. Reads that classified in the Isotrichidae were further classified to two genera, *Dasytricha* and *Isotricha*. The variation among the camels in protozoal population was clearly observed and seemed to be higher than other microbial communities; however, the protozoal community composition was similar among the camels (Table 4, Fig. 1c). The most dominant protozoal genera were *Diplodinium, Ophryoscolex* and *Entodinium*. Camels in G1 had the highest population of *Entodinium* and *Epidinium.* Camels in G2 had the greatest population of *Eudiplodinium, Ophryoscolex, Isotricha* and *Dasytricha* and camels in G3 had the greatest population of *Diplodinium, Polyplastron* and *Trichostomatia*. On the sample fraction level, the solid fraction had a higher representation of *Ophryoscolex, Polyplastron, Eudiplodinium, Epidinium* and *Diplodinium,* while the liquid fraction had a higher representation of *Entodinium, Isotricha* and *Dasytricha* (Table 4, Fig. 1c).

Anaerobic rumen fungal community

Reads that classified as rumen fungi were further classified to two phyla: Neocallimastigomycota and Chytridiomycota. Reads that classified in the Neocallimastigomycota were further classified to family Neocallimasticeceae that includes three genera, *Neocallimastix, Piromyces* and *Cyllamyces*. *Neocallimastix* dominated the fungal community, followed by *Piromyces* and *Cyllamyces* (Table 5, Fig. 1d). These anaerobic fungal genera represented > 99.5 % of the fungal population. In addition, reads that classified in the Chytridiomycota were further classified to family Spizellomycetaceae that includes genus *Spizellomyces*, which was noted in a very small proportion (<0.5 %) (Table 5). *Neocallimastix* was more abundant in the SF-G1 samples while *Piromyces* and *Cyllamyces* were more abundant in LF-G2 and SF-G3 respectively (Table 5, Fig. 1d).

Effect of feeding system on the composition of microbial communities

Multivariate analysis separated libraries by feeding system distinctly (Figs. 2 and 3). Also, bacteria, dominated by phylum Firmicutes were the main driver of differences between animals (Fig. 3). Furthermore, *Entodinium*, Thermoplasmatales, *Neocallimastix* were the main drivers of differences in protozoal, archaeal and fungal communities, respectively. PERMANOVA analysis revealed that the difference between camel groups was significant ($P < 0.01$) in all microbial groups (Supplementary Table S4). Pairwise comparison between camel groups based on Bonferroni-corrected p-value demonstrated that the difference was significant ($P < 0.05$) between camels of G2 and G3 in bacterial and archaeal communities (Supplementary Table S4). Moreover, the difference was significant between the three groups in the protozoal community ($P < 0.05$), whereas, in the fungal community, the difference was significant only between camels in group G1 and G2 (Supplementary Table S4).

Pearson correlation between microbes in the rumen of dromedary camel

Pearson correlation analysis (Fig. 4A, 4B), revealed many significant positive and negative correlations ($P < 0.05$). For example, in active bacteria, Bacteroidetes correlated positively with *Cyllamyces* and negatively with *Butyrivibrio*, *Methanosphaera* and *Trichostomatia*. Prevotellaceae correlated positively with *Neocallimastix* and *Entodinium* and negatively with Ruminococcaceae, *Methanosphaera* and *Diplodinium*. *Fibrobacteres* correlated positively with *Cyllamyces, Methanomicrobium,* Thermoplasmatales and *Diplodinium* and negatively with *Methanosphaera, Epidinium*, Ruminococcaceae and *Butyrivibrio*. Firmicutes correlated positively with *Methanosphaera* and negatively with *Piromyces*, Thermoplasmatales and *Methanomicrobium*.

In active archaea, Thermoplasmatales correlated positively with *Diplodinium* and negatively with *Methanobrevibacter* and *Methanosphaera.* In active protozoa, there was a negative correlation between *Polyplastron*, *Entodinium*, *Ophryoscolex* and *Epidinium.* In active fungi, a negative correlation was observed between *Cyllamyces*, *Neocallimastix* and *Piromyces* and between *Piromyces* and *Entodinium*.

Discussion

Rumen microbes can ferment a wide variety of feed components, including cellulose, xylan, amylose and protein and produce volatile fatty acids that provide the animal with approximately 70% of daily energy requirements (Bergman, 1990; Henderson et al., 2015). Furthermore, the rumen fermentation generates methane, which contributes to global warming and represents 2–12% loss of feed energy for the animal (Johnson and Ward, 1996; Carberry et al., 2012; Jami et al., 2014). Therefore, investigation of these microbial communities is the key to understand their roles and maximize ruminal fermentation and fiber digestion and reduction of greenhouse gas emissions (Lee et al., 2012).

Camels groups fed different diets and reared in different locations. However, the diet type has the main effect on the diversity and relative abundance of microbial communities. This speculation is supported by the similarity of microbial groups across the samples. Furthermore, the variation in the relative abundance of microbial groups was associated with diet composition, more details could be seen in Supplementary note S1. In addition, PCoA, LDA and PERMANOVA analyses confirmed the finding of this study and was in agreement with the results of other ruminant studies (Henderson et al., 2015). Camels in the present study were fed on different forages; Egyptian clover and wheat straw (Supplementary Table S1). Egyptian clover is the most balanced and nutritious fodder widely used for feeding camels (Carberry et al., 2012; Bakheit, 2013; Shrivastava et al., 2014), which might supported the high microbial diversity in G2 camels compared to other groups (Table 1). This was consistent with previous studies on cows (Pitta et al., 2010; Shanks et al., 2011; Kumar et al., 2015). Highly degradable carbohydrates support the bacterial and protozoal growth (Dijkstra and Tamminga, 1995; Kumar et al., 2015), which could demonstrate their higher population in G1 camels. Additionally, the higher bacterial population slows the fungi growth (Stewart et al., 1992; Orpin and Joblin, 1997), which was illustrated by the low fungal population in G1 camels.

Bacterial community

Firmicutes phylum was appeared more abundant than Bacteroidetes and both phyla comprised > 75% of all bacterial reads (Table 2), which is in agreement with previous studies on different animals including camels (Samsudin et al., 2011), Surti Buffalo (Pandya et al., 2010) and Muskoxen (Salgado-Flores et al., 2016). The majority of Firmicutes' members have a potential role in fiber digestion, which might illustrate their higher population in G2 camels that were fed on high-quality forage and also in solid fraction. This speculation was supported by the high proportion of Ruminococcaceae and Lachnospiraceae families that found to be active in fiber digestion in the rumen (Pitta et al., 2014a; Nathani et al., 2015). Bothe *Blautia* and *Acetitomaculum* genera have a key role as reductive acetogens (Le Van et al., 1998; Yang et al., 2016) and varied with feeding system among the camel groups in this study. This finding could indicate that the reductive acetogenesis pathway could be maximized by diet to minimize methane production (Le Van et al., 1998).

Bacteroidetes were higher in poor-quality diet (G3), which was similar to results found in cattle (Pitta et al., 2014b). The phylum was dominated by family Prevotellaceae, which confirms Gharechahi et al. (2015). The members of Bacteroidetes possess diverse enzymes that can target cellulose, pectin and soluble polysaccharides released in the liquid phase (Mackenzie et al., 2015). Additionally, *Prevotella* genus is involved in propionate production that is used for energy by the

host (Nathani et al., 2015). We speculate that Bacteroidetes species contribute to the adaptation of camels to arid conditions.

The RC9_gut_group found in this study belongs to uncultured genera and was found also in the gut of Rhinoceros hindgut (Bian et al., 2013). Unclassified Bacteroidetes are specialized in lignocellulose degradation (Mackenzie et al., 2015), which could support their high proportion in G3 camels. Fibrobacteres was higher (3.1%) in this study compared to the other findings on camels (Gharechahi et al., 2015); this phylum is the principal cellulolytic bacteria in the rumen (Ransom-Jones et al., 2012; Nathani et al., 2015), which might illustrate its higher relative abundance in solid fraction and in the rumen of camels fed on wheat straw (G3) (Table 2). The members of Proteobacteria were lower in G2 and G3 camels that were fed on diet rich in fiber contents. These findings highlighted this phylum's function as a protein-degrading bacteria as it was reported by Liu et al. (2017). The abundance of *Treponema* was higher in the solid fraction and in G3 camels (Figure 1a). *Treponema* is the dominant genus in Spirochaetes phylum and it is fiber-associated bacteria, which could indicate to its cellulytic and xylanolytic activities (Ishaq and Wright, 2012).

The dominant bacterial genera in this study were *Butyriovibrio,* RFN8-YE57*, Ruminococcus*, *Prevotella*, *Fibrobacter*, *Treponema* and VadinHA. These genera were higher in the SF except RFN8-YE57 compared to the LF; this finding was consistent with a previous study on camels (Gharechahi et al., 2015), and confirms that the solid-attached microbes could play a major role in ruminal fiber digestion (Jewell et al., 2015; Noel et al., 2017).
Most of Elusimicrobia in this study were uncultured; some members of this phylum were isolated from the termite's gut that degrades cellulose (Herlemann et al., 2009). Therefore, we speculate that this phylum has a role in fiber digestion and that might illustrate their high proportion in G3 camels. Actinobacteria observed also in the rumen of moose and some members of this phylum have acetogenic activities (Ishaq et al., 2015). Some members of *Victivallis* within Lentisphaerae phylum were involved in cellobiose degrading activity (Zoetendal et al., 2003).

Unclassified bacteria in our study (39% of total bacterial reads) were less than the percentage found in a study of Muskoxen (53.7-59.3%) (Salgado-Flores et al., 2016). The presence of unclassified bacteria in the gut was commonly observed (Gruninger et al., 2016) and could be a result of the presence of new bacteria that ferment plant biomass (Salgado-Flores et al., 2016) or related to short reads were generated from RNAsequncing (Li et al., 2016).

Archaeal community

The archaeal population has important roles in methane emission mitigation strategies as they convert the H_2 and CO_2 produced in the rumen to methane (Hook et al., 2010). Additionally, acetate produced in fiber breakdown is used to provide a methyl group for methanogenesis; therefore, methanogens population could be shifted by alteration of diet composition or feed additives and plant compounds (Hook et al., 2010; Tapio et al., 2017), which could demonstrate the variation in the relative abundance of archaea between camel groups. Camels of the second group (G2) that fed fresh clover, showed the highest archaeal population (Table 2) and the archaeal community was dominated by Thermoplasmatales, a methylotrophic methanogens order (Table 3) which was consistent with the results on cattle (Carberry et al., 2014) and camels (Gharechahi et al., 2015). Thermoplasmatales produces methane from methyl amine and its population was decreased by the addition of rapeseed oil to animal diet, making it a high potential target in future strategies to mitigate methane emissions (Poulsen et al., 2013). The *Methanobrevibacter,*

Methanosphaera, *Methanomicrobium* and *Methanobacterium* (Table 4) are the other dominant archaea in this study and in accordance with the results found in beef cattle (Carberry et al., 2014). *Methanobrevibacter* dominated the methanogens in other ruminant (Henderson et al., 2015) and was associated with high methane emissions (Tapio et al., 2017). Moreover, *Methanomicrobium* had its highest proportion with the feeding system of poor quality forage diet (G3), which was similar to results found in buffalo (Franzolin and Wright, 2016), and *In vitro* (Wang et al., 2018). In rumen, *Methanomicrobium* has been shown to be responsible for the conversion of H_2 and/or formate into CH_4 (Leahy et al., 2013). The abundance of Thermoplasmatales was also negatively correlated with *Methanobrevibacter* which is consistent with previous results (Danielsson et al., 2017; McGovern et al., 2017).

Protozoal community

The majority of protozoal reads were related to *Diplodinium*, *Ophryoscolex*, *Entodinium*, *Polyplastron*, *Eudiplodinium* and *Epidinium* (Table 4). Similar findings were observed in other study on different ruminants (Baraka, 2012). The relative abundance of protozoal was influenced by feeding system, which was in the same line with results on cattle (Hristov et al., 2001; Weimer, 2015). The *Diplodinium* dominated the protozoal community and was prevalent in the G3 camels, which highlighted the cellulolytic activity of this genus (Coleman et al., 1976). Also, some species of genus *Diplodinium* were discovered in the rumen of Egyptian camel and is considered to be peculiar in camel such as *Diplodinium cameli*, (Kubesy and Dehority, 2002). In addition, *Entodinium* was higher in G1 camels that were fed on concentrates feed mixture that increase the protozoa. Also, previous studies showed that this genus was dominant in rumen of camels (Selim et al., 1999; Ghali et al., 2005) and cattle (Carberry et al., 2012). Moreover, the study of Kittelmann and Janssen (2011) showed that the *Polyplastron* was the dominant genus in cattle. On the function level, all the genus *Eudiplidinum*, *Epidinum* and *Diplodinum* have cellulolytic activity (Coleman et al., 1976), whereas, *Polyplastrone* and *Epidinium* have a xylanolytic activity (Devillard, 1999; Béra-Maillet et al., 2005).

Anaerobic rumen fungal community

The highest fungal population was observed in the solid fraction and rumen of G3 camels (Table 1). These findings were in agreement with the results of different studies stated that the fibre-based diets stimulated the fungal growth (Orpin, 1977; Roger et al., 1993; Kamra et al., 2005; Haitjema et al., 2014). This speculation could explain the low fungal population in G1 camels in our study. Moreover, the longer retention time and neutral pH in camel's forestomach (Russell and Wilson, 1996) make it more suitable for the survival of rumen fungi. The genus *Neocallimastix* dominated the fungal community and found to be higher in the G1 camels which was similar to other results on sheep and camels (Kittelmann et al., 2013, Rabee et al., 2019). This genus produces enzymes capable of hydrolyzing cellulose, xylan and starch (Pearce and Bauchop, 1985). *Cyllamyces* that was observed in small population, has the ability to degrade poor-quality feeds (Sridhar et al., 2014), which might explain its high population in solid fraction and G3 camels. *Piromyces* was the second dominant genus in the camel rumen of this study and has been shown to produce cellulolytic and xylanolytic enzymes (Teunissen et al., 1992). Therefore, the fungi were more prevalent in ruminants of G2 camels, which fed high-quality forage with high fiber contents than in G2 and G3 camels. The genus *Spizellomyces* is closely related to Chytridiomctes (Bowman et al., 1992), and common in grassland and crop soil (Lozupone and Klein, 2002, Kittelmann et al.,

2012). Thus, the presence of this fungus in the camel rumen in the current study could be explained by a contamination of the forages by soil.

Correlation between rumen microbes

The interactions between rumen microbes are the main driver of feed degradation and methane formation in the rumen, which influence the animal production and the environment (Williams et al., 1994; Lee et al., 2012; Henderson et al., 2015). Positive and negative correlations were observed within and between microbial communities in this study (Fig. 4). Methanogens colonize the protozoa and this relationship was believed to enhance methane formation (Newbold et al., 1995), which highlighted some positive correlations between protozoa and archaea. Additionally, the fibrolytic bacteria produce the important substrates mainly hydrogen and methyl groups that methanogens use for growth (Johnson and Johnson, 1995), which demonstrated the positive correlations found between *Fibrobacteres* and some methanogens. Also, positive correlation between the methylotrophic *Methanosphaera* and *Lachnospiraceae* that has been implicated in pectin degradation and provides methanol as a substrate for the methylotrophs (Dehority, 1969). On the other hand, *Prevotella* is a hydrogen utilizer and produces propionate which has a negative impact on methanogenesis in the rumen (Pitta et al., 2014a; Liu et al., 2017), which also illustrated the negative correlation obtained in this study between Prevotellaceae and archaea.

Since the rumen anaerobic fungi produce abundant H_2 through the fermentation of carbohydrate; they can interact positively with H_2 utilizers such as archaea, Prevotellaceae, *Blautia* and *Acetitomaculum* (Orpin and Joblin, 1997; Le Van et al., 1998; Yang et al., 2016; Liu et al., 2017). Additionally, the anaerobic fungi penetrate plant tissue, which provides an increased surface area for bacterial colonization (Orpin and Joblin, 1997), which could explain the positive correlation between fungi and both *Butyrivibrio* and *Fibrobacteres* in this study. However, fungi are known to be negatively impacted by the presence of some bacteria and protozoa as the fungal zoospores are likely to be a prey for protozoa (Morgavi et al., 1994), which demonstrated the negative correlation between both *Neocallimastix* and *Piromyces* with *Diplodinium* and *Entodinium.* Furthermore, *Ruminococcus* produces compounds that inhibit the growth of rumen fungi (Stewart et al., 1992), which support the negative correlation between *Neocallimastix* and Ruminococcaceae. *Polyplastron* predates upon other protozoa like *Epidinium, Eudiplodinium, Diplodinium,* and *Ostracodinium* (Eadie, 1967), which might explained the negative correlation between Polyplastron and other Protozoa.

Conclusions

This study applied total rRNA sequencing to get insight into the active microbial groups in the rumen of dromedary camels. However, using the DNA-amplicon sequencing with RNA sequencing is recommended in the future studies to compare the composition of active microbial groups (from RNA sequencing) with the composition of the whole microbial community. Furthermore, it is recommended to use larger population in future studies. As a major conclusion of our study, the microbial community in camel rumen was diverse and similar in composition between the camels. However, the feeding system impacted the relative abundance of active microbial communities where the fresh Egyptian clover provided the highest microbial diversity. The majority of camel rumen microbes (bacteria, fungi, and protozoa) were fibrolytic or have a possible role in fiber digestion, which might illustrate the ability of camel to live in desert harsh conditions under poor feeds. Moreover, the structure of microbial community in rumen of camel

found to be similar to other ruminant studies with a shown difference in the relative abundances. The present results should open new perspectives for further cultivation and isolation studies on the unclassified microorganisms found in the rumen of camels to classify them and assign their functions.

References

Andrews S. 2010. FastQC A Quality Control Tool for High Throughput Sequence Data. Available at: http://www.bioinformatics.babraham.ac.uk/projects/fastqc/

Bakhei, BR. 2013. Egyptian clover (*Trifolium alexandrinum*) breeding in Egypt: A review. *Asian Journal of crop science* 5: 325-337 DOI 10.3923/ajcs.2013

Bailey TL, Boden M, Buske FA, Frith M, Grant CE, Clementi L, Ren J, Li WW, Noble WS. 2009. MEME SUITE: tools for motif discovery and searching. *Nucleic acids research* 37(Web Server issue): W202–W208 DOI 10.1093/nar/gkp335

Baraka TA. 2012. Comparative studies of rumen pH, total protozoa count, generic and species composition of ciliates in camel, buffalo, cattle, sheep and goat in Egypt. *Journal of american science* 8: 448-462.

Bergman EN. 1990. Energy contributions of volatile fatty acids from the gastrointestinal tract in various species. *Physiological reviews* 70: 567-90 DOI 10.1152/physrev.1990.70.2.567.

Béra-Maillet C, Devillard E, Cezette M, Jouany J, Forano E. 2005. Xylanases and carboxymethylcellulases of the rumen protozoa *Polyplastron multivesiculatum*, *Eudiplodinium maggii* and *Entodinium sp*. *FEMS Microbiology letters* 244: 49-156 DOI 10.1016/j.femsle.2005.01.035.

Bhatt VD, Dande SS, Patil NV, Joshi CG. 2013. Molecular analysis of the bacterial microbiome in the forestomach fluid from the dromedary camel (*Camelus dromedarius*). *Molecular biology reports* 40: 3363-3371 DOI 10.1007/s11033-012-2411-4.

Bian G, Ma L, Su Y, Zhu W. 2013. The microbial community in the feces of the white Rhinoceros (*Ceratotherium simum*) as determined by barcoded pyrosequencing analysis. *PLoS One* 8: e70103 DOI 10.1371/journal.pone.0070103.

Bolger AM, Lohse M, Usadel B. 2014. Trimmomatic: a flexible trimmer for Illumina sequence data. *Bioinformatics* 30: 2114–2120 DOI 10.1093/bioinformatics/btu170.

Bowman BH, Taylor JW, Brownlee AG, Lee J, Lu SD, White TJ. 1992. Molecular evolution of the fungi: relationship of the Basidiomycetes, Ascomycetes, and Chytridiomycetes. *Molecular biology and evolution* 9: 285-296 DOI 10.1093/oxfordjournals.molbev.a040720.

Carberry CA, Waters SM, Kenny DA, Creevey CJ. 2014. Rumen methanogenic genotypes differ in abundance according to host residual feed intake phenotype and diet type. *Applied and environmental microbiology* 80: 586-594 DOI 10.1128/AEM.03131-13.

Carberry CA, Kenny DA, Han S, McCabe MS, Waters SM .2012. Effect of phenotypic residual feed intake and dietary forage content on the rumen microbial community of beef cattle. *Applied and environmental microbiology* 78: 4949-4958 DOI 10.1128/AEM.07759-11.

Coleman GS, Laurie J I, Bailey JE, Holdgate SA. 1976. The cultivation of cellulolytic protozoa isolated from the rumen. *Journal of general microbiology* 95: 144-150 DOI 10.1099/00221287-95-1-144.

Dagar SS, Kumar S, Griffith GW, Edwards JE, Callaghan TM, Singh R, Nagpal AK, Puniya A K. 2015. A new anaerobic fungus (*Oontomyces anksri gen. nov., sp. nov*.) from the digestive tract of the Indian camel (*Camelus dromedarius*). *Fungal biology* 119:731-737 DOI 10.1016/j.funbio.2015.04.005.

Danielsson R, Dicksved J, Sun L, Gonda H, Müller B, Schnürer A, Bertilsson J. 2017. Methane production in dairy cows correlates with rumen methanogenic and bacterial community structure. *Frontiers in microbiology* 8: 226 DOI 10.3389/fmicb.2017.00226.

Dehority BA. 1969. Pectin-fermenting bacteria isolated from the bovine rumen. *Journal of bacteriology* 99: 189–196.

Devillard E, Newbold CJ, Scott KP, Forano E, Wallace RJ, Jouany JP, Flint HJ. 1999. A xylanase produced by the rumen anaerobic protozoan *Polyplastron multivesiculatum* shows close sequence similarity to family 11 xylanases from Gram-positive bacteria. *FEMS Microbiology letters* 181: 145-152 DOI 10.1111/j.1574-6968.1999.tb08837.x.

Dijkstra J, Tamminga S. 1995. Simulation of the effects of diet on the contribution of rumen protozoa to degradation of fibre in the rumen. *British journal of nutrition* 74: 617-634 DOI 10.1079/BJN19950166.

Eadie JM. 1967. Studies on the ecology of certain rumen ciliate protozoa. *Journal of general microbiology* 49: 175-194 DOI 10.1099/00221287-49-2-175.

Elekwachi CO, Wang Z, Wu X, Rabee A, Forster RJ. 2017. Total rRNA-Seq analysis gives insight into bacterial, fungal, protozoal and archaeal communities in the rumen using an optimized RNA isolation method. *Frontiers in microbiology* 8: 1814 DOI 10.3389/fmicb.2017.01814.

Faye B. 2013. Camel farming sustainability: The challenges of the camel farming system in the XXIth century. *Journal of sustainable development* 6: 74-82 DOI 10.5539/jsd.v6n12p74.

Franzolin R, Wright AG. 2016. Microorganisms in the rumen and reticulum of buffalo (*Bubalus bubalis*)fed two different feeding systems. *BMC Research notes* 9: 243 DOI 10.1186/s13104-016-2046-y.

Gaidos E, Rusch A, Ilardo M. 2011. Ribosomal tag pyrosequencing of DNA and RNA from benthic coral reef microbiota: community spatial structure, rare members and nitrogen-cycling guilds. *Environmental microbiology* 13: 1138–1152 DOI 10.1111/j.1462-2920.2010.02392.x.

Ghali MB, Scott PT, Jassim RAM. 2005. Effect of diet change on population of rumen protozoa in dromedary camel. *Recent advances in animal nutrition in Australia.* 15: 27A. Available at: http://livestocklibrary.com.au/handle/1234/20056. Accessed June 8, 2020.

Gharechahi J, Zahiri HS, Noghabi KA, Salekdeh GH. 2015. In-depth diversity analysis of the bacterial community resident in the camel rumen. *Systematic and applied microbiology* 38: 67-76 DOI 10.1016/j.syapm.2014.09.004.

537 Gruninger RJ, McAllister TA, Forster RJ. 2016. Bacterial and archaeal diversity in the
538 gastrointestinal tract of the orth American Beaver (*Castor canadensis*). *PLoS ONE* 11: e0156457
539 DOI 10.1371/journal.pone.0156457.

540 Guo J, Cole J, Zhang Q, Brown C, Tiedje J. 2015. Microbial community analysis with ribosomal
541 gene fragments from shotgun metagenomes. *Applied and environmental microbiology* 82: 157-
542 166 DOI 10.1128/AEM.02772-15.

543 Haitjema CH, Solomon KV, Henske JK, Theodorou MK, O'Malley MA. 2014. Anaerobic gut
544 fungi: Advances in isolation, culture, and cellulolytic enzyme discovery for biofuel production.
545 *Biotechnology and bioengineering* 111: 1471-1482 DOI 10.1002/bit.25264.

546 Hammer Ø, Harper DAT, Ryan PD. 2001. PAST: Paleontological statistics software package for
547 education and data analysis. *Palaeontologia electronica* 4: 9 https://palaeo-
548 electronica.org/2001_1/past/issue1_01.htm.

549 Henderson G, Cox F, Ganesh S, Jonker A, Young W, Janssen PJ. 2015. Rumen microbial
550 community composition varies with diet and host, but a core microbiome is found across a wide
551 geographical range. *Scientific reports* 5: 14567 DOI 10.1038/srep14567.

552 Herlemann DPR, Geissinger O, Ikeda-Ohtsubo W, Kunin V, Sun H, Lapidus A, Hugenholtz P, d
553 Brune A. 2009. Genomic analysis of "*Elusimicrobium minutum*," the first cultivated
554 representative of the phylum "*Elusimicrobia*" (formerly termite group 1). *Applied and*
555 *environmental microbiology* 70: 2841-2849 DOI 10.1128/AEM.02698-08.

556 Hook SE, Wright ADG, McBride BW. 2010. Methanogens: methane producers of the rumen and
557 mitigation strategies. *Archaea* 945785 http://dx.doi.org/10.1155/2010/945785.

558 Hristov AN, Ivan M, Rode LM, McAllister TA. 2001. Fermentation characteristics and ruminal
559 ciliate protozoal populations in cattle fed medium- or high-concentrate barley-based diets.
560 *Journal of animal science* 79: 515-524 DOI 10.2527/2001.792515x.

561 Ishaq SL, Wright AG. 2012. Insight into the bacterial gut microbiome of the North American
562 moose (*Alces alces*). *BMC Microbiology* 12: 212 DOI 10.1186/1471-2180-12-212.

563 Ishaq SL, Wright ADG. 2014. High-throughput DNA sequencing of the ruminal bacteria from
564 moose (*Alces alces*) in Vermont, Alaska, and Norway. *Microbial ecology* 68: 185-195 DOI
565 10.1007/s00248-014-0399-0.

566 Ishaq S, Sundset M, Crouse J, Wright A. 2015. High-throughput DNA sequencing of the moose
567 rumen from different geographical locations reveals a core ruminal methanogenic archaeal
568 diversity and a differential ciliate protozoal diversity. *Microbial genomics* 1: e000034 DOI
569 10.1099/mgen.0.000034.

570 Jami E, White BA, Mizrahi I. 2014. Potential role of the bovine rumen microbiome in
571 modulating milk composition and feed efficiency. *PLoS ONE* 9: e85423 DOI
572 10.1371/journal.pone.0085423.

573 Jewell KA, McComirck C, Odt CL, Weimer PJ, Suen G. 2015. Ruminal bacterial community
574 composition in dairy cows is dynamic over the course of two lactations and correlates with feed
575 efficiency. *Applied and environmental microbiology* 18: 4697-4710 DOI 10.1128/AEM.00720-
576 15.

Johnson KA, Johnson DE. 1995. Methane emissions from cattle. *Journal of animal science* 73: 2483-2492 DOI 10.2527/1995.7382483x.

Johnson DE, Ward GM. 1996. Estimates of animal methane emissions. *Environmental monitoring and assessment* 42: 113-141 DOI 10.1007/BF00394046.

Kamra DN. 2005. Rumen microbial ecosystem. *Current science* 89 124–135.

Kay RNB, Maloiy MO. 1989. Digestive secretions in camels. Options Méditerranéennes – Série Séminaires - n." 2 : 83-87.

Kittelmann S, Naylor GE, Koolaard JP, Janssen PH. 2012. A proposed taxonomy of anaerobic fungi (Class Neocallimastigomycetes) suitable for large-scale sequence-based community structure analysis. *PLoS ONE* 7: e36866 DOI 10.1371/journal.pone.0036866.

Kittelmann S, Seedorf H, Waters WA, Clemente JC, Knight R, Gordon J, Janssen PH. 2013. Simultaneous amplicon sequencing to explore co-occurrence patterns of bacterial, archaeal and eukaryotic microorganisms in rumen microbial communities. *PloS ONE* 8: e47879 DOI 10.1371/journal.pone.0047879.

Kittelmann S, Janssen PH. 2011. Characterization of rumen ciliate community composition in domestic sheep, deer, and cattle, feeding on varying diets, by means of PCR-DGGE and clone libraries. *FEMS Microbiology ecology* 75: 468-481 DOI 10.1111/j.1574-6941.2010.01022.x.

Kubesy AA, Dehority BA. 2002. Forestomach ciliate Protozoa in Egyptian dromedary camels (*Camelus dromedarius*). ZOOTAXA 51: 1-12.

Kumar S, Indugu N, Vecchiarelli B, Pitta DW. 2015. Associative patterns among anaerobic fungi, methanogenic archaea, and bacterial communities in response to changes in diet and age in the rumen of dairy cows. *Frontiers in microbiology* 6: 781 DOI 10.3389/fmicb.2015.00781.

Le Van TD, Robinson JA, Ralph J, Greening RC, Smolenski WJ, Leedle JA, Schaefer DM. 1998. Assessment of reductive acetogenesis with indigenous ruminal bacterium populations and *Acetitomaculum ruminis*. *Applied and environmental microbiology* 64: 3429-36.

Leahy S, Kelly W, Ronimus R, Wedlock N, Altermann E, Attwood G. 2013. Genome sequencing of rumen bacteria and archaea and its application to methane mitigation strategies. *Animal* 7: 235-243 DOI 10.1017/S1751731113000700.

Lechner-Doll M, Engelhardt WV. 1989. Particle size and passage from the forestomach in camels compared to cattle and sheep fed a similar diet. *Journal of animal physiology and animal nutrition* 61:120–128 https://doi.org/10.1111/j.1439-0396.1989.tb00091.x.

Lee HJ, Jung JY, Oh YK, Lee SS, Madsen EL, Jeon CO. 2012. Comparative survey of rumen microbial communities and metabolites across one Caprine and three Bovine groups, using bar-coded pyrosequencing and ^{1}H nuclear magnetic resonance spectroscopy. *Applied and environmental microbiology* 78: 5983-5993 DOI 10.1128/AEM.00104-12.

Li W. 2009. Analysis and comparison of very large metagenomes with fast clustering and functional annotation. *BMC Bioinformatics* 10:359 DOI 10.1186/1471-2105-10-359.

Li F, Henderson G, Sun X, Cox F, Janssen PH, Guan LL. 2016. Taxonomic assessment of rumen microbiota using total RNA and targeted amplicon sequencing approaches. *Frontiers in microbiology* 7 : 987 DOI 10.3389/fmicb.2016.00987.

Liu K, Xu Q, Wang L, Guo W, Zhou M. 2017. The impact of diet on the composition and relative abundance of rumen microbes in goat. *Asian-Australasian journal of animal sciences* 30: 531–537 DOI 10.5713/ajas.16.0353.

Lozupone CA, Klein DA. 2002. Molecular and cultural assessment of chytrid and *Spizellomyces* populations in grassland soils. *Mycologia* 94: 411-420 DOI 10.1080/15572536.2003.11833206.

Mackenzie AK, Naas AE, Kracun SK, Schuckel J, Fangel JU, Agger JW, Willats WG, Eijsink VG, Pope PB. 2015. A polysaccharide utilization locus from an uncultured Bacteroidetes phylotype suggests ecological adaptation and substrate versatility. *Applied and environemental microbiology* 81: 187-195 DOI 10.1128/AEM.02858-14.

McGovern E, McCabe MS, Cormican P, Popova M, Keogh K, Kelly AK, Kenny DA, Waters S M. 2017. Plane of nutrition affects the phylogenetic diversity and relative abundance of transcriptionally active methanogens in the bovine rumen. *Scientific reports* 7: 13047 https://doi.org/10.1038/s41598-017-13013-y.

Morgavi DP, Sakurada M, Mizokami M, Tomita Y, Onodera R. 1994. Effects of ruminal protozoa on cellulose degradation and the growth of an anaerobic ruminal fungus, *Piromyces sp* strain OTS1, in vitro. *Applied and environmental microbiology* 60: 3718-3723.

Nathani NM, Patel AK, Mootapally CS, Reddy B, Shah SV, Lunagaria PM, Kothari RK, Joshi CG. 2015. Effect of roughage on rumen microbiota composition in the efficient feed converter and sturdy Indian Jaffrabadi buffalo (*Bubalus bubalis*). *BMC Genomics* 16: 1116 DOI 10.1186/s12864-015-2340-4.

Newbold CJ, Lassalas B, Jouany JP. 1995. The importance of methanogens associated with ciliate protozoa in ruminal methane production in vitro. *Letters in applied microbiology* 21: 230-234 DOI 10.1111/j.1472-765X.1995.tb01048.x.

Noel SJ, Attwood GT, Rakonjac J, Moon CD, Waghorn GC, Janssen PH. 2017. Seasonal changes in the digesta-adherent rumen bacterial communities of dairy cattle grazing pasture. *PLoS ONE* 12: e0173819 DOI 10.1371/journal.pone.0173819.

Orpin CG. 1977. The rumen flagellate *Piromonas communis*: its life cycle and invasion of plant material in the rumen. *Journal of general microbiology* 99: 107-117 DOI 10.1099/00221287-99-1-107.

Orpin CG, Joblin KN. 1997. The rumen anaerobic fungi. In: The Rumen Microbial Ecosystem (Hobson PN & Stewart CS, eds), pp. 140-184. Blackie Academic and Professional, London.

Pandya PR, Singh KM, Parnerkar S, Tripathi AK, Mehta HH, Rank DN, Kothari RK, Joshi CG. 2010. Bacterial diversity in the rumen of Indian Surti buffalo (*Bubalus bubalis*), assessed by 16S rDNA analysis. *Journal of applied genetics* 51: 395-402 DOI 10.1007/BF03208869.

Pearce PD, Bauchop T. 1985. Glycosidases of the rumen anaerobic fungus *Neocallimastix frontalis* grown on cellulosic substrates. *Applied and environmental microbiology* 49: 1265-1269.

Pitta DW, Parmar N, Patel AK, Indugu N, Kumar S, Prajapathi KB, Patel AB, Reddy B, Joshi C. 2014b. Bacterial diversity dynamics associated with different diets and different primer pairs in the rumen of Kankrej cattle. *PLoS ONE* 9: e111710 DOI 10.1371/journal.pone.0111710.

Pitta DW, Kumar S, Veiccharelli B, Parmar N, Reddy B, Joshi CG. 2014a. Bacterial diversity associated with feeding dry forage at different dietary concentrations in the rumen contents of Mehshana buffalo (*Bubalus bubalis*) using 16S pyrotags. *Anaerobe* 25: 31-41 DOI 10.1016/j.anaerobe.2013.11.008.

Pitta DW, Pinchak E, Dowd SE, Osterstock J, Gontcharova V. 2010. Rumen bacterial diversity dynamics associated with changing from Bermuda grass hay to grazed winter wheat diets. *Microbial ecology* 59: 511-522 DOI 10.1007/s00248-009-9609-6.

Poulsen M, Schwab C, Jensen B, Engberg R, Spang A, Canibe N, Højberg O, Milinovich G, Fragner L, Schleper C, Weckwerth W, Lund P, Schramm A, Urich T. 2013. Methylotrophic methanogenic Thermoplasmata implicated in reduced methane emissions from bovine rumen. *Nature communications* 4: 1428 DOI 10.1038/ncomms2432.

Qin J, Li Y, Cai Z, Li S, Zhu J, Zhang F, Liang S, Zhang W, Guan Y, Shen D, Peng Y, Zhang D, Jie Z, Wu W, Qin Y, Xue W, Li J, Han L, Lu D, Wu P, Dai Y, Sun X, Li Z, Tang A, Zhong S, Li X, Chen W, Xu R, Wang M, Feng Q, Gong M, Yu J, Zhang Y, Zhang M, Hansen T, Sanchez G, Raes J, Falony G, Okuda S, Almeida M, LeChatelier E, Renault P, Pons N, Batto JM, Zhang Z, Chen H, Yang R, Zheng W, Li S, Yang H, Wang J, Ehrlich SD, Nielsen R, Pedersen O, Kristiansen K, Wang J. 2012. A metagenome wide association study of gut microbiota in type 2 diabetes. *Nature* 490: 55–60 DOI 10.5524/100036.

Rabee AE, Forster RJ, Elekwachi CO, Kewan KZ, Sabra EA, Shawket SM, Mahrous HA, Khamiss OA. 2019. Community structure and fibrolytic activities of anaerobic rumen fungi in dromedary camels. *Journal of basic microbiology* 49: 1-10 DOI 10.1002/jobm.201800323

Ransom-Jones E, Jones DL, McCarthy AJ, McDonald JE. 2012. The Fibrobacteres: an important phylum of cellulose-degrading bacteria. *Microbial ecology* 63: 267-281 DOI 10.1007/s00248-011-9998-1.

Ren Q, Si H, Yan X, Liu C, Ding L, Long R, Li Z, Qiu Q. 2020. Bacterial communities in the solid, liquid, dorsal, and ventral epithelium fractions of yak (*Bos grunniens*) rumen. MicrobiologyOpen 9: e963 https://doi.org/10.1002/mbo3.963.

Roger V, Bernalier A, Grenet E, Fonty G, Jamot J, Gouet P. 1993. Degradation of wheat straw and maize stem by a monocentric and a polycentric rumen fungus, alone or in association with rumen cellulolytic bacteria. *Animal feed science and technology* 42: 69-82 DOI 10.1016/0377-8401(93)90024-E.

Russell JB, Wilson DB. 1996. Why are ruminal cellulolytic bacteria unable to digest cellulose at low pH?. *Journal of dairy science* 79: 1503-1509 DOI 10.3168/jds.S0022-0302(96)76510-4.

Salgado-Flores A, Bockwoldt M, Hagen L, Pope P, Sundset M. 2016. First insight into the faecal microbiota of the high Arctic muskoxen (*Ovibos moschatus*). *Microbial genomics* 2 DOI 10.1099/mgen.0.000066.

Samsudin AA, Evans PN, Wright AD, Al Jassim R. 2011. Molecular diversity of the foregut bacteria community in the dromedary camel (*Camelus dromedarius*). *Environmental microbiology* 13: 3024-3035 DOI 10.1111/j.1462-2920.2011.02579.x.

Schloss PD, Westcott SL, Ryabin T, Hall JR, Hartmann M, Hollister EB, Lesniewski RA, Oakley BB, Parks DH, Robinson CJ, Sahl JW, Stres B, Thallinger GG, Van Horn DJ, Weber C F. 2009. Introducing mothur: Open-source, platform-independent, community-supported

699 software for describing and comparing microbial communities. *Applied and environmental*
700 *microbiology* 75: 7537-7541 DOI 10.1128/AEM.01541-09.

701 Selim HM, Imai S, ElSheik AK, Attia H, Okamot E. 1999. Rumen ciliate protozoal fauna of
702 native sheep, Friesian cattle and Dromedary camel in Libya. *Journal of veterinary medical*
703 *science* 61: 303-305 DOI 10.1292/jvms.61.303.

704 Shanks OC, Kelty CA, Archibeque S, Jenkins M, Newton RJ, McLellan SL, Huse SM, Sogin M
705 L. 2011. Community structures of fecal bacteria in cattle from different animal feeding
706 operations. *Applied and environmental microbiology* 77: 2992-3001 DOI 10.1128/AEM.02988-
707 10.

708 Shrivastava B, Jain KK, Kalra A, kuhad RC. 2014. Bioprocessing of wheat straw into
709 nutritionally rich and digested cattle feed. *Scientific reports* 4: 6360 DOI 10.1038/srep06360.

710 SPSS. 1999. Statistical package for social science "Release 15, SPSS INC, Chicago. USA.

711 Sridhar M, Kumar D, Anadan S. 2014. *Cyllamyces icaris sp. nov*., a new anaerobic gut fungus
712 with nodular sporangiophores isolated from Indian water buffalo (*Bubalus bubalis*).
713 *International journal of current research and academic review* 2: 7- 24.

714 Stewart CS, Duncan SH, Richardson AJ. 1992. The inhibition of fungal cellulolysis by cell-free
715 preparations from Ruminococci. *FEMS Microbiology letters* 97: 83-87 DOI 10.1111/j.1574-
716 6968.1992.tb05444.x.

717 Tapio I, Snelling TJ, Strozzi F, Wallace RJ. 2017. The ruminal microbiome associated with
718 methane emissions from ruminant livestock. *Journal of animal science and biotechnology* 8: 7
719 DOI 10.1186/s40104-017-0141-0.

720 Teunissen MJ, Dekort GM, Opdencamp HM, Huistveld HJ. 1992. Production of cellulolytic and
721 xylanolytic enzymes during growth of the anaerobic fungus *Piromyces* sp. on different
722 substrates. *Journal of general microbiology* 138: 1657-1664 DOI 10.1099/00221287-138-8-
723 1657.

724 Wang K, Nan X, Chu K, Tong J, Yang L, Zheng S, Zhao G, Jiang L, Xiong B. 2018. Shifts of
725 hydrogen metabolism from methanogenesis to propionate production in response to replacement
726 of forage fiber with non-forage fiber sources in diets *in vitro*. *Frontiers in microbiology* 9:2764
727 DOI 10.3389/fmicb.2018.02764.

728 Weimer PJ. 2015. Redundancy, resilience, and host specificity of the ruminal microbiota:
729 implications for engineering improved ruminal fermentations. *Frontiers in microbiology* 6: 296
730 DOI 10.3389/fmicb.2015.00296.

731 Williams AG, Withers SE, Naylor GE, Joblin KN. 1994. Effect of heterotrophic ruminal bacteria
732 on xylan metabolism by the anaerobic fungus *Piromyces communis*. *Letters in applied*
733 *microbiology* 19: 105-109 DOI 10.1111/j.1472-765X.1994.tb00917.x.

734 Yanagita K, Kamagata Y, Kawaharasaki M, Suzuki T, Nakamura Y, Minato H. 2000.
735 Phylogenetic analysis of methanogens in sheep rumen ecosystem and detection of
736 *Methanomicrobium mobile* by fluorescence in situ hybridization. *Journal bioscience,*
737 *biotechnology, and biochemistry* 64: 1737-1742 DOI 10.1271/bbb.64.1737.

738 Yang CL, Mi L, Hu XL, Liu JX, Wang JK. 2016. Investigation into host selection of the cecal
739 acetogen population in rabbits after weaning. *PLoS One* 11: e0158768 DOI
740 10.1371/journal.pone.0158768.

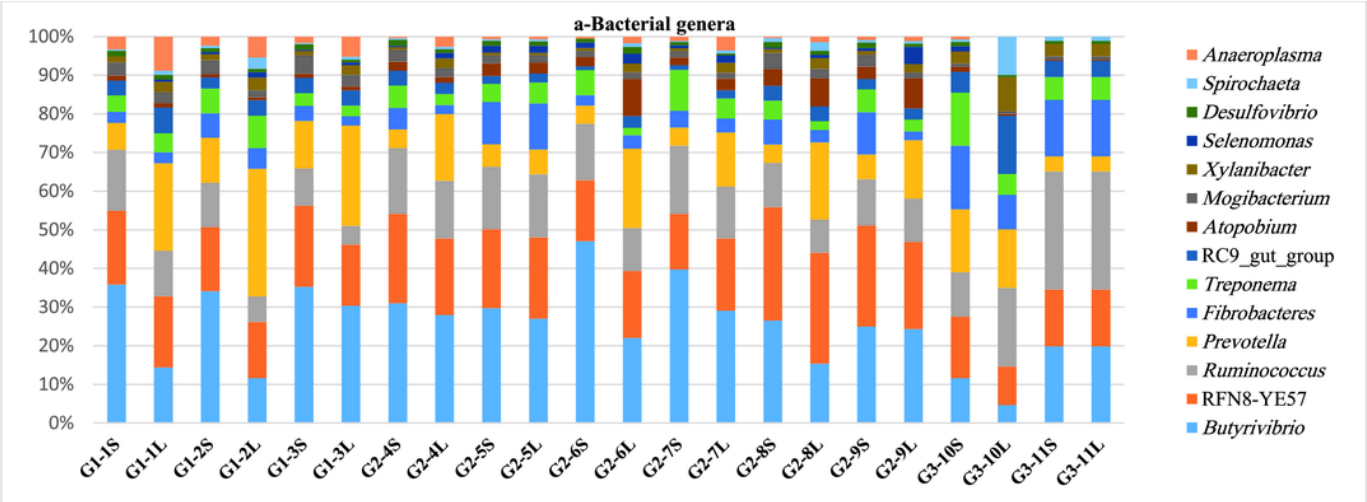
741 Zhang J, Kobert K, Flouri T, Stamatakis A. 2014. PEAR: a fast and accurate Illumina Paired-End
742 read merger. *Bioinformatics* 30: 614–620 DOI 10.1093/bioinformatics/btt593.

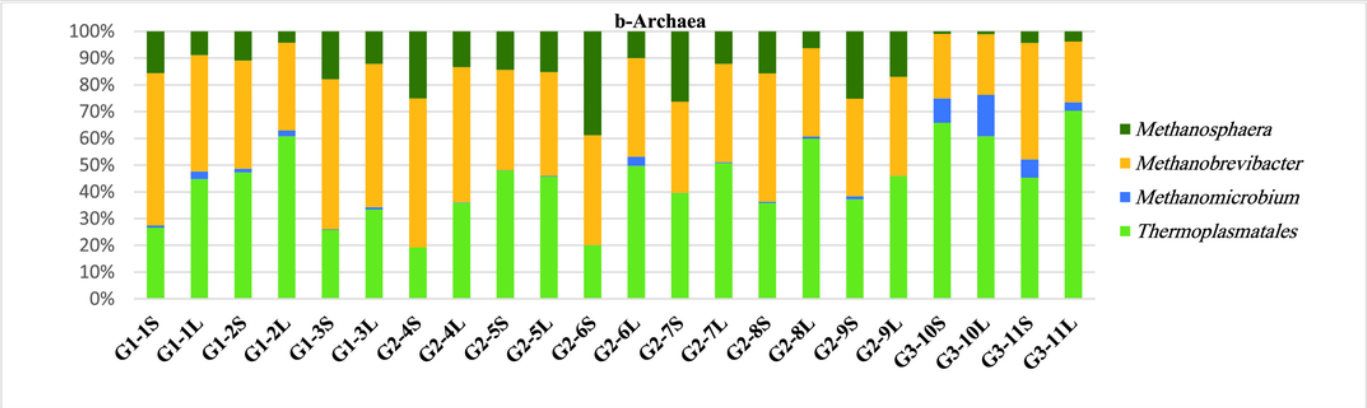
743 Zoetendal E, Plugge CM, Akkermans ADL, Vos WM. 2003. *Victivallisvadensis gen. nov*., sp.
744 nov., a sugar-fermenting anaerobe from human faeces. *International journal of systematic and*
745 *evolutionary microbiology* 53: 211-215 DOI 10.1099/ijs.0.02362-0.
746

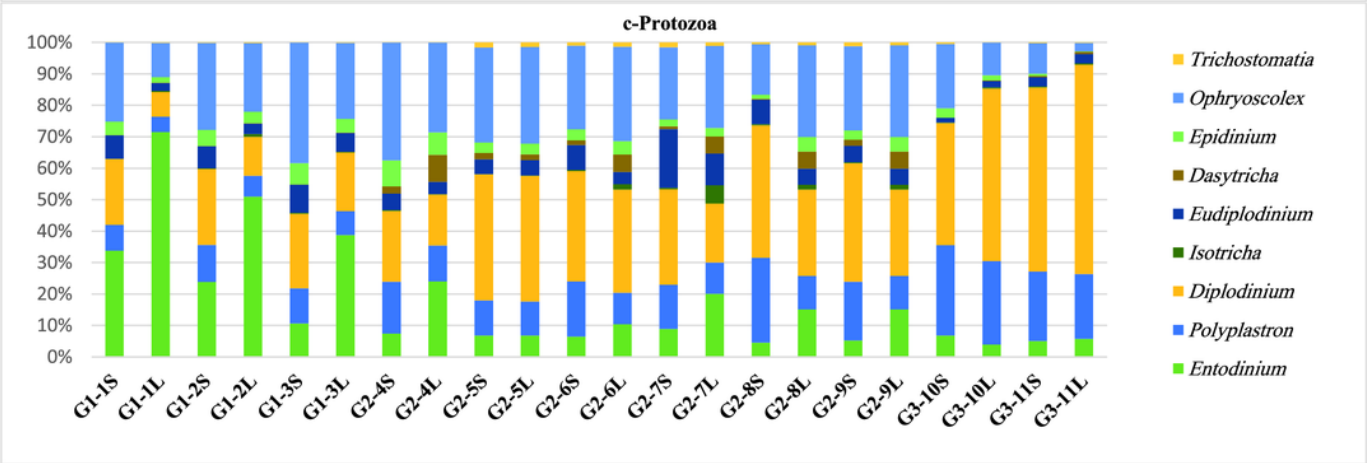
Figure 1

The relative abundance of microbial groups

Figure 1: Comparison of relative abundance of genera of the microbiota in dromedary camel. bacterial (a), archaeal (b), protozoal (c) and fungi (d) in ruminal solid (SF) and liquid (LF) fractions of camels under different feeding systems.


a-Bacterial genera
100%
90%
80%
70%
60%
50%
40%
30%
20%
10%
0%
G1-1S G1-1L G1-2S G1-2L G1-3S G1-3L G2-4S G2-4L G2-5S G2-5L G2-6S G2-6L G2-7S G2-7L G2-8S G2-8L G2-9S G2-9L G3-10S G3-10L G3-11S G3-11L
Anaeroplasma
Spirochaeta
Desulfovibrio
Selenomonas
Xylanibacter
Mogibacterium
Atopobium
RC9_gut_group
Treponema
Fibrobacteres
Prevotella
Ruminococcus
RFN8-YE57
Butyrivibrio


b-Archaea
100%
90%
80%
70%
60%
50%
40%
30%
20%
10%
0%
G1-1S G1-1L G1-2S G1-2L G1-3S G1-3L G2-4S G2-4L G2-5S G2-5L G2-6S G2-6L G2-7S G2-7L G2-8S G2-8L G2-9S G2-9L G3-10S G3-10L G3-11S G3-11L
Methanosphaera
Methanobrevibacter
Methanomicrobium
Thermoplasmatales


c-Protozoa
100%
90%
80%
70%
60%
50%
40%
30%
20%
10%
0%
G1-1S G1-1L G1-2S G1-2L G1-3S G1-3L G2-4S G2-4L G2-5S G2-5L G2-6S G2-6L G2-7S G2-7L G2-8S G2-8L G2-9S G2-9L G3-10S G3-10L G3-11S G3-11L
Trichostomatia
Ophryoscolex
Epidinium
Dasytricha
Eudiplodinium
Isotricha
Diplodinium
Polyplastron
Entodinium

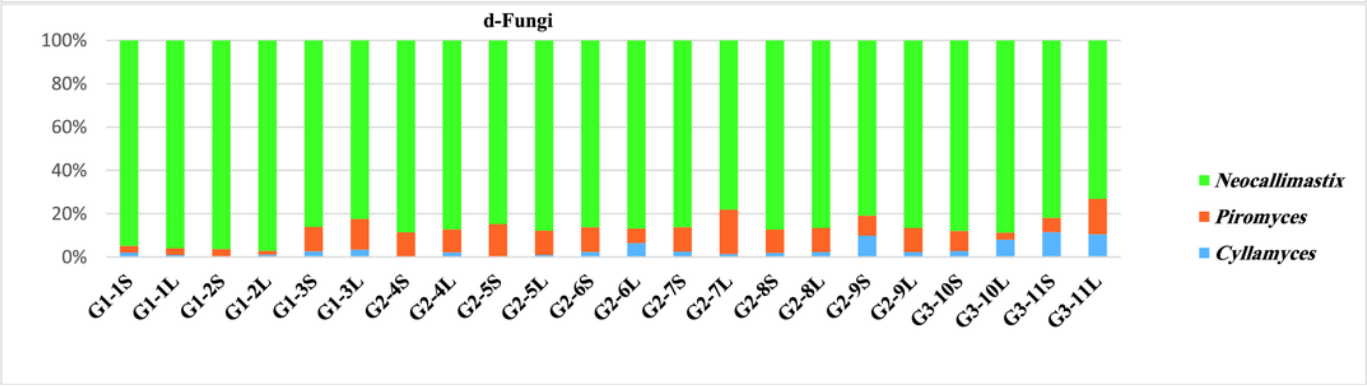

d-Fungi
100%
80%
60%
40%
20%
0%
G1-1S G1-1L G1-2S G1-2L G1-3S G1-3L G2-4S G2-4L G2-5S G2-5L G2-6S G2-6L G2-7S G2-7L G2-8S G2-8L G2-9S G2-9L G3-10S G3-10L G3-11S G3-11L
Neocallimastix
Piromyces
Cyllamyces

Figure 2

Principal Co-ordinated analysis

Figure 2: Principal Co-ordinated analysis derived from OTUs from twenty-two ruminal liquid (LF) and solid (SF) samples distributed on three camel groups. G1 camels (red circles), G2 (white circle and G3(blue circles).

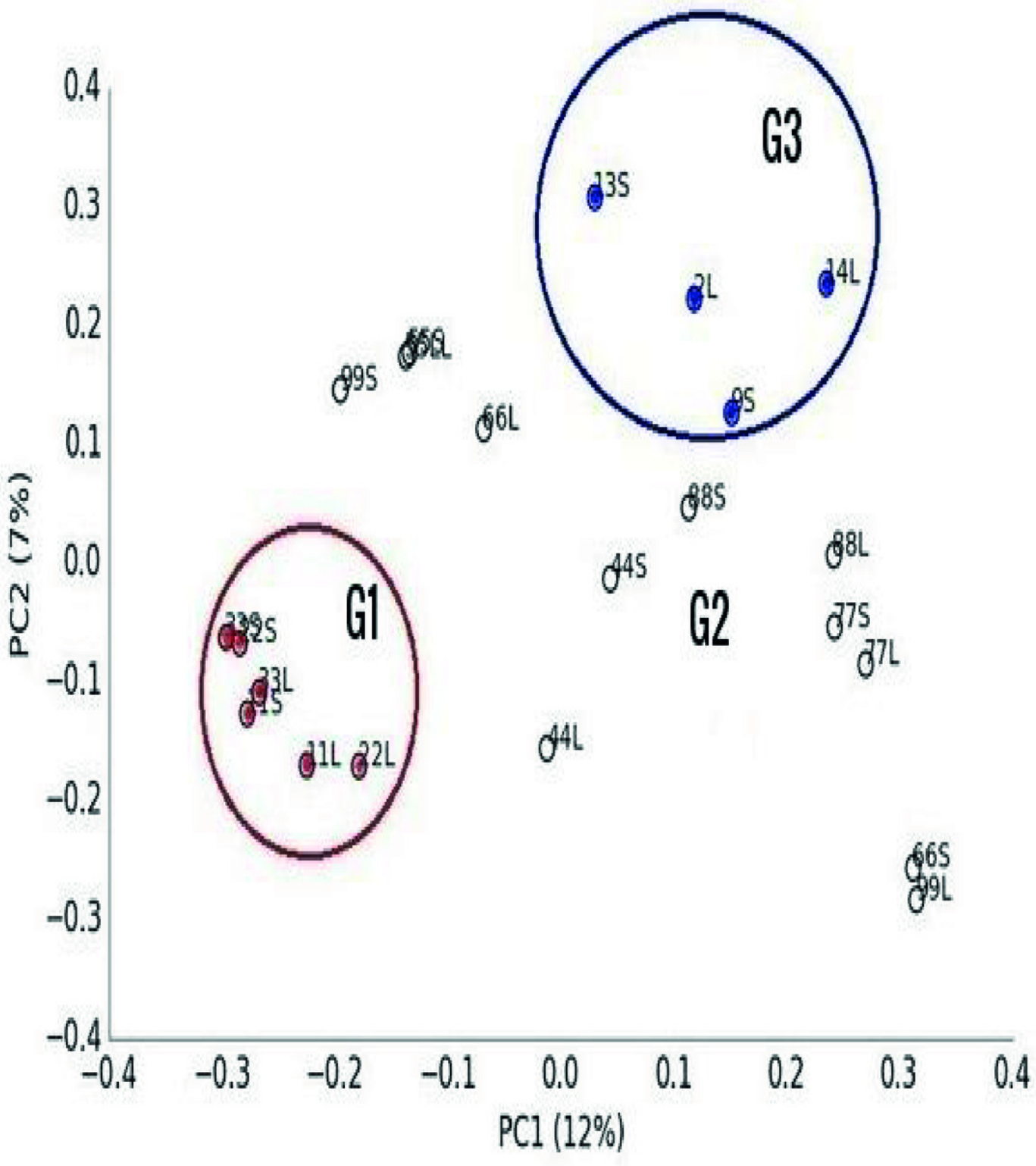
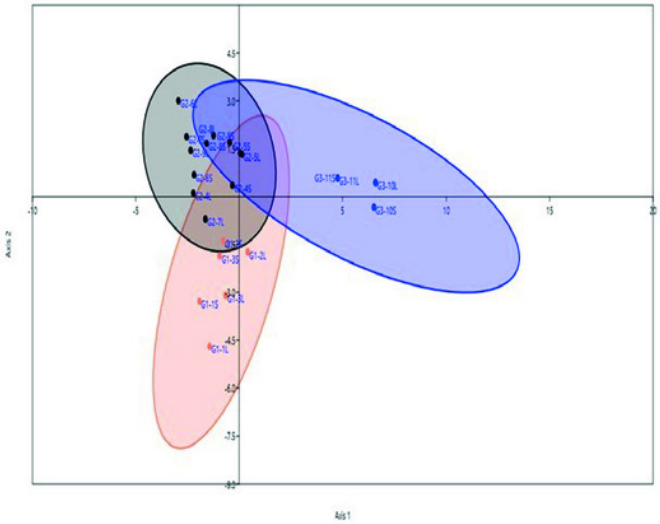

G1
G2
G3
PC1 (12%)
PC2 (7%)
0.4
0.3
0.2
0.1
0.0
−0.1
−0.2
−0.3
−0.4
−0.4
−0.3
−0.2
−0.1
0.0
0.1
0.2
0.3
0.4
13S
14L
2L
9S
6L
8S
8L
4S
7S
7L
4L
6S
9L
11L
12L
3L
1S

Figure 3

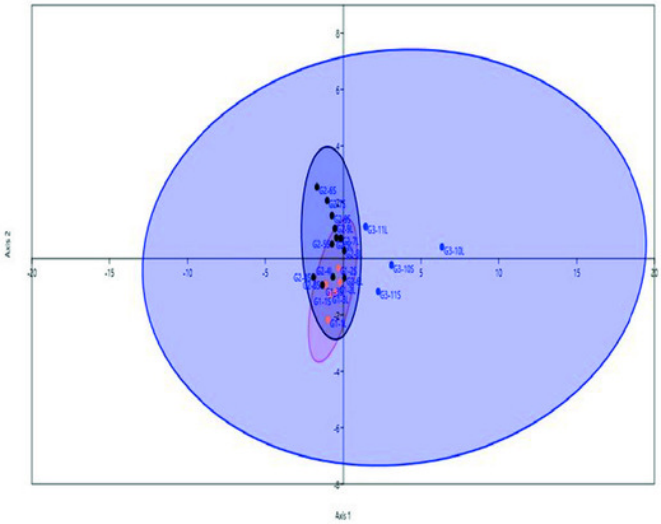
Linear Discriminant analysis

Figure 3: Linear Discriminant analysis of microbial communities in the samples based on the relative abundance of genera of active bacteria (a), archaea (b), protozoa (c) and fungi i (d) in ruminal solid (SF), and liquid (LF) fractions of camels under three feeding systems, G1 (black dots), G2 (blue squares) and G3 (coral triangles).

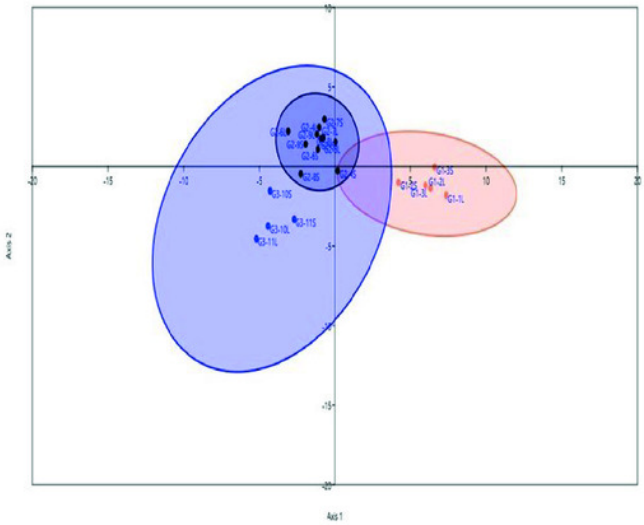
a-Bacteria



B-Archaea



c-Protozoa



d-Fungi

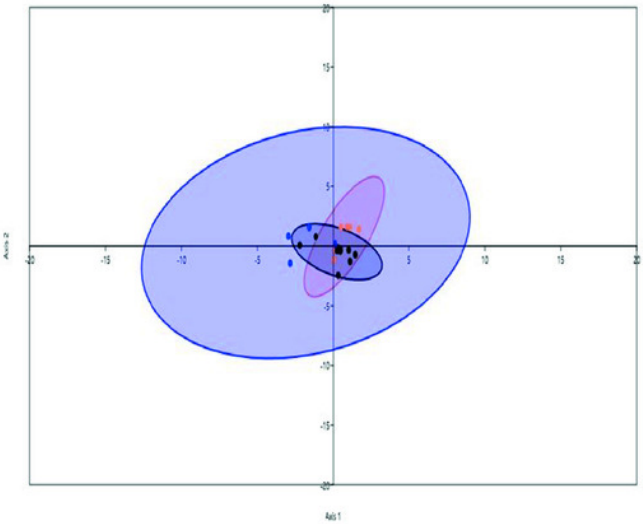
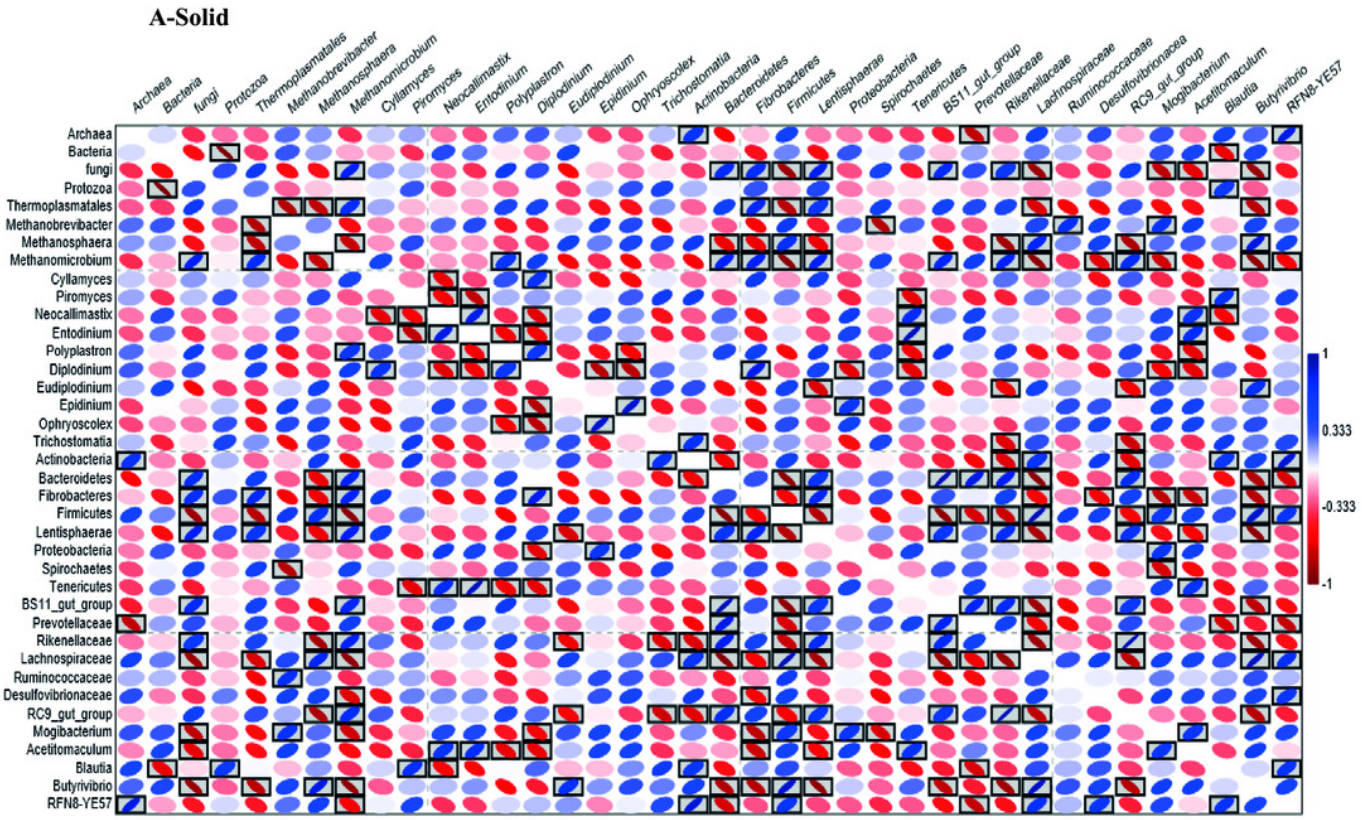


Figure 4

Heatmap based on Pearson correlation

Figure 4: Heatmap based on Pearson correlation coefficients between and within the relative abundance of bacteria, archaea, protozoa and fungi in solid (A) and liquid (B) rumen fractions of dromedary camel. The black boxed ellipses refer to the significant correlations at $P < 0.05$.


A-Solid
Archaea
Bacteria
fungi
Protozoa
Thermoplasmatales
Methanobrevibacter
Methanosphaera
Methanomicrobium
Cyllamyces
Piromyces
Neocallimastix
Entodinium
Polyplastron
Diplodinium
Eudiplodinium
Epidinium
Ophryoscolex
Trichostomatia
Actinobacteria
Bacteroidetes
Fibrobacteres
Firmicutes
Lentisphaerae
Proteobacteria
Spirochaetes
Tenericutes
BS11_gut_group
Prevotellaceae
Rikenellaceae
Lachnospiraceae
Ruminococcaceae
Desulfovibrionacea
RC9_gut_group
Mogibacterium
Acetitomaculum
Blautia
Butyrivibrio
RFN8-YE57
1
0.333
-0.333
-1

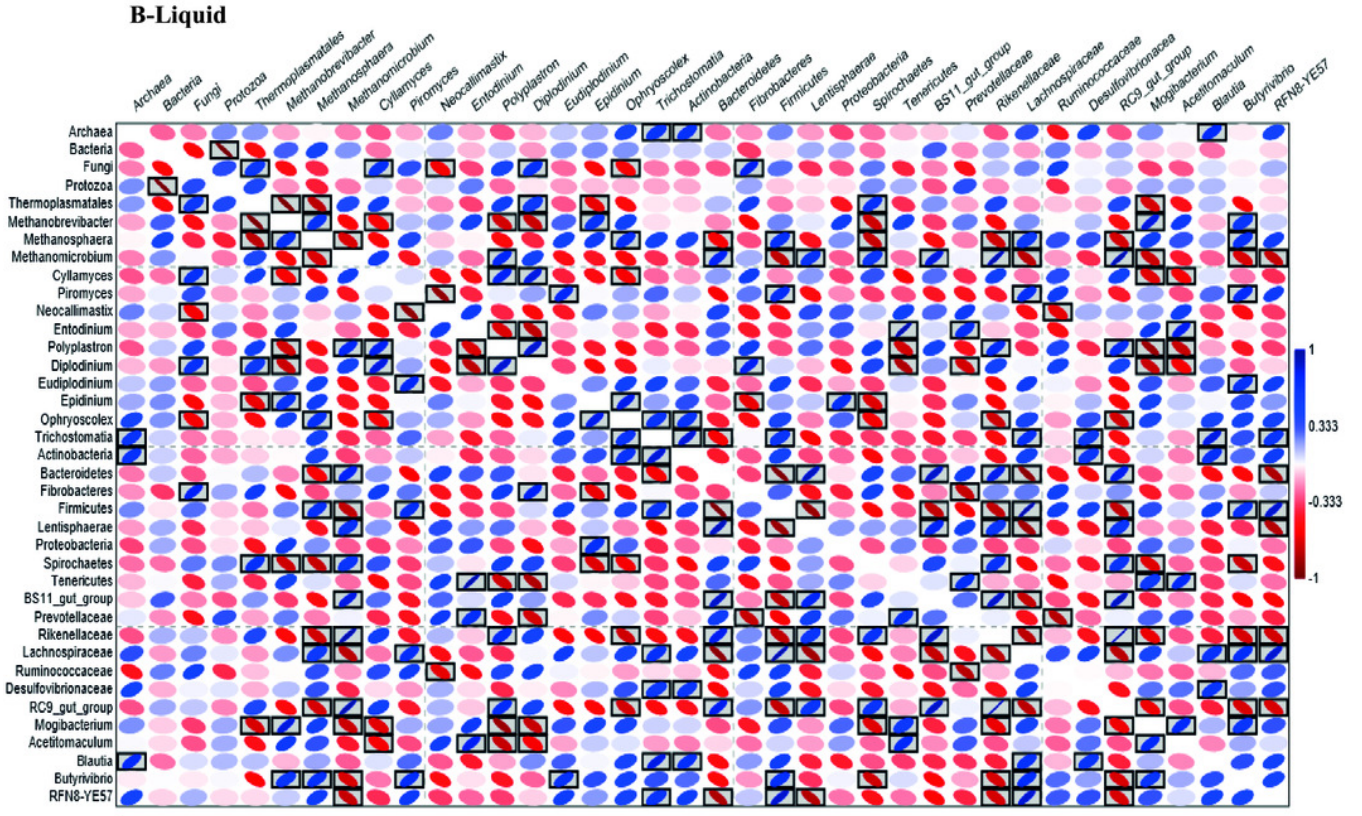

B-Liquid
Archaea
Bacteria
Fungi
Protozoa
Thermoplasmatales
Methanobrevibacter
Methanosphaera
Methanomicrobium
Cyllamyces
Piromyces
Neocallimastix
Entodinium
Polyplastron
Diplodinium
Eudiplodinium
Epidinium
Ophryoscolex
Trichostomatia
Actinobacteria
Bacteroidetes
Fibrobacteres
Firmicutes
Lentisphaerae
Proteobacteria
Spirochaetes
Tenericutes
BS11_gut_group
Prevotellaceae
Rikenellaceae
Lachnospiraceae
Ruminococcaceae
Desulfovibrionacea
RC9_gut_group
Mogibacterium
Acetitomaculum
Blautia
Butyrivibrio
RFN8-YE57
1
0.333
-0.333
-1

Table 1(on next page)

The Relative abundance (%) of bacteria, archaea, protozoa and fungi and diversity indices

Table 1: The Relative abundance (%) of bacteria, archaea, protozoa and fungi and OTU numbers and values of Shannon, Chao1 and Inverse Simpson indices in the ruminal solid (SF) and liquid (LF) fractions of dromedary camels fed a mixed ration (G1), high-quality forage(G2) and low-quality- forage (G3) (Mean ± Standard error (SE)).

1

Item	G1	G2	G3	Overall mean
Bacteria SF	92±1	88.5±2	89±2	90±1
Bacteria LF	85± 4	90.5± 2	87± 8	88± 2
Archaea SF	2.3±0.17	3.4±0.4	2.2±1	3±0.3
Archaea LF	2.2± 0.2	2.8± 0.4	1.75± 0.2	2± 0.25
Protozoa SF	5±1	7±2	6±2	6±1
Protozoa LF	12± 4	6± 1.6	8± 5	8± 1.6
Fungi SF	0.13± 0.05	0.9± 0.3	3± 1.1	1± 0.4
Fungi LF	0.35± 0.1	0.5± 0.1	3± 3	0.95± 0.5
OTUs SF	1012± 43	1201± 38	1135± 148	1137± 39
OTUs LF	1076± 26	1229± 38	1147± 53	1172± 30
Shannon SF	6± 0.1	7± 0.10	7± 0.3	7± 0.1
Shannon LF	6.5± 0.06	7± 0.09	7± 0.1	7± 0.1
Chao1 SF	6644± 650	9329±714	9028± 1985	8542± 608
Chao1 LF	7280± 521	10839± 724	7688± 625	9295± 672
Invsimpsone SF	117± 14	863± 306	644± 398	620± 196
Invsimpsone LF	135± 21	983± 492	612± 142	684± 282

2
3

Table 2(on next page)

Relative abundance (%) of bacterial phyla

Table 2: Relative abundance (%) of bacterial phyla in the ruminal solid (SF) and liquid (LF) fractions of camels fed a mixed ration(G1), high-quality forage(G2) and low-quality forage (G3) (Mean ± Standard Error (SE)).

1

Bacterial Phylum	G1	G2	G3	Overall mean
Firmicutes SF	63±1.5	65±0.1	48±9.5	60±2.5
Firmicutes LF	46±3.1	56±2	45±13	50±2.7
Bacteroidetes SF	20±1	15.5±1	27±8	19±2
Bacteroidetes LF	31±0.5	21.5±1.5	31.5±12	26±2.5
Proteobacteria SF	5±1	3.5±0.25	3±0.5	4±0.3
Proteobacteria LF	6.5±1	6±2	3±0.1	5.5±1
Spirochaetes SF	3±0.6	5±1	6±1.5	4.5±0.6
Spirochaetes LF	3.7±1	2.6±0.5	5.6±0.8	3.5±0.5
Fibrobacteres SF	2.5±0.6	4±0.7	9±1	4.5±1
Fibrobacteres LF	1.6±0.5	2.5±1	7±3	3±1
Actinobacteria SF	2±0.2	4.5±0.3	1.5±0.3	3±0.5
Actinobacteria LF	1.5±0.14	5.5±1	1±0.1	3.6±0.8
Lentisphaerae SF	0.7±0.03	0.7±0.1	1.5±0.2	0.8±0.1
Lentisphaerae LF	3.2±0.3	2±0.5	3.2±2	2.6±0.4
Tenericutes SF	2±0.4	0.8±0.1	0.6±0.25	1±0.2
Tenericutes LF	3.7±0.6	1.5±0.25	0.4±0.1	1.8±0.4
Verrucomicrobia SF	0.26±0.1	0.22±0.1	0.57±0.4	0.33±0.1
Verrucomicrobia LF	2.2±0.4	1±0.3	1.3±0.3	1.3±0.3
Chloroflexi SF	0.4±0.03	0.5±0.06	0.24[a]	0.4±0.04
Chloroflexi LF	0.3±0.03	0.3±0.05	0.24[a]	0.3±0.02
Cyanobacteria SF	0.3±0.04	0.3±0.05	0.5[a]	0.35±0.04
Cyanobacteria LF	0.3±0.05	0.3±0.05	0.25[a]	0.3±0.03
Elusimicrobia SF	0.2±0.05	0.15	0.3±0.14	0.2±0.04
Elusimicrobia LF	0.3±0.07	0.2±0.04	0.8±0.4	0.4±0.1

2 [a] The value was calculated from one animal.

3

Table 3(on next page)

Relative abundance (%) of archaeal orders and genera

Table 3: Relative abundance (%) of archaeal orders and genera observed in the ruminal solid (SF), and liquid (LF) fractions of camels under different feeding systems. Animals in G1 fed a mixed ration, animal in G2 fed high-quality forage and animal in G3 fed low quality-forage (Mean ± Standard Error (SE)).

Archaea	G1	G2	G3	Overall mean
Thermoplasmatales SF	33 ± 7	33 ± 4	55 ± 10	37 ± 4
Thermoplasmatales LF	46 ± 8	48 ± 3	67 ± 5	51 ± 3
Methanomicrobium SF	0.8 ± 0.3	0.3± 0.2	8 ± 1	2 ± 0.9
Methanomicrobium LF	2 ± 0.5	0.9 ± 0.5	9 ± 6	3 ± 1
Methanobrevibacter SF	51± 5	42± 3	34± 9	43± 3
Methanobrevibacter LF	43± 5	39± 2.4	23± 0.01	37± 2
Methanosphaera SF	15± 2	24 ± 3	3± 1	18± 3
Methanosphaera LF	8± 2	12± 1.5	2.5± 1	9.5± 1.5
Methanobacterium SF	0.05	0.06	0	ND
Methanobacterium LF	0.2 ± 0.02	0.07± 0.02	0	ND

ND: Non Determined

Table 4(on next page)

Relative abundance (%) of protozoal genera

Table 4: Relative abundance (%) of protozoal genera in the ruminal solid (SF) and liquid fraction (LF) of camels under different feeding systems. Animals in G1 fed a mixed ration, animals in G2 fed high-quality forage and animals in G3 fed low-quality forage (Mean ± SE).

1

Protozoa	G1	G2	G3	Overall mean
Entodinium SF	23±6	6.5±0.6	6±0.8	11±3
Entodinium LF	54±9.5	15±2.5	5±0.8	24±6
Polyplastron S F	10±1	17.5±2	25±3	17±2
Polyplastron LF	6±0.8	11±0.2	24±3	12±2
Diplodinium S F	23±1	35±3	49±10	34±3
Diplodinium LF	13±3	27±3	61±6	29±5
Eudiplodinium SF	8±0.6	8±2	2±0.7	7±1
Eudiplodinium LF	4±1	5.5±0.9	2.5±0.4	4.5±0.6
Epidinium SF	5±0.76	4±1	2±1	4±0.1
Epidinium LF	3±0.8	4.5±0.6	1±0.7	3.5±0.5
Ophryoscolex SF	30±4	27±3	15±5	26±2.5
Ophryoscolex LF	19±4	29±0.6	6.5±3.8	22±3
Trichostomatia SF	0.1±0.02	1±0.25	0.3±0.15	1±0.2
Trichostomatia LF	0.2±0.04	1±0.2	1±0.1	1±0.2
Isotricha SF	0.2±0.04	0.3±0.05	0.3±0.004	0.3±0.03
Isotricha LF	0.5±0.2	2±0.85	0.3±0.007	1±0.5
Dasytricha SF	0.04±0.008	1.5±0.3	0.2±0.15	1±0.3
Dasytricha LF	0.1±0.002	5.5±0.8	0.5±0.3	3±1

2

Table 5(on next page)

Relative abundance (%) of fungal genera

Table 5: Relative abundance (%) of fungal genera in the ruminal solid (SF) and liquid fraction (LF) of camels under different feeding systems. Camels in G1 fed a mixed ration, animals in G2 fed high- quality forage, and animals in G3 fed low- quality forage (Mean ± SE).

1

Fungi	G1	G2	G3	Overall mean
Spizellomyces SF	0	0.09	0.017	ND
Spizellomyces LF	0.3 ± 0.1	0.25 ± 0.1	0	ND
Cyllamyces SF	2± 0.6	3± 1.5	7± 4	3.5± 1
Cyllamyces LF	2± 0.78	3± 0.8	10± 1	4± 1
Piromyces SF	6± 3	12± 0.7	8± 1	9± 1
Piromyces LF	6±4	12±2	10±6	10±2
Neocallimastix SF	92±3	85±1	85±3	87±1
Neocallimastix LF	92±4	85±1.5	81±7	86±2

2 ND: Non Determined

3