

ORIGINAL ARTICLE

Specific microbiome-dependent mechanisms underlie the energy harvest efficiency of ruminants

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Ruminants have the remarkable ability to convert human-indigestible plant biomass into human-digestible food products, due to a complex microbiome residing in the rumen compartment of their upper digestive tract. Here we report the discovery that rumen microbiome components are tightly linked to cows' ability to extract energy from their feed, termed feed efficiency. Feed efficiency was measured in 146 milking cows and analyses of the taxonomic composition, gene content, microbial activity and metabolic composition was performed on the rumen microbiomes from the 78 most extreme animals. Lower richness of microbiome gene content and taxa was tightly linked to higher feed efficiency. Microbiome genes and species accurately predicted the animals' feed efficiency phenotype. Specific enrichment of microbes and metabolic pathways in each of these microbiome groups resulted in better energy and carbon channeling to the animal, while lowering methane emissions to the atmosphere. This ecological and mechanistic understanding of the rumen microbiome could lead to an increase in available food resources and environmentally friendly livestock agriculture.

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Introduction

Ruminants hold enormous significance for man, as they convert the energy stored in plant biomass polymers, which are indigestible for humans, to digestible food products. Humans domesticated these animals for this purpose in the Neolithic era (Ajmone-Marsan *et al.*, 2010) and have been farming them ever since for the production and consumption of animal protein in the form of meat and milk. In today's extensive production regimes, ruminants consume 30% of the crops grown on earth and occupy another 30% of the earth's land mass (Thornton, 2010). These animals also emit methane—a highly potent greenhouse gas—to the atmosphere and are considered to be responsible for a considerable portion of its emission because of anthropogenic activities (McMichael *et al.*, 2007). One way to tackle these problems is to increase the animals' energetic

efficiency, that is, the efficiency with which they convert energy from feed, thereby increasing food availability, while lowering the environmental burden, as these animals would produce more and eat less (Bradford, 1999; Thornton, 2010).

Different methods are used to evaluate an animal's energetic efficiency; of these, the residual feed intake (RFI) method (Koch *et al.*, 1963) is highly accepted and widely used (Herd and Arthur, 2009) as it takes into account growth and body size and is thus suitable for comparisons between animals. This parameter is an estimation of the difference between an animal's actual feed intake and its predicted feed intake based on its production level and body weight. The energetic efficiency varies considerably between different individuals from the same breed. Specific genomic regions, such as one that is suggested to be associated with a role in controlling energy metabolism, have been found to correlate to feed efficiency using genome-wide association studies (Pryce *et al.*, 2012). Nevertheless, only a moderate genetic component (heritability ranging from 0.26 to 0.58) affects energy utilization, as has also been demonstrated by elevation of feed efficiency via selection of animals according to their RFI (Archer *et al.*, 1999; Moore *et al.*, 2009).

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One important factor that could greatly contribute to variations in these animals' feed efficiency is the rumen microbiome. The ability of these animals to digest plant biomass polymers is attributed to this complex microbiome that resides in their upper digestive tract in a compartment termed the rumen (Mizrahi, 2013). The anaerobic environment in the rumen and the highly complex food webs sustained by the rumen microbiome enable the fermentation of plant material into metabolic end products such as short-chain fatty acids (SCFAs) and methane. Although SCFAs are absorbed through the rumen wall and serve to fulfill the animal's energy needs, methane is not absorbed; it is emitted to the atmosphere together with its retained energy, thereby contributing to energy loss from the feed, as well as global warming (Mizrahi, 2011). Microbial composition of the rumen has been described using various techniques (Brulc *et al.*, 2009; Hess *et al.*, 2011; Jami and Mizrahi, 2012; Henderson *et al.*, 2015). In addition, differences between high and low RFI animals have been reported in terms of methane production, as well as of some differences in microbial composition (Nkrumah *et al.*, 2006; Mizrahi, 2011; Hernandez-Sanabria *et al.*, 2012; Jami *et al.*, 2014; Kittelmann *et al.*, 2014; Shi *et al.*, 2014; Wallace *et al.*, 2015). Nevertheless, a comprehensive and thorough understanding of microbiome structure patterns and how to translate them to functionality at the animal level is still lacking.

Here we determined the feed efficiency phenotype of a cohort of 146 animals; this phenotype was mapped to its underlying microbiome determinants by sampling and analyzing rumen fluid and feces from the animals in the two extreme quartiles (78 animals—40 efficient and 38 inefficient; Supplementary Figure S1 and Supplementary Data 1). We characterized the taxonomic composition, genetic functional potential, metabolomic composition and activity of these rumen microbiomes to explore the hypothesis of a link between the animal's feed efficiency and these microbiome components, and to uncover the potential mechanisms that might explain this link. Our data provide detailed and novel insight into the characteristics and components of the rumen microbiome related to feed efficiency—their ecological context, the underlying mechanisms and their potential as markers to predict the feed efficiency phenotype.

Materials and methods

Trial design

The experimental procedures used in this study were approved by the Faculty Animal Policy and Welfare Committee of the Agricultural Research Organization (ARO), approval number IL-386/12, Volcani Research Center, and were in accordance with the guidelines of the National Council for Animal Experimentation.

A total of 146 Holstein Friesian dairy cows were selected for the experiment and housed at the ARO's

experimental dairy farm in Bet Dagan, Israel. Cows with history of diseases, miscarriages and twin pregnancies or that were above first trimester were not included in the experiment. The experimental dairy farm is equipped with a facility that is specially designed to individually monitor all of the animal's functions, feed intake and different physiological parameters (Halachmi *et al.*, 1998). The animals were divided into seven groups according to lactation period such that each cow was between 50 and 150 days of lactation when monitored. Each group contained between 19 and 21 cows that were monitored for 42–49 days. The animals were fed *ad libitum* a standard lactating cow diet consisting of 30% roughage and 70% concentrate and had free access to water. The cows were habituated with the aforementioned diet for 3 weeks before the start of the experiment so that they would become accustomed to their individual feeding station.

The following parameters were automatically monitored three times a day during the experiment: dry matter (DM) intake (kg), weight (kg), milk yield (kg), milk lactose, fat and protein (g) and somatic cell count using the Afimilk program (Afimilk Ltd, Kibbutz Afikim, Israel). Milk samples were sent to an authorized milk quality lab (National Service for Udder Health and Milk Quality, Caesarea, Israel) three times for each group to verify the Afimilk program analysis. Body conditioning score was measured once a week by the same person throughout the experiment.

Feed efficiency parameters RFI and conversion ratio were calculated according to National Research Council (2001) formulas. In order to increase the statistical power compared with random sampling, extreme phenotypes sampling approach (Li *et al.*, 2011) was applied. Twelve cows with the most extreme and stable RFI values were selected from each group for rumen fluid sampling, six with low and six with high RFI values. Tukey's test was used to verify that the RFI value of each cow was steady throughout the experiment and significantly different from cows in the reciprocal efficiency group. Overall, 78 cows were chosen for sampling and represented the 25% most efficient and 25% most inefficient animals of the whole cohort ($P < 0.0001$; Supplementary Figure S1 and Supplementary Data 1). Our previous work showed that samples from 16 cows are sufficient in order to cover all of the microbial diversity in the bovine rumen on a specific diet (Jami and Mizrahi, 2012). Therefore, this extreme phenotype characterization and sampling approach, together with the large cohort from which the extreme cows were chosen from, ensured increased adequate power to detect microbiome components connected to the host phenotype.

Sample collection

Rumen samples were collected on 3 consecutive days. The cows were sampled 6 h after feeding in which they were not offered feed; 500 ml of rumen contents were collected using a stainless-steel

stomach tube with a rumen vacuum sampler, and pH was immediately determined. Samples for DNA and metabolite extraction were snap frozen in liquid nitrogen and stored at -80°C until analysis. Rumen samples for metabolic assays were filtered through six layers of cheese cloth to remove big feed particles, transferred to CO_2 -containing bottles and flushed with CO_2 to maintain anaerobic conditions. Immediately after collection, the rumen samples were maintained at 39°C up to 1 h until use, and processed in the laboratory, located 100 m away.

Fresh fecal samples were obtained three times a day for 4 consecutive days. Samples were immediately frozen at -20°C .

In-vitro digestibility assay

The *in-vitro* digestibility of plant cell wall fibers, represented by neutral detergent fiber or total feed polymers (in DM), was determined according to the two-stage technique by Tilley and Terry (1963). Briefly, cows' feed was dried for 72 h in an aerated 60°C oven and then ground to pass a 1-mm screen. The feed was incubated with rumen fluid and artificial rumen buffer, in sealed glass tubes.

Artificial rumen buffer was formulated as described previously (McDougall, 1948; Tilley and Terry, 1963). Briefly, 100 ml of buffer A (98 g NaHCO_3 , 93 g $\text{NaH}_2\text{PO}_4 \cdot 12\text{H}_2\text{O}$, 5.7 g KCl, 4.7 g NaCl, 1.2 g $\text{MgSO}_4 \cdot 7\text{H}_2\text{O}$, added to double-distilled water (DDW) to final volume of 1 liter) was added to 800 ml DDW. The solution was flushed with CO_2 to reduce the pH to 6.8–7.0. Then, 1 ml of buffer B (40 g CaCl_2 added to DDW to final volume of 1 liter), 50 ml of buffer C (30 g NH_4HCO_3 added to DDW to final volume of 1 liter) and 100 μl of buffer D (10 g $\text{MnCl}_2 \cdot 4\text{H}_2\text{O}$, 1 g $\text{CoCl}_2 \cdot 6\text{H}_2\text{O}$, 8 g $\text{FeCl}_3 \cdot 6\text{H}_2\text{O}$ added to DDW to final volume of 1 liter) were added and the buffer was brought to a final volume of 1 liter with DDW.

The tubes were flushed with CO_2 and closed with a unidirectional valve cap, which only allowed emission of gas from the tube. The tubes were incubated for 24 or 48 h at 39°C and were shaken five times a day, followed by incubation with acid pepsin. At the end of this procedure, the undigested solids were precipitated by centrifugation at 1000 g for 10 min and dried in an aerated oven at 60°C for 72 h. The precipitates were used for residual DM determination by weighing or for residual neutral detergent fiber determination by following the procedure of Van Soest *et al.* (1991). The results are expressed as mean feed digestibility in the rumen from two consecutive sampling days.

In-vivo digestibility

Fecal grab samples were pooled for each cow, dried at 60°C for 72 h in a forced-air oven and ground to pass a 1-mm screen. The indigestible neutral detergent fiber content was determined in the ration and in the fecal samples according to a previously reported

method (Lippke *et al.*, 1986) after incubation with rumen fluid for 72 h and was used as an internal marker for the apparent total-tract DM digestibility analysis. Each cow's *in-vivo* DM and neutral detergent fiber digestibility of the ration was calculated using its average DM intake and fecal output.

In-vitro methane emission assay

Samples were diluted 1:2 (v/v) with artificial rumen buffer. Duplicates of 5 ml aliquots from each diluted sample were transferred to screw-cap glass tubes (ISI, Israel Scientific Instruments Ltd, Petah-Tikva, Israel) suitable for methane measurement using a gas chromatography (GC) system HP-5890 series II, (Hewlett-Packard, Palo Alto, CA, USA) with a FID detector. The samples were incubated at 39°C for 24 h with 0.5 g DM feed, and then analyzed by GC for methane emission. Samples of 0.5 ml gas from the tube headspace were injected into a $182.88\text{ cm} \times 0.3175\text{ cm} \times 2.1\text{ mm}$ packed Supelco analytical-45/60 Molecular sieve 5 A column (Supelco Inc., Bellefonte, PA, USA) with helium carrier gas set to a flow rate of 10 ml min^{-1} and an oven temperature of 200°C . The oven temperature remained steady for a total run time of 5 min. A standard curve was generated using pure methane gas.

Methane production was quantified for 36 rumen microbiome samples of the most extreme animals of the feed efficiency groups (18 efficient and 18 inefficient), with two biological repeats of each animal.

Identification and quantification of rumen fluid metabolites

Frozen rumen fluid samples were thawed at 25°C and centrifuged at 10 000 g for 15 min. The supernatant was filtered through a sterile $0.45\text{ }\mu\text{m}$ filter (Merck Millipore Ltd., Tullagreen, County Cork, Ireland). Rumen fluid samples were kept on ice during metabolite extraction in the gas chromatography mass spectrometry and GC metabolite identification and quantification pipelines to minimize metabolite degradation.

The rumen samples were analyzed by gas chromatography mass spectrometry for polar metabolites and by GC with a FID detector for SCFAs. The extraction and derivatization protocol for the gas chromatography mass spectrometry analysis was adapted from a previously described method (Saleem *et al.*, 2013). Derivatized extracts were analyzed using an Agilent 5975C GC and an Agilent 7890A MS (Agilent Technologies, Palo Alto, CA, USA) operating in electron impact (EI) ionization mode. Aliquots (1 μl) were injected (splitless) into a $30\text{ m} \times 0.25\text{ mm} \times 0.25\text{ }\mu\text{m}$ HP-5MS Ultra Inert column (Agilent Technologies, Berkshire, UK) with helium carrier gas set to a flow rate of 1 ml min^{-1} and initial oven temperature of 70°C . The oven temperature was held constant at the initial temperature for 2 min, and thereafter increased at $10^{\circ}\text{C min}^{-1}$ to a final

temperature of 310 °C, and a final run time of 45 min. Samples were run using full scan in a mass range of 50–500 m/z (1.7 scan s⁻¹) with a detection delay of 4 min. Retention indices were calculated using a C8-C20 alkane standard mixture solution (Sigma-Aldrich, Buchs, Switzerland) as the external standard. Quantification and identification of trimethylsilylated metabolites were performed using the NIST database and high performance liquid chromatography grade standards.

For SCFA identification and quantification, 400 µl of filtered rumen fluid were mixed with 100 µl of 25% metaphosphoric acid solution (w/v in DDW) and vortexed for 1 min. The samples were incubated at 4 °C for 30 min and subsequently centrifuged for 15 min at 10 600 g. The supernatant was decanted into new tubes, then 250 µl methyl tert-butyl ether (Sigma-Aldrich) was added and the tubes were vortexed for 30 s. Another cycle of centrifugation was performed for 1 min at 10 600 g. The upper phase, which contained methyl tert-butyl ether +SCFAs, was analyzed using an Agilent 7890B GC system (Agilent Technologies, Santa Clara, CA, USA) with a FID detector. The temperatures at the inlet and detector were 250 °C and 300 °C, respectively. Aliquots (1 µl) were injected with a split ratio of 1 : 25 into a 30 m × 0.32 mm × 0.25 µm ZEBRON ZB-FFAP column (Phenomenex, Torrance, CA, USA) with helium carrier gas set to a flow rate of 2.4 ml min⁻¹ and initial oven temperature of 100 °C. The oven temperature was held constant at the initial temperature for 5 min, and thereafter increased at 10 °C min⁻¹ to a final temperature 125 °C, and a final run time of 12.5 min.

Quantification and identification of metabolites were performed using high performance liquid chromatography-grade standards. All metabolites were normalized to the organic matter content of the rumen fluid they were extracted from. Rumen samples were filtered through a sterile 0.45 µm Supor Membrane filter (PALL Life Sciences Ann Arbor, MI, USA). The organic C in the rumen samples was analyzed with a Formacs, combustion total organic carbon analyzer (Skalar, De Breda, The Netherlands).

Microbial DNA extraction

The rumen microbial fraction was separated according to Stevenson and Weimer (2007), with minor modifications to suit the needs of these experiments as described in Jami *et al.* (2013). The DNA extraction was performed as described by Stevenson and Weimer (2007).

Shotgun DNA sequencing and analysis

Metagenomic DNA libraries were constructed with the TruSeq DNA Sample Prep kit (Illumina, San Diego, CA, USA). Libraries were pooled and sequenced on two lanes for 151 cycles

from each end on a HiSeq2500 (Illumina) and processed with Casava 1.8.2 (Illumina). On average, 35 581 041 ± 6 899 269 paired end reads were obtained from each sample and 2 775 321 186 paired end reads were obtained overall. In all, 18.6% of the reads did not pass artifact filtering and trimming using MOCAT pipeline (Kultima *et al.*, 2012).

To obtain a more comprehensive metagenome, a joint assembly of all data from the 78 cows was created. This compensated for the lower sequencing depth of each individual sample and any bias caused by assembly of individual samples. Reads from all samples were pooled and assembled into one metagenome using CLC Bio, package CLC Assembly Cell version 3.2.2 (Qiagen, Redwood, CA, USA) with K-mer = 21 and default parameters; 16 784 830 contigs were obtained. A QC pipeline of dereplication and screening for *Bos taurus* reads was performed using the MG-RAST pipeline. No redundancies were found and 0.43% of the contigs were discarded after removing *Bos taurus* contaminants. The phylogenetic origin of each contig was annotated with RefSeq database (Pruitt *et al.*, 2007) ($E \leq 10^{-5}$) using the MG-RAST pipeline (Meyer *et al.*, 2008).

Gene calling was performed on the contigs using FragGeneScan (Rho *et al.*, 2010); 21 531 511 genes were identified over all. Each sample's reads were recruited against the overall genes using burrows-wheeler alignment tool (Li and Durbin, 2009) with 98% identity and default parameters; a threshold of one read for gene identification was chosen to include rare genes in the analysis. On average, 52.4% of the reads from each sample were mapped to the obtained genes, without differences between the efficiency groups (Supplementary Figure S2). An average of 4 079 212 genes were identified in each sample. The abundance of a specific gene was calculated by the number of reads uniquely recruited, normalized to the length of the gene and total reads obtained from the sample. The number of genes detected had no dependence on the number of mapped reads (Supplementary Figure S3).

16S ribosomal DNA sequencing and analysis

The 16S V3 region was amplified using the primers 357F 5'-CCTACGGGAGGCAGCAG-3' and 926R 5'-CCGTC AATTCMTTTRAGT-3' (Peterson *et al.*, 2009). The libraries were pooled and sequenced on one MiSeq flowcell (Illumina) for 251 cycles from each end of the fragments and analyzed with Casava 1.8. Overall, 49 760 478 paired end reads were obtained for all 3 sampling days, with an average of 212 652 reads per sample per day.

Data quality control and analyses were performed using the QIIME pipeline version 1.7.0 (Caporaso *et al.*, 2010). Species were defined at 97% identity using UCLUST (Edgar, 2010). Taxonomy assignment of species was performed using BLAST against the 16S rRNA reference database RDP (version 10) (Cole *et al.*,

2003). All singletons and doubletons were removed from the data set, resulting in 85 225 species with an average of 5039 per sample. Species were binned at different taxonomic levels to receive taxon abundances for each phylogenetic level (Supplementary Figure S4 and Supplementary Data 2).

Biodiversity analysis

Within-sample (alpha) diversity was calculated using Shannon index and dominance was determined according to 1–Simpson index (Harper, 1999). The indices of 16S rRNA gene profiles were calculated using bootstrapping with 9999 replicates. Richness of genes and taxa are presented as simple counts of genes and taxa.

Identifying differential species and genes

The statistical significance of differences in species and gene abundance between the efficiency groups was tested by Wilcoxon rank-sum test coupled with a bootstrapping approach adopted from Le Chatelier *et al.* (2013): 70% of the whole sampled cohort was randomly chosen 30 times and significance was determined at $P < 0.05$ with bootstrap = 0.8 as a threshold. This process was repeated with another 30 iterations on the 48 most extreme cows (24 efficient and 24 inefficient). Overall, 18 significantly different species and 34 166 significantly different genes common to all 60 tests were further analyzed. Species and genes that were significantly different were correlated to the RFI parameter using Spearman correlation. Functional annotation of significant genes was achieved using BLASTP with $E \leq 10^{-6}$ against KEGG PATHWAY, MODULE, BRITE, GENES and ORTHOLOGY databases (2014; 25% annotation) (Kanehisa *et al.*, 2011). These genes were also blasted against the NR database (Pruitt *et al.*, 2007), and their phylogenetic annotation was determined according to the best hit (BLASTP with $E \leq 10^{-6}$; 89% annotation).

Statistical tests and estimation of false discovery rate

Tukey's, Student's *t* and Wilcoxon rank-sum tests were conducted depending on the normality of distribution of the input data. All tests were corrected for false discovery rate using the method described by Benjamini and Hochberg (1995) unless otherwise noted. In permutation *t*-test, significance of the difference between means was inferred by performing *t*-test between the two groups and comparing the resulting *t*-statistic to the *t*-statistics resulting from 9999 permutations of random group assignments (two-tailed, $P < 0.05$) (Davis, 1986). For multiple hypothesis correction, the distribution of *t*-test *P*-values was compared with the lowest *P*-values distribution resulting from 9999 permutations of random group assignments according to Westfall and Young (1993). This procedure was

performed using the R bioconductor package mult-test (Pollard *et al.*, 2005), function *mt.maxT*, individually for each metabolic or activity test, namely polymers, SCFAs, methane and all other measured metabolites. Variance similarity was tested where required by the statistical test.

Predictions of different physiological parameters

Feature selection of microbial species and genes was conducted by choosing species or genes that were significantly different in their presence/absence using the Fisher's exact test. Species and genes were sorted separately according to their *P*-value in ascending order and grouped into bins of 100 features. Each bin was used as predictive features for the feed efficiency phenotype using the *k*-nearest neighbors algorithm (Aha, 1997) with $k = 3$. The mean accuracy of the prediction was calculated using cross-validation of 1000 iterations for each bin, in which 70% of the samples were used as a training set and the remaining 30% were used as a test set to measure the accuracy of the prediction. Changing the bin size (bins ranging in size from 50 to 1000 features per bin) did not affect the accuracy of the prediction (Supplementary Data 3). To check the significance of the classifications accuracy, a permutations technique was used. The classification procedure was repeated 100 times, each time after randomly shuffling (permutating) the sample labels. The *P*-value for each classification accuracy was then obtained by the percentage of permutation runs in which the accuracy achieved was greater than the classification accuracy achieved with the original non-permuted data. The same prediction methodology, accuracy and *P*-value determination were applied to several other metabolic parameters—conversion ratio, milk yield, milk energy, milk lactose, milk fat, milk protein, body conditioning score, pH and DM intake. For each metabolic parameter prediction test, the cows were separated into two groups, by the physiological parameter's mean value (Supplementary Data 4).

For each physiological index, receiver operation characteristics curves and area under curve (AUC) measures were obtained based on the average of 1000 *k*-nearest neighbors cross-validation iterations. The analysis was performed with the Metrics class that is part of the SKLEARN python machine-learning framework.

Recruitment to microbial genomes and metabolic pathways

Reads from each sample were subsampled according to the sample with lowest number of reads (21 486 100). The reads from each sample were aligned using burrows-wheeler alignment tool to a data set of 59 microbial genomes downloaded from NCBI using burrows-wheeler alignment tool with 98% identity and default parameters. Reads were

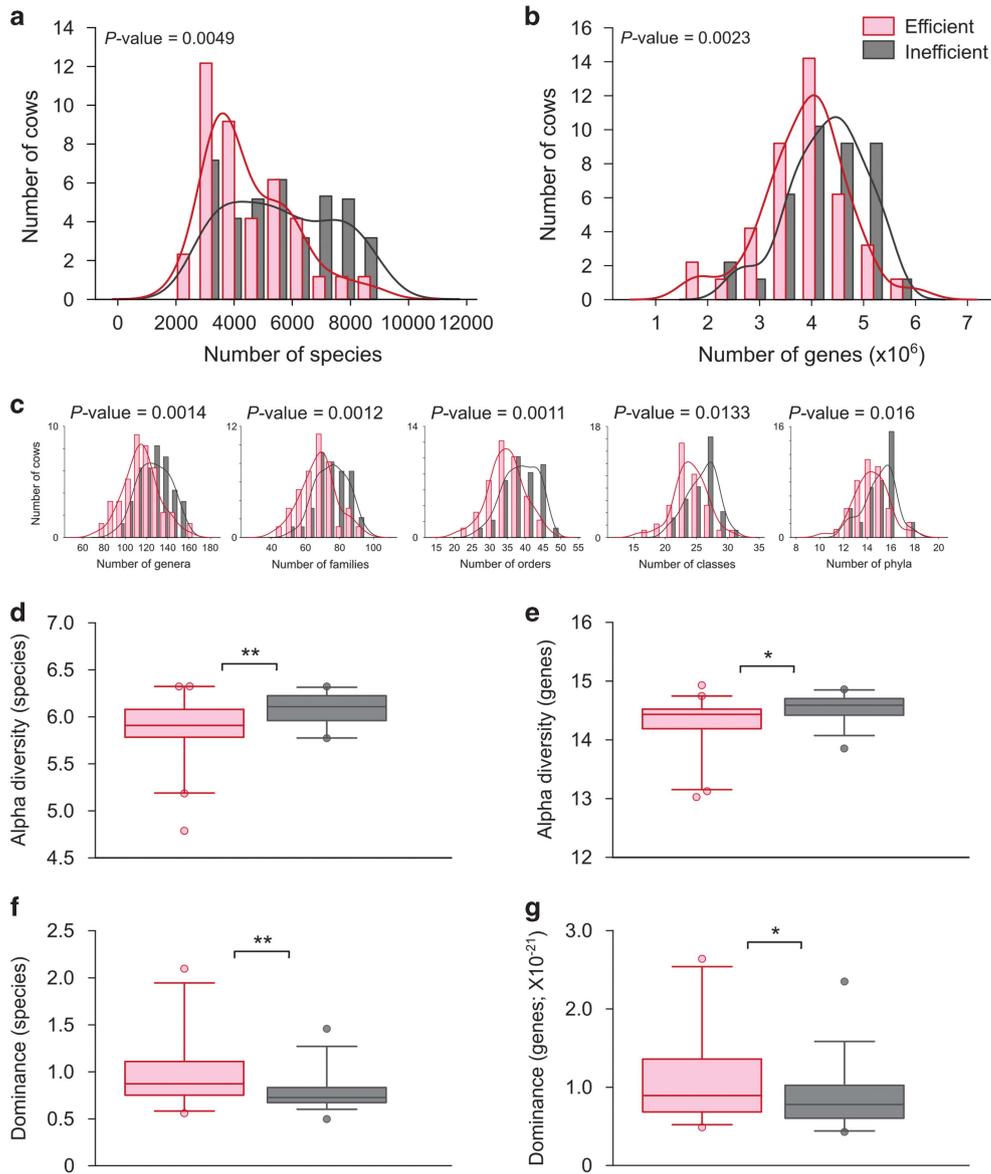


Figure 1 Community parameters of efficient and inefficient cows' microbiomes. (a, b) Microbiome richness. Species (based on 16S amplicon sequencing) (a) and gene (based on metagenomics sequencing) (b) counts were calculated and expressed as simple richness. Kernel density of the efficient and inefficient histograms emphasizes the different distribution of counts in each microbiome group. *P*-values of the difference in richness between efficient and inefficient cows are shown. (c) Microbiome richness at different phylogenetic levels. (d, e) Alpha diversity (Shannon index) measurements according to species (d) and genes (e). (f, g) Dominance of the microbiome according to species (f) and genes (g). Data are expressed as mean \pm s.e.m. Wilcoxon rank-sum, **P*<0.05, ***P*<0.01.

also recruited to metabolic pathways of the significantly different metabolites (*P*<0.05) using the same method. Our database consisted of all possible KEGG enzymes for each metabolic pathway. The EC numbers used for each metabolic pathway are described in Supplementary Table S1.

The existence of the propionate production acrylate pathway in the genomes of the examined lactate utilizers *Selenomonas ruminantium* and *Anaerobivrio lipolyticus* was additionally tested by blasting them against all possible KEGG enzymes belonging to the acrylate pathway (EC 1.3.8.7, 2.8.3.1 and 4.2.1.54) using a threshold of above 70% identity,

70% alignment length of the subject gene and $E \leq 10^{-5}$.

Results

Construction of a rumen metagenome reference data set
To determine whether there are microbiome features that are associated with the cow's energetic efficiency, the individual feed efficiency of 146 Holstein Friesian cows was first determined. Each animal was automatically monitored for multiple parameters used to calculate feed efficiency (using the RFI approach).

For further analyses, the upper and lower 25% of the animals that exhibited extreme feed efficiency values were chosen, for a total of 78 animals—40 efficient and 38 inefficient (Supplementary Figure S1 and Supplementary Data 1). Metagenomic DNA samples of these animals' rumen microbiomes were subjected to 16S rRNA gene sequencing and whole-genome shotgun sequencing. The metagenomics reads of all samples were pooled and assembled, and the predicted genes served as a reference data set (see Materials and methods section). The metagenome contained 96.72% bacterial sequences, 1.73% archaeal sequences and 1.34% eukaryotic sequences, similar to what was previously described for rumen microbiome metagenomes (Brulc *et al.*, 2009). None of the eukaryotic sequences showed significance in the analyses.

Microbiome features differ and can predict feed efficiency phenotype

A comparison of microbiome richness across the animals revealed significantly lower richness in the efficient cows' microbiomes in both species ($P=0.0049$) and gene content ($P=0.0023$; Figures 1a and b). The differences in taxon richness were apparent up to the phylum level (Figure 1c), further stressing the intensity of this phenomenon. Taxon composition and gene content were derived from two different procedures of sequencing and analysis, and therefore the agreement between these findings highlights the robustness of the observation. The differences in richness were also accompanied by significantly lower diversity and higher dominance in the efficient animals' microbiomes at the species and gene levels ($P<0.01$ and $P<0.05$, respectively, for both diversity and dominance; Figures 1d–g and Supplementary Table S2). These differences were apparent up to the family level (Supplementary Figure S5). These differences in microbiomes of efficient and inefficient cows begged the question of whether microbiome features could be used as markers for the feed efficiency trait.

Thereupon, the species and gene composition of the rumen microbiomes were used to successfully predict the animals' feed efficiency phenotypes with up to 91% accuracy using the k-nearest neighbors algorithm (Aha, 1997). For the feature selection process, a Fisher's exact test was used to measure differences in presence/absence between microbiomes of efficient and inefficient animals. The species and genes were ranked separately according to their P -values in ascending order and divided into bins of 100 features to be used for prediction. Each bin was tested for its ability to predict high or low feed efficiency. The mean prediction accuracy was calculated using cross-validation for each bin (1000 iterations). The first species bin's prediction accuracy was 80%, while the first gene bin reached an accuracy of 91% (Figure 2). The species prediction accuracy declined to 50% (accuracy of a random guess) after the fifth bin, whereas the decline in prediction accuracy for the genes followed a much more moderate slope, with the first four predictive bins at above 90% accuracy with highly significant P -values. These differences in the slope of prediction accuracy could stem from the fact that each species represents a single genome containing thousands of genes therefore declining more rapidly compared to bins composed of hundreds of single genes. Supplementary Data 5 and 6 contain the identity of species and genes of the first five bins.

The microbiome features were also highly predictive of other physiological parameters, such as milk lactose content and milk yield (Supplementary Figures S6 and S7). The sensitivity and specificity of the predictive bins was further assessed by performing receiver operating characteristic analysis for the first five bins, for both the species and genes data of each physiological parameter (Supplementary Figures S8 and S9). This analysis showed high sensitivity and specificity of the predictions of the host physiological traits based on these microbiome features, as the AUC index had high values that are considered to be good for the species data, and excellent for the genes data

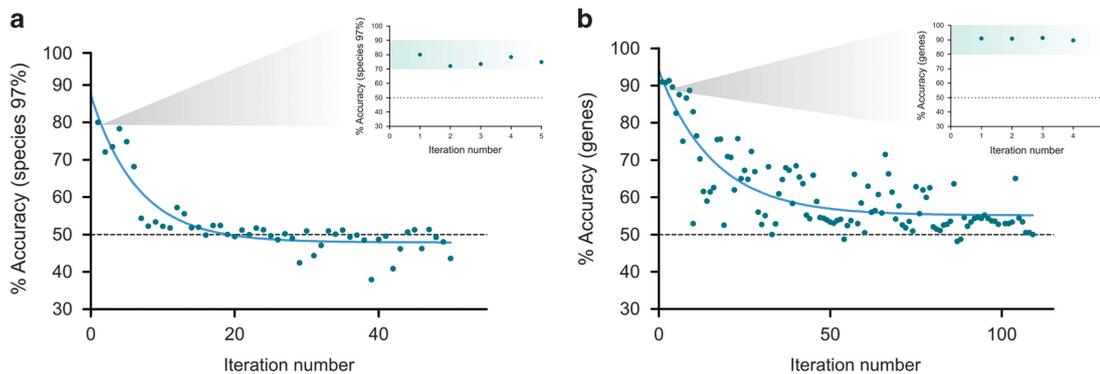


Figure 2 Feed efficiency predictions according to species and genes. Species (a) and genes (b) that differed in presence/absence between efficient and inefficient cows were ranked according to their P -values and grouped into bins of 100. The bins were used as predictive features for the RFI feed efficiency parameter using the k-Nearest Neighbors (KNN) algorithm with $k=3$. Each iteration used a different bin as predictive features, in ascending P -value order. Inset in both graphs represents the first five prediction accuracy values (permutations of random classes shuffling, P -value = 0.009).

(AUC > 0.8, AUC > 0.9 respectively). This high prediction accuracy indicated that the differences in microbiome gene content and taxonomic composition could be used to classify and predict the cow's energetic efficiency.

Microbiome metabolic activity varies in cows with different feed efficiencies

Diversity, richness and dominance are key ecological determinants that, when altered in a given ecosystem, are expected to have a marked effect on its functionality (Hooper *et al.*, 2005). Hence, following the findings of evident differences in these parameters (Figure 1, Supplementary Figure S5 and Supplementary Table S2), the functionality of the rumen ecosystem was further investigated. Several microbial activity assays, as well as a series of 41 metabolites, were targeted and measured, representing the processes and products of different trophic levels of the rumen microbiome from efficient and inefficient cows, starting from degradation of the ingested plant fiber to the end products (Figure 3).

Significant differences were discovered in most SCFAs. Out of the six SCFAs measured, four—

propionate, butyrate, valerate and isovalerate—were at higher concentrations in the rumen of efficient cows (Figure 3, metabolic end products and Supplementary Table S3). In addition, the total concentration of SCFAs was higher in the efficient animals showing an increase of 10% between the two efficiency groups ($P < 0.01$; Figure 4a). These differences are considered to have a marked effect on animal productivity, given that approximately 70% of the net energy requirements of the animal are supplied by SCFAs (Seymour *et al.*, 2005).

Interestingly, the propionate-to-acetate ratio in the efficient animals was also significantly higher than in the inefficient ones ($P < 0.05$; Figure 4b); an increase in this ratio is associated with a decline in methane production and increased energy retention by cattle (Russell, 1998). This finding was congruent with the measurements of the microbiomes' methanogenesis potential, where it was evident that the efficient cows' microbiomes produce significantly less methane than their inefficient counterparts ($P < 0.01$; Figure 3, metabolic end products). The finding of higher concentrations of SCFAs and lower methane emission from the efficient rumen microbiomes is consistent with the notion that

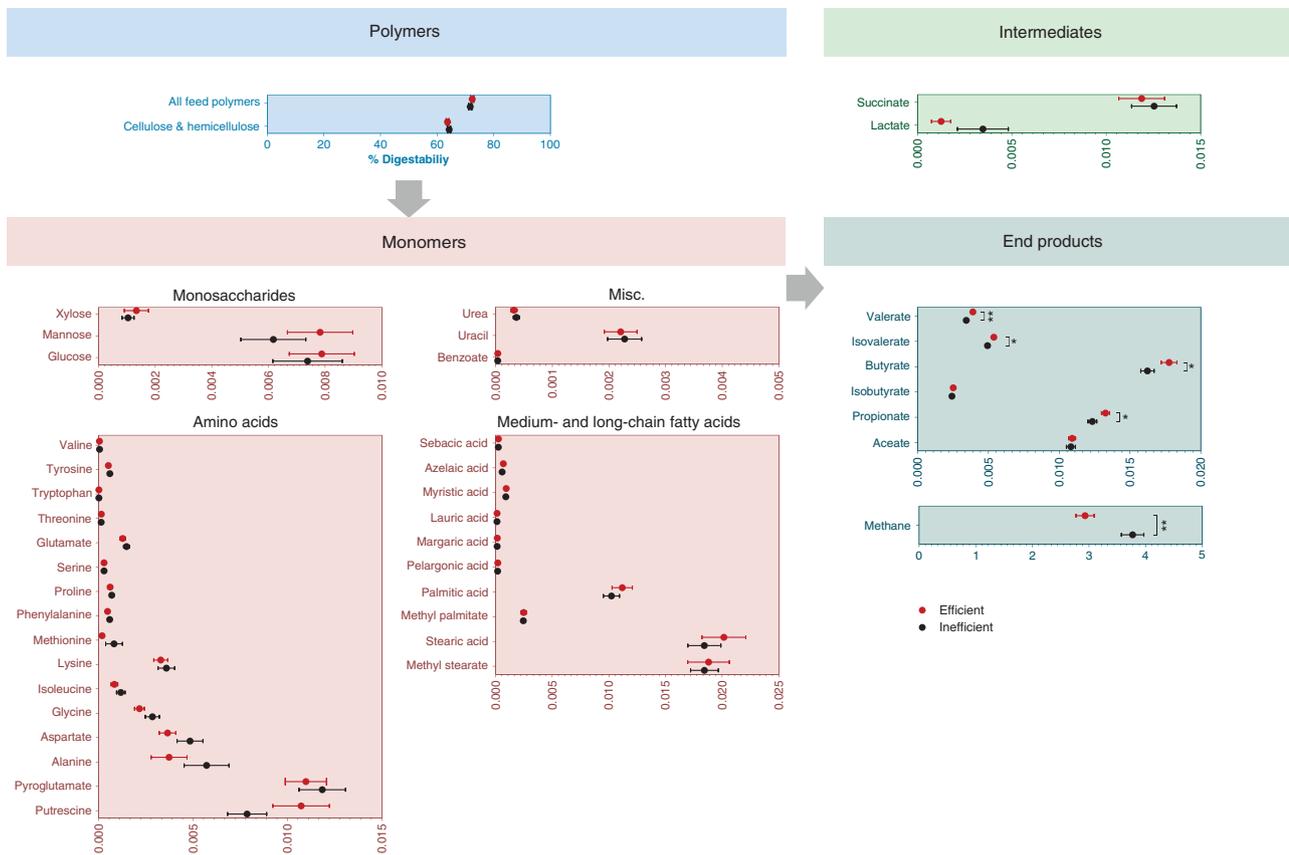


Figure 3 Metabolome and microbial activity of rumen microbiomes of efficient and inefficient cows. *In-vivo* and *in-vitro* digestibility methods were performed on rumen fluid of efficient and inefficient cows in addition to extraction, identification and quantification of 41 different metabolites by GC and gas chromatography mass spectrometry. These metabolites were normalized to the organic matter content of the rumen fluid from which they were extracted. Metabolites are organized according to trophic levels. Multiple hypothesis correction with 9999 permutations was performed individually for each metabolic or activity test using the *t*-statistic (Materials and methods section). Data are expressed as mean \pm s.e.m. * $P < 0.05$, ** $P < 0.01$.

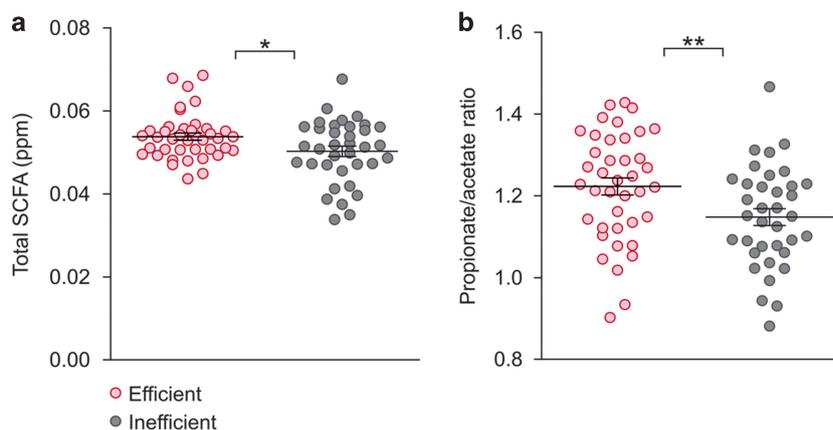


Figure 4 SCFA concentration in rumen fluids of efficient and inefficient cows. (a) Total SCFA concentrations in efficient and inefficient rumen samples. (b) Propionate/acetate ratio in the efficient and inefficient rumen samples. Data are expressed as mean \pm s.e.m. * $P < 0.05$, ** $P < 0.01$.

propionate and butyrate production competes with methanogenesis for hydrogen and presents an alternative mechanism that serves as an electron sink (Ungerfeld, 2015). The production of more SCFAs and less methane by the efficient cows' microbiomes is in agreement with the higher energetic efficiency.

Our analysis did not reveal any significant differences in the microbiomes' ability to degrade the plant cell wall in the diet, *in vitro* or *in vivo* (Figure 3, polymers and Supplementary Figure S10).

Differential abundance of rumen microbes and metabolic pathways

The lower diversity and higher dominance in gene content and taxonomic composition apparent in the microbiomes of efficient cows, together with changes in metabolite assortments, suggested that the flux through collective metabolic pathways is different in microbiomes of this efficiency group. This raised the hypothesis that this may be due to changes in the occupancy of specific rumen microbial niches, defined by metabolic and physical characteristics, by functional groups that differ in their resource demands or output products.

To explore this hypothesis, a permutative Wilcoxon rank-sum test was conducted in which gene and taxonomic profiles were compared between the microbiomes of efficient and inefficient animals (see Materials and methods section). Overall, 18 species and 34 166 genes differentiated the microbiomes of efficient and inefficient cows (Supplementary Figures S11 and S12, and Supplementary Data 7 and 8); of these, 2 species and 227 genes were more abundant in efficient cows. These species and genes were not only differentially abundant in cows with different RFI values, but were also significantly correlated to the intensity of the phenotype (Figure 5a and Supplementary Data 7). The lower numbers of species and genes that were more abundant in the efficient cows' microbiomes are compatible with the higher dominance and lower

richness in species and gene composition of these microbiomes. The annotation and analysis of the differentiating genes against the KEGG database (Kanehisa *et al.*, 2011) were also in agreement with these findings, as well as with the metabolomic analysis. Among the KEGG pathways and resultant metabolites that were enriched in the inefficient cows' microbiomes were enzymes from the protein digestion and absorption category, amino-acid biosynthesis and the methane metabolism category (ko numbers of these pathways are detailed in Supplementary Table S4). Furthermore, a significantly lower number of KEGG pathways were enriched in the efficient cows' microbiomes, resulting in a significantly lower number of potential products (Supplementary Figures S13 and S14, Supplementary Data 9).

These findings suggest that there is more diverse use of resource compounds, such as dietary proteins, pyruvate, acetyl-CoA and hydrogen, in the inefficient cows' microbiomes, resulting in a more diverse array of produced metabolites, some of which affect the animal's energy harvest in a negative manner or cannot be utilized by the animal for its energy requirements. In the efficient cows' microbiomes, the use of these compounds is dominated by a limited number of metabolic pathways that are more relevant and valuable for the energy needs of the animal.

The phylogenetic annotations of genes that were enriched in the efficient cows' microbiomes were dominated by the rumen bacterial species *Megasphaera elsdenii*, a highly potent utilizer of lactate for the production of butyrate and propionate (Figure 5b). This annotation, or any other closely related annotation, did not appear in the inefficient cow microbiomes' enriched genes. Overall, the inefficient cows' microbiomes were less dominated by a specific taxon unique to that microbiome group (Figure 5b), further supporting the hypothesis of higher dominance of specific functional groups in the microbiomes of efficient cows. This was also reinforced by the annotation of the two species that were significantly more abundant in the efficient cows' microbiomes in the 16S rRNA gene analysis. One

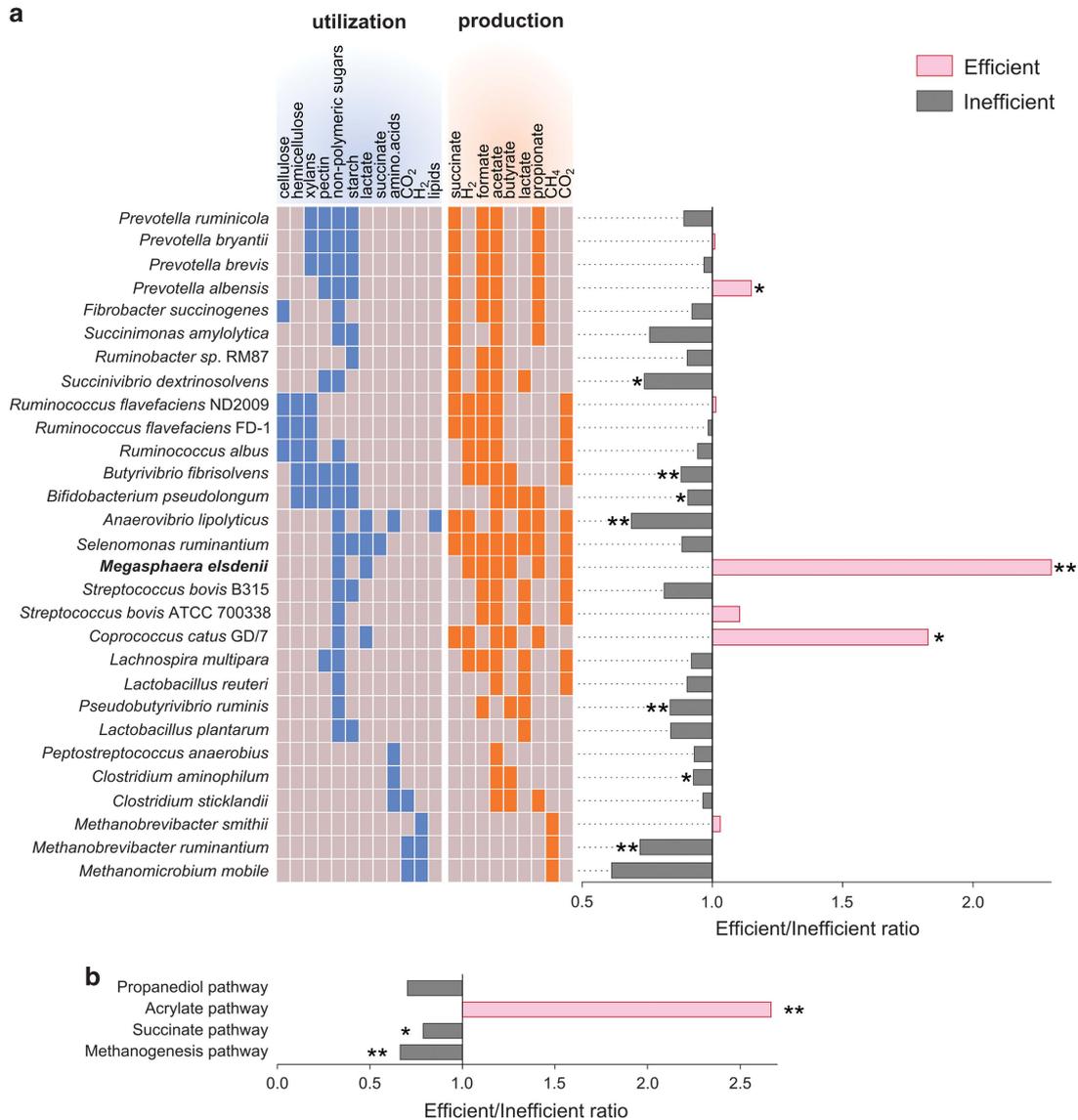


Figure 6 Microbiome features enriched in each microbiome group. (a) Reads from each sample were aligned to sequenced genomes of known rumen microorganisms using the burrows-wheeler alignment tool. The ratios between alignments of efficient/inefficient samples to each genome are presented. The utilization and production of metabolites for each microorganism based on the known growth characteristics (Holdman and Moore, 1974; Russell and Rychlik, 2001; Duncan *et al.*, 2009) are colored in blue and orange, respectively. (b) Reads from each sample were aligned to KEGG enzymes of different metabolic pathways using the burrows-wheeler alignment tool. Propanediol, acrylate and succinate pathways are different propionate production pathways. The ratios between alignments of efficient/inefficient samples to each pathway are presented. Data are expressed as ratio of means. Permutations *t*-test, **P*<0.05, ***P*<0.01.

aligned to this pathway are predominantly annotated as *M. elsdenii* and *C. catus*, however, annotations of *Clostridium propionicum* and *Clostridium botulinum* were also detected (Supplementary Figure S16). This highlights the acrylate pathway as the main contributor to the increase in propionate and decrease in lactate observed in the metabolomic analysis of the efficient cows' microbiome group (Figure 3).

Discussion

Our analyses of multiple animals feeding on the exact same diet and kept under the same conditions showed

that there are large variations in the individual animals' ability to extract energy from their feed. These variations are tightly linked to several microbiome features that include a decrease in richness and increase in dominance of taxonomic and coding capacity in the efficient cow's microbiome. They are reflected as changes in this ecosystem's functionality, where changes in the dominance of specific functional components affect the overall availability of ecosystem goods that are of high value to the hosting animal. Higher microbiome richness and changes in specific functional groups have been recently described to affect host productivity in plants (Wagg *et al.*, 2014) as well as humans, where lower diversity

and richness have been associated with higher energy harvest from feed in obese humans (Turnbaugh *et al.*, 2009; Le Chatelier *et al.*, 2013). A possible explanation for this phenomenon could stem from a more diverse use of resource compounds in the inefficient cow's microbiomes that are enriched in species, genes and KEGG pathways resulting in a wider array of output metabolites (Figures 3, 6 and 7); this was also confirmed by significantly more

KEGG output metabolites (Supplementary Figures S13 and S14). On the other hand, in the efficient cow's microbiome, simpler metabolic pathway networks result in increased dominance of specific functional components, which leads to higher concentrations of ecosystem goods that are relevant to the host (Figure 7b). Therefore, the efficient microbiomes are less complex but more specialized to support the host's energy requirements.

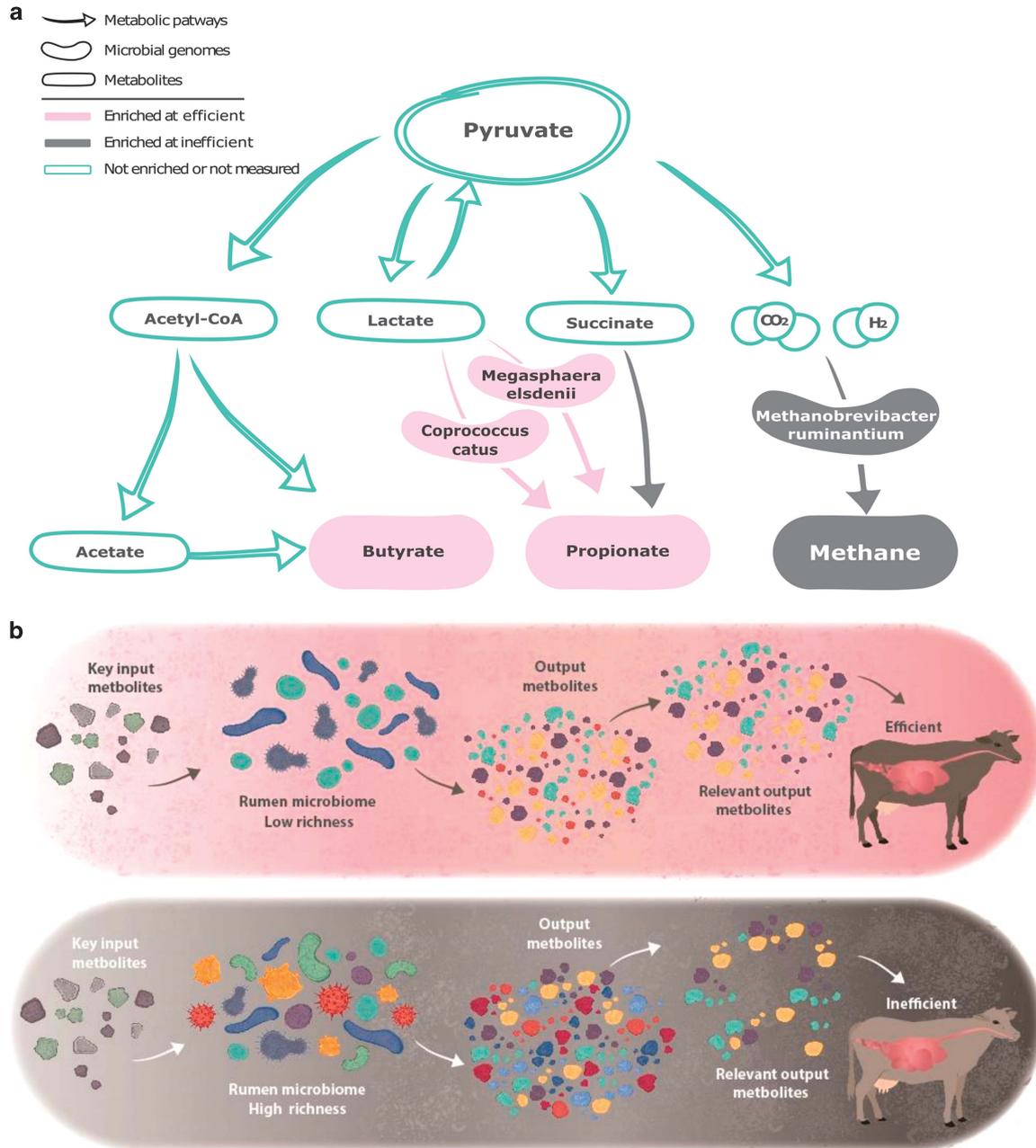


Figure 7 Consolidated results and model. (a) Consolidation of results from the metabolomics, genome and pathway recruitment analyses. Green: pathways and metabolites that were not significantly different or that were not assessed. Pink: enriched in inefficient microbiomes. Grey: enriched in efficient microbiomes. (b) Proposed model. From left to right: identical key input metabolites are ingested by the cow and presented to either an efficient microbiome (top panel) with lower richness and diversity, or an inefficient microbiome (bottom panel) with higher richness and diversity. Differences in richness result in the production of different metabolites. The efficient microbiome produces a smaller range of output metabolites than the inefficient microbiome, however, with larger amounts of relevant output metabolites, which are available for the animal's energetic needs.

This notion is exemplified by the finding of higher concentrations of SCFAs, which are valuable to the hosting animal. SCFAs are absorbed through the rumen wall to serve the energetic needs of the animal; propionate, for example, is the main precursor for gluconeogenesis in animals (Russell and Wilson, 1996; Mizrahi, 2011; Mizrahi, 2013). This is not the case with methane as the energy retained in it cannot be absorbed by the animals, and is lost to the atmosphere. Such metabolic changes are usually achieved via the use of antibiotic growth promoters that increase the animal's feed efficiency (Duffield *et al.*, 2012). Such is the case with monensin, a carboxylic polyether ionophore that selectively affects some of the rumen microbes, therefore changing the structure of the rumen microbiome and subsequently the ratio of SCFAs in the rumen, increasing propionic acid and decreasing methane production (Thornton and Owens, 1981; Callaway *et al.*, 2003; Weimer *et al.*, 2008; Duffield *et al.*, 2012). It has been shown that when administered orally, monensin improves feed efficiency in cattle in a dose-dependent manner. Therefore, it has been used for this purpose extensively since its approval for cattle agriculture in the mid-1970s (Duffield *et al.*, 2012). This effect of rumen microbiome manipulation achieved via antibiotics further supports the connection of the rumen microbiome with the feed efficiency of the animal.

Here we show that these metabolomic changes are the outcome of microbiome structures that are naturally occurring and are highly correlated with, and predictive of the feed efficiency phenotype. Therefore, these findings could be harnessed to reduce the use of antibiotic growth promoters in agriculture. Such prospects, together with examination of the true causal nature of the rumen microbiome on its host, would be made possible when non-antibiotic rumen microbiome manipulation techniques, as well as adult cow germ-free facilities, will be better established.

From an ecological perspective, the lower abundance of methanogenesis pathways and methanogenic archaea in the efficient cow's low-richness microbiome concurs with the notion that processes that are performed by small taxonomic groups, such as the methanogenic archaea that occupy only small percentages of the rumen microbiome, are more sensitive to changes in diversity and richness (Hooper *et al.*, 1995). These changes are usually accompanied by occupation and dominance of the available niche by different species using the same resources (Grime, 1998). Such is the case with *M. elsdenii* and *C. catus*, independently found to be enriched in the efficient animals' microbiomes in different analyses (Figures 5 and 6a, Supplementary Figure S15), which use electrons for the production of the valuable SCFAs propionate and butyrate, thereby diverting them from reducing CO₂ to methane (Prabhu *et al.*, 2012; Ungerfeld, 2015). A similar principle was shown to apply in Tamar wallabies, where

Succinivibrio bacteria were suggested to utilize hydrogen for the production of succinate, therefore lowering its availability for methanogenesis (Pope *et al.*, 2011). It is also possible that the Lachnospiraceae detected in the efficient animals' microbiomes are butyrate producers (Figure 5) and are contributing further to this effect (Louis and Flint, 2009; Meehan and Beiko, 2014). Nevertheless, as other SCFAs are enriched in this microbiome group and most of the carbon flux in the system goes to acetyl-CoA, formate or hydrogen and carbon dioxide, it is likely that more genes and pathways are involved in this effect.

A cardinal point that emerges from our findings is that the functional characteristics of a small number of species can have a large impact on community structure and ecosystem functioning. This, in turn, can change the productivity of the supraorganism—the host and its residing rumen microbiome.

These findings could potentially be harnessed to increase the production of food resources for mankind in a more sustainable manner, as well as to understand the underlying ecological mechanisms that govern complex microbial communities and their interactions with their hosts.

Conflict of Interest

The authors declare no conflict of interest.

Acknowledgements

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Data deposition

Assembled Contigs were deposited in the MG-RAST server under IDs 4547164.3 and 4548996.3.

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