Optimize sequencing depth required for reliable profiling of microbiome by rarefaction and multimodel inference, using shotgun metagenomic data from pollination system and a modular profiling pipeline

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Declaration:

All datasets used in this project are provided by Dr. Peter Graystock and the project was conducted under his supervison. I am responsible for the development and conduction of the analyses presented.

Abstract

Bee-associated microbiome plays a crucial role in host health and relevant investigations have been boosted by shotgun metagenomics, which provides unique advantages by capturing diversity of multiple taxonomic clades and providing information on function potentiality. However, utilization of shotgun metagenomics is hindered by complexity of data analysis and high cost of sequencing. Here, an integrated pipeline combining assembly-dependent and -independent methods was introduced for taxonomic and functional profiling of shotgun metagenomic data, and a framework of rarefaction and multimodel inference was constructed for optimizing sequencing depth. Both the pipeline and the framework were used for analysis of real metagenomic datasets ($2 \times 150bp$ read pairs) from honey bees, bumble bees and flower washes. The integrated pipeline illustrated taxon composition and metabolic potentiality of metagenome, and provided improvement in species identification compared with an assembly-dependent procedure. Sequencing depth can be optimized for species diversity estimation especially when little emphasis is given to rare species, while functional profiling requires deeper sequencing than taxon profiling. These results are helpful for cost-effective utilization of shotgun metagenomics in investigations of pollination system.

1 Introduction

Pollinators such as bees are crucial to maintain global food security and provide stability of natural systems (Hristov et al., 2020a, Bänsch et al., 2021, Khalifa et al., 2021) but their health is under increasing concern (Amiri et al., 2020). Diverse pathogens and environmental factors threaten bee health and are contributing to their declines. A combination of stressors including parasites, pesticide exposure, invasive species, habitat loss and climate change are all contributing to declines of bee populations. (Brown and Paxton, 2009, Hristov et al., 2020b, Cheng and Ashton, 2021, Zattara and Aizen, 2021).

Bees are associated with a community of microorganisms influencing host health (Engel et al., 2016, Raymann and Moran, 2018). Within the gut of bees, stable microbial communities (microbiomes) play crucial roles in food digestion, parasite defence and chemical detoxification. They mediate food digestion processes including polysaccharides breakdown (Zheng et al., 2019), sucrose hydrolysis (Engel et al., 2012, Lee et al., 2015), and mannose metabolism (Engel et al., 2012, Lee et al., 2015). They also provide protection against pathogens including *Crithidia*, (Koch and Schmid-Hempel, 2011b, Cariveau et al., 2014), *Paenibacillus larvae*, (Ebeling et al., 2016, Forsgren et al., 2010) and *Nosema sp.* (Cariveau et al., 2014, Maes et al., 2016). Besides, bee-associated symbionts can aid in detoxification. They are involved in resistance to both metal and metalloid toxins including cadmium (Rothman et al., 2019b), copper (Rothman et al., 2020) and selenate (Rothman et al., 2019a).

Amplicon sequencing, in which a species-specific barcode region is amplified and sequenced, is a powerful

method for investigations of microbiome (Abdelfattah et al., 2018). It is vastly used in bee microbiome investigations to illustrate the taxonomic diversity of bee-associated bacteria and fungi (*e.g.* Geldert et al. (2021), Wang et al. (2021), Powell et al., Kapheim et al. (2021)). However, bees visit numerous niches during environment exploration and foraging activities, and can get contact with diverse eDNA signatures, which provide insight into bee ecology by reflecting interactions between bees and other organisms including bacteria, fungi, plants, arthropods and viruses (Bovo et al., 2018, Ribani et al., 2020, Bovo et al., 2020, Matsuzawa et al., 2020). It is difficult to explore this diversity via amplicon sequencing because it only captures a fraction of the whole community since analysis of different taxonomic groups is based on different barcode regions, *e.g.* 16S ribosomal RNA (rRNA) for bacteria (Hayashi et al., 2002, Eckburg et al., 2005), internal transcribed spacer (ITS) for fungi (Nilsson et al., 2008), cytochrome c oxidase subunit I (COI) for Animalia (Hebert et al., 2003) and plastid genes for plants (Group et al., 2009). As a result, amplicon sequencing only captures taxon diversity within a certain clade. Besides, it is difficult to illustrate function potentiality of bee-associated microbiome using amplicon sequencing since it does not provide information on content of functional gene clusters (FGCs), *i.e.* aggregates of genes with same function. As a result, functional capacity needs to be inferred based on reference genomes (Aßhauer et al., 2015, Douglas et al., 2018). However, bee bacterial symbionts are diversified at strain level (Engel et al., 2012, Powell et al., 2016, Ellegaard et al., 2020) and bacterial strains are often highly variable in gene content (Cordero and Polz, 2014, Brockhurst et al., 2019). As a result, amplicon sequencing-based inference of functional capacity of bee microbiome may not be reliable.

Shotgun metagenomics provides an alternative for microbiome investigations to overcome drawbacks of amplicon sequencing. By capturing and sequencing DNA fragments unselectively, shotgun metagenomics is capable of providing comprehensive inventories of taxa and FGCs (Quince et al., 2017, New and Brito, 2020, Galloway-Peña and Hanson, 2020). However, utilization of shotgun metagenomics is hindered by challenges in bioinformatics. Typical goal of metagenomics is to provide taxonomic and functional profile of microbiome, and there is not a golden standard for bioinformatics of metagenome. Generally, one of the first steps in metagenomic analysis is assembling short reads into long contigs, which can help improve accuracy of metagenomic annotation (Wommack et al., 2008, Carr and Borenstein, 2014, Tran and Phan, 2020) and is necessary for discovery of novel taxa and genes (Culligan et al., 2014, Youngblut et al., 2020). However, metagenome assembly is complex, compromised by fragmental assembly, chimaeras (Mikheenko et al., 2016) and loss of taxon/function diversity due to unassembled reads (Vollmers et al., 2017, Ayling et al., 2020). Probably because of these shortcomings, assembly is skipped in some researches and short reads are directly proceeded for annotation (Tringe et al., 2005, Abubucker et al., 2012, Vermote et al., 2018, Bovo et al., 2018), although the accuracy can be compromised due to low information load of short

reads (Wommack et al., 2008, Carr and Borenstein, 2014, Tran and Phan, 2020). A combination of both assembly-dependent and -free methods could overcome the complexity and improving accuracy of metagenomic profiling (Becker et al., 2020).

A remaining challenge of metagenomics is the determination of a sequencing depth that provides reliable estimation of taxon/FGC diversity without overspending. It is recommended to retrieve as many reads as possible (Quince et al., 2017) since insufficient sequencing causes compromise in metagenome profiling (Cattonaro et al., 2018, Zaheer et al., 2018, Pereira-Marques et al., 2019, Gweon et al., 2019). However, deep metagenomic sequencing is expensive, which hinders its utilization, especially in large-scale projects. Currently, there are few published guidelines for the sufficient sequencing depth of a given environment or study type in order to reach a trade-off between sequencing effort and reliable output.

In order to balance sequencing cost and reliable estimation of taxon/FGC diversity, expected diversity represented by given sequencing depth need to be computed. Since a metagenomic dataset can be viewed as a random sample of an assemblage of genomic sequences, and profiling is the process by which reads are assigned to taxa or FGCs, the relationship between sequencing depth and diversity can be illustrated by rarefaction (randomly subsampling the original dataset without replacement) and quantified by model fitting, if the original dataset can provide an almost complete inventory of taxa/FGCs (Heck Jr et al., 1975, Hortal and Lobo, 2005, Gómez-Anaya et al., 2014, Hughes et al., 2021). The sequencing depth is sufficient for reliable estimation of taxon/FGC diversity if and only if the slope of the model is small (Hortal and Lobo, 2005, Chao and Jost, 2012, Roswell et al., 2021).

In this project, I utilised metagenomes from three environmental types: the gut of a honey bee (*Apis mellifera*), the gut of a common North American bumble bee (*Bombus impatiens*) and the surface of a flower (*Erigeron annuus*). I aimed to (1) develop an integrated pipeline combining assembly-dependent and -independent methods to deliver improved taxon annotation of sequencing data and (2) optimize sequencing depth to balance sequencing cost and requirement for reliable analysis of microbial species diversity, or the description of their functional diversity. In the integrated pipeline (Figure 1), assembly-dependent taxon profiling is conducted after quality filtering and removing host contamination. To address high negative rate of assembly-dependent search, a reference database of genomes from species present in the assembly is constructed and used for filtering non-host reads. Reads not recruited by the reference database are subjected to assembly-free taxonomic search. This integrated pipeline was used to profile metagenomic datasets involved in this project. Then in order to estimate optimal sequencing depth for reliable representing taxonomic/functional diversity of metagenomes, I simulated different sequencing depth by rarefaction and profiled subsampled datasets using the integrated pipeline, generating inventories of species and FGCs represented by Kyoto Encyclopedia of Genes and Genomes (KEGG) orthologies (KOs)

(Kanehisa and Goto, 2000). Diversity of species and KOs was measured by Hill numbers of order 0, 1 and 2. Sequencing depth was measured by number of clean read pairs (150 bp read length). The relationship between Hill numbers and sequencing depth was quantified by fitting and averaging asymptotic species accumulation models, and estimation of optimal sequencing depth was given by the point where the slope of rarefaction curve drops to cut-off values.

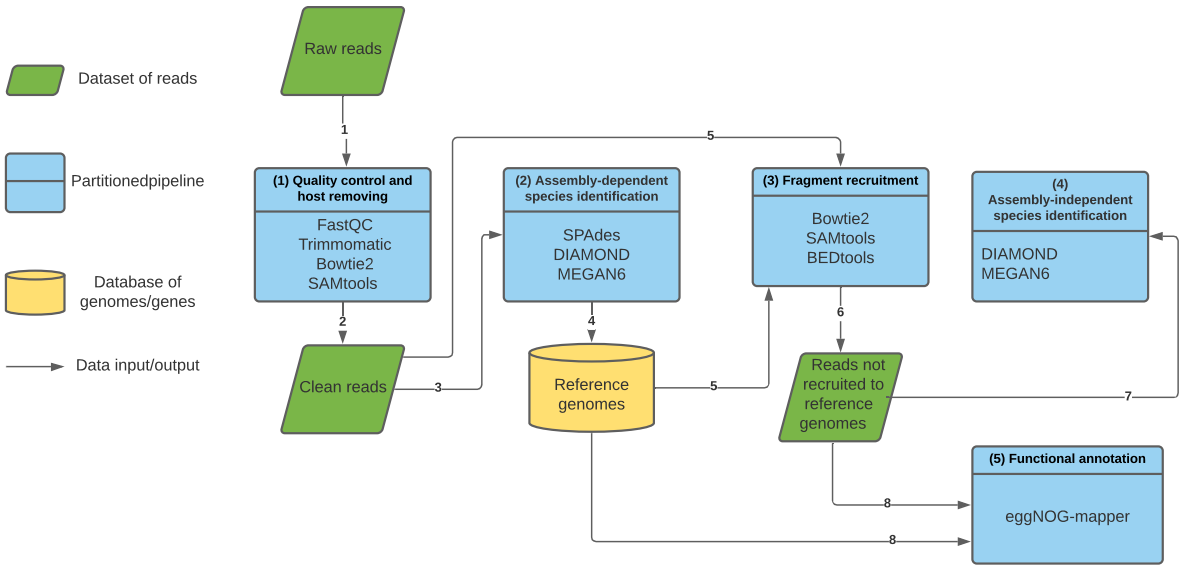

Figure 1: An overview of the integrated pipeline. The pipeline is separated into 5 modules, indicated by blue boxes. Green boxes indicate dataset of reads, while yellow boxes indicate database of genomes. Black arrows indicate the input and output of each step and the numbers on them indicate the order that each step is utilized.

2 Materials and Methods

2.1 Samples, DNA extraction and sequencing

Samples include four honey bees (two from the same hive (hive 13), one from a hive in the same apiary (hive 15) and one caught foraging), three bumble bees from commercially supplied (Biobest) bumblebee colonies (two from the same colony), and one buffer wash of a wild flower (*Erigeron annuus*).

DNA extraction was performed as in Graystock et al. (2020), followed by library preparation using a template protocol with Illumina Nextera Library Prep kits. Briefly, this involved tagmentation into fragments of 300 bases before eight samples of 10ng were pooled together and sent to Beijing Genomics Institute for further quality control and sequencing using a full lane in the X-ten platform.

2.2 Metagenomic profiling using integrated pipeline

2.2.1 Integrated pipeline

The integrated pipeline was designed for shotgun metagenomic profiling, *i.e.* assigning reads to taxa or FGCs. It is separated into five modules (Figure 1).

In quality control and host removing, raw sequencing data quality is checked using FastQC v.0.11.5 (Andrews et al., 2010) and filtered by Trimmomatic v.0.39 (Bolger et al., 2014). Then clean reads are mapped to host genome using Bowtie2 v.2.4.2 (Langmead and Salzberg, 2012) and non-host reads are extracted by SAMtools v.1.11 (Li et al., 2009).

The non-host reads are subject to the module of assembly-dependent species identification. *De novo* assembly is conducted by SPAdes v.3.15.2 (Prjibelski et al., 2020). Assembled contigs are aligned to NCBI non-redundant (nr) database by DIAMOND v.2.0.7.145 (Buchfink et al., 2015), and assigned to taxa by MEGAN6 (Huson et al., 2007).

Then fragment recruitment is conducted. A reference database is constructed and it comprises reference genome dataset, *i.e.* genomic sequences in FASTA format and corresponding genome annotation in general feature format (gff). For each species represented by assembly, its reference genome dataset, if available, is downloaded from NCBI using its *datasets* command-line tool and added to the reference database. Then non-host reads are mapped to the reference database by Bowtie2, and unmapped reads are extracted by SAMtools.

Reads not recruited by the reference database are subjected to assembly-independent species identification. They are aligned to NCBI nr database through DIAMOND and assigned to taxa by MEGAN6.

Finally, functional annotation is conducted by EggNOG-mapper v.2.1.2 (Huerta-Cepas et al., 2017). It takes coding sequences (CDSs) of genomes in the reference database and reads subject to assembly-independent species identification as input and assigns them to KOs.

The integrated pipeline was used for analyzing metagenomic datasets involved in this study and details in parameter settings of each module are described in Supplementary 7.1.

2.2.2 Taxon/function quantification and metabolic pathway reconstruction

After profiling, identified species and KOs were quantified by calculating relative sequence abundance, *i.e.* proportion of reads assigned to a species/KO in all reads annotated. For species without available reference genomes, their abundances were calculated using reads assigned to them in assembly-independent search. As for taxa with available reference genomes, they may be identified in both assembly-dependent and -independent search due to strain-specific genomic structures that are not present in reference genomes. Their abundances were calculated by summating number of reads that (1) mapped to coding sequences

(CDSs) of reference genomes and (2) assigned to them in assembly-independent search. Reads mapped to non-coding regions were not taken into consideration in order to avoid overestimation since the assembly-independent search was based on aligning reads to nr database, which is composed of proteins. As for KO quantification, CDSs with zero-coverage were excluded. Abundances of KOs were calculated by summating number of reads that (1) mapped to CDSs assigned to KOs and (2) assigned to KOs directly. Extraction of CDSs and calculation of their coverage were conducted by BEDtools v.2.30.0 (Quinlan and Hall, 2010).

Metabolic pathways were inferred based on KOs. Reads assigned to plants and arthropods were not included since they were unlikely to represent living organisms. MinPath v.1.6 was used for pathway inference (Ye and Doak, 2009). It finds a minimal set of KEGG pathways that can explain all KOs provided as input.

2.3 Estimation of optimal sequencing depth required for metagenomic profiling

2.3.1 Simulating different sequencing depth by subsampling and measuring diversity by Hill numbers

Species/KO inventories obtained from different sequencing depth was simulated by rarefaction. Since the ratio between numbers of raw and clean reads is dependent on sequencing process and is not influenced by sample type, sequencing depth here refers to number of clean read pairs to exclude variance caused by different proportion of low quality reads in samples of same type. Besides, the expected ratio between host and non-host reads in a metagenomic dataset is dependent on the DNA sample and not impacted significantly by sequencing depth. Thus the proportion of non-host reads in each simulation is expected to be the same with that in the original dataset.

Based on these considerations, I randomly subsampled non-host dataset of each sample, taking 10%-100% of read pairs at interval of 10% by *reformat.sh* script of BBmap v.38.90 (Bushnell, 2014), and profiled subsampled datasets by the integrated pipeline (Figure 1). The sequencing depth of each subsampled dataset equals number of subsampled non-host read pairs divided by ratio between non-host and clean read pairs. Thus, each subsampled dataset of non-host reads is corresponded to an imaginary dataset of clean reads, whose proportion of non-host reads is the same with that of the original metagenomic dataset.

After profiling subsampled datasets, species/KO diversity was measured by Hill numbers of order q, defined as Equation 1 (Hill, 1973).

$$D^{(q)} = (\sum_i (p_i)^q)^{\frac{1}{1-q}} \tag{1}$$

p_i represents the relative abundance of ith species/KO, and q determines sensitivity to relative abundances. When $q = 0$, abundances are not taken into consideration and $D^{(0)}$ equals species/KO richness. When $q = 1$, Hill number is defined as the limit of Equation 1 as q tends to 1 (2) and emphasis is given to species/KOs with general abundances.

$$D^{(1)} = e^{-\sum_i p_i log p_i} \tag{2}$$

When $q = 2$, high leverage is provided to abundant species/KOs and Hill number equals the inverse of Simpson index (Equation 3).

$$D^{(2)} = \frac{1}{\sum_i (p_i)^2} \tag{3}$$

2.3.2 Quantification of relationship between sequencing depth and Hill numbers by fitting rarefaction curves

Hill number of order q (Equation 1) measures diversity of an inventory as the number of equally abundant categories in an imaginary inventory with the same diversity (Chao et al., 2014, Roswell et al., 2021). Order q determines leverage given to abundant categories. All Hill numbers behave in the following way: if a proportion of categories in an inventory was removed randomly, all Hill numbers would decrease by that proportion (Roswell et al., 2021). Thus, it can be hypothesized that as sequencing depth (number of clean read pairs) increases, the detection of novel species/KOs leads to increase of Hill numbers, and when sequencing depth is so big that all species/KOs present in the metagenomic DNA sample have been detected, Hill numbers level off. Such a relationship can be fitted by asymptotic species accumulation models.

Let Hill number of order q (Equation 1) be a function of sequencing depth x, which takes million read pairs as the unit. This function was fitted using a multimodel inference method. First, a total of five candidate models (Table S1) were fitted to rarefaction curves which plots Hill numbers against sequencing depth. R package *minpack.lm* v.1.2.1, which employs Levenberg-Marquardt nonlinear least-square algorithm, was used for model fitting. Then small sample unbiased Akaike information criterion (AICc) (Anderson, 2007) of each candidate model was calculated (Equation 4):

$$AICc = -2L + 2k + \frac{2k(k+1)}{(n-k-1)} \tag{4}$$

where n is number of observed data points ($n = 10$ in this study), k is the number of fitted coefficients, and L is maximized log-likelihood, given by Equation 5.

$$L = -0.5nlog(\frac{Rss}{n}) \tag{5}$$

Rss represents residual sum of squares.

Then model averaging was conducted. First, differences of AICc scores between ith candidate models and the model with lowest AICc value were calculated using Equation 6.

$$\Delta_i = AICc_i - AICc_{min} \tag{6}$$

$AICc_i$ is the AICc score of ith plausible model and $AICc_{min}$ is the lowest AICc score among all candidate models. The Akaike weight of ith model is given by Equation 7 (Anderson, 2007).

$$w_i = \frac{e^{(-0.5\Delta_i)}}{\sum_i e^{(-0.5\Delta_i)}} \tag{7}$$

Denote ith candidate model by $D_i^{(q)} = D_i^{(q)}(x)$, the averaged model is given by Equation 8.

$$D^{(q)}(x) = \sum_i w_i D_i^{(q)}(x) \tag{8}$$

The slope of rarefaction curve was calculated by first derivative of averaged model (Equation 9). It reflects the increase rate of the curve.

$$\frac{dD^q}{dx} = \sum_i w_i \frac{dD_i^{(q)}}{dx} \tag{9}$$

The asymptote of rarefaction curve as sequencing depth tends to infinity is given by Equation 10. It provides an estimation of the total diversity and is comparable among metagenomic DNA samples with different sequencing depth. However, the accuracy of asymptotic estimators is controversial (Colwell and Coddington, 1994, Chazdon et al., 1998, Jimenez-Valverde et al., 2006, Hortal et al., 2006).

$$\lim_{x\to+\infty} D^{(q)}(x) = \sum_i w_i \lim_{x\to+\infty} D_i^{(q)}(x) \tag{10}$$

2.3.3 Estimating optimal sequencing depth using rarefaction curves

Optimal sequencing depth is defined as the point at which diversity starts to level off as sequencing depth increases, and its precise estimation via rarefaction is based on the assumption that the original dataset is sufficient for detection of almost all species/KOs present. This assumption can be verified by looking at

the rarefaction curve that plots species/KO richness (Hill number of order 0) against sequencing depth. The original dataset is sufficient for providing a reliable inventory if and only if the rarefaction curve of richness is characterized by a small final slope (Heck Jr et al., 1975, Hortal and Lobo, 2005, Chao and Jost, 2012). Then an estimation of optimal sequencing depth is provided by the point at which the slope of rarefaction curve decreases to a given cut-off value (Hortal and Lobo, 2005, Gómez-Anaya et al., 2014).

3 Results

3.1 Sequence reads

Eight samples (four honey bees, three bumble bees and one flower eDNA) were sequenced. The quality reports of raw reads showed low-quality 3'-end (Figure S1a), uneven base content in 5'-end (Figure S1b) and the present of adaptors (Figure S1c). The quality control procedure covered these aspects and improved data quality (Figure S1d, S1e and S1f).

After quality control, read pairs aligned to host genome were removed. Table S2 reports numbers of raw, clean and non-host read pairs. Honey bee sample *Bee_Amellifera_13_1* was filtered from further analysis since its low raw read pair number (1.10 million compared to the other samples at 59 million) is suggestive of a poor quality sample. After quality control, 62.08%-76.52% of raw read pairs were retained for these three honey bee samples. Then a different proportion of non-host read pairs (29.52%-86.19%) were retained. As for bumble bee samples, about 58 million raw read pairs were obtained for each sample and 82.1%-83.8% were retained after quality control. After host removing, 7.35%-10.08% of clean reads were retained. For flower eDNA sample, 1.44 million raw read pairs were obtained and 61.15% of them were retained.

3.2 Application of integrated pipeline

The integrated pipeline was used to profile metagenomic datasets from pollination system, illustrating its capacity in presenting taxon composition and functional potentiality of microbiome. Although samples are different in proportion of host contamination (Table S2, Figure 2a), diverse communities composed of multiple taxonomic clades were identified (Figure 2b), and most species identified are common in pollination system.

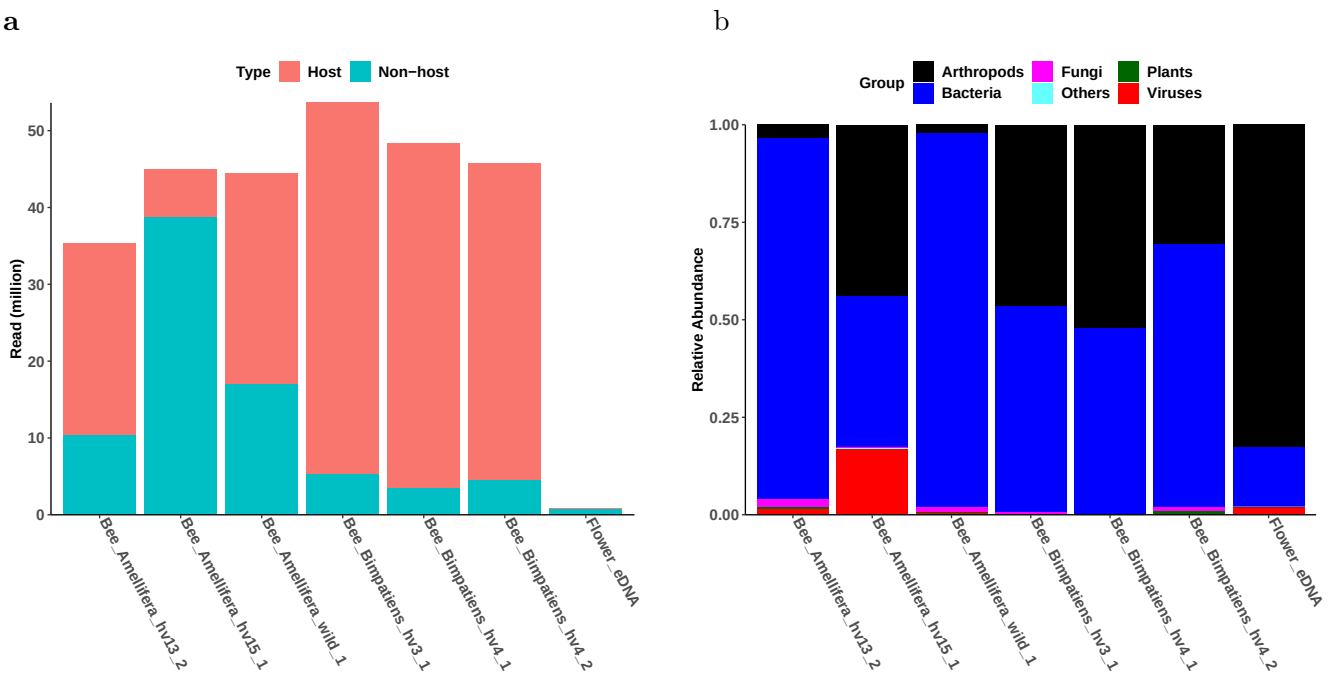

Figure 2: a. The number of host and non-host reads in each sample. b. The relative abundance of species under six taxonomic groups: superkingdom Viruses, superkingdom Bacteria, kingdom Viridiplantae (plants), kingdom Fungi, phylum Arthropoda and others (species that are not in the other five groups).

The present of core members of bee-associated bacterial community including species within *Bifidobacterium*, *Frischella*, *Gilliamella*, *Snodgrassella*, *Lactobacillus*, *Apilactobacillus* and *Bombilactobacillus* (Figure 3) (Koch and Schmid-Hempel, 2011a, Moran, 2015, Kwong et al., 2017, Zheng et al., 2020) suggests good sample quality. As for other abundant bacteria, *Fructobacillus sp.* are often found in fructose-rich environments like flowers (Endo and Dicks, 2014); *Bartonella apis* is related to animal pathogens (Kešnerová et al., 2016) and is widespread in honey bee workers (Raymann and Moran, 2018); *Candidatus Schmidhempelia bombi* is a known uncultured symbiont of *Bombus impatiens* (Martinson et al., 2014). It should be noted that some typical bee-associated bacteria were also found on the flower, including *Bartonella apis*, *Bifidobacterium asteroides*, *Bombilactobacillus mellis* and *Gilliamella apicola*.

Composition of arthropods and plants indicating interactions within pollination networks. Most arthropods identified are pollinators within *Apis* and *Bombus* (Figure S2). However, some of them might be considered as false positive. For example, *Apis cerana*, *Apis dorsata* and *Apis florea* are mainly found in Asia and unlikely to present in the area where samples were collected. These might derive by similarity between genomes of *Apis mellifera* and other *Apis* species. As for plant species (Figure S3), they indicate foraging areas of bees. Several crop species were identified, including *Brassica napus* (rape), *Brassica oleracea*, *Cicer arietinum* (chickpea), *Glycine max* (soybean), *Helianthus annuus* (sunflower), *Nicotiana sylvestris* (flowering tabacco) and *Raphanus sativus* (radish).

Fungal and viruse species were identified. In fungal communities of most samples, *Nosema ceranae*, a widespread bee pathogen, was the dominant species. (Figure S4). Besides, three yeast species (*Clavispora lusitaniae*, *Saprochaete ingens* and *Wickerhamiella sorbophila*) were also found in bees. As for viruses, most of them are phages or arthropod-infecting species (Figure S5). Phage species include *Bifidobacterium phage BitterVaud1* infecting bee-commensal bacterium *Bifidobacterium asteroides* (Bonilla-Rosso et al., 2020); *Bacteriophage sp.* infecting *Pseudomonas aeruginosa* (Essoh et al., 2015), an opportunistic pathogen that might contaminate bees (Bailey, 1968, Papadopoulou-Karabela et al., 1992, 1993); and unclassified species within Myoviridae and Siphoviridae. Listed arthropod-infecting viruses including *Apis mellifera filamentous virus* and *Bombus cryptarum densovirus* that infect bees; and several parvoviruses (*Blattodean pefuambidensovirus 1*, *Hemipteran scindoambidensovirus 1*, *Hymenopteran scindoambidensovirus 1* and *Orthopteran scindoambidensovirus 1*) (Pénzes et al., 2020).

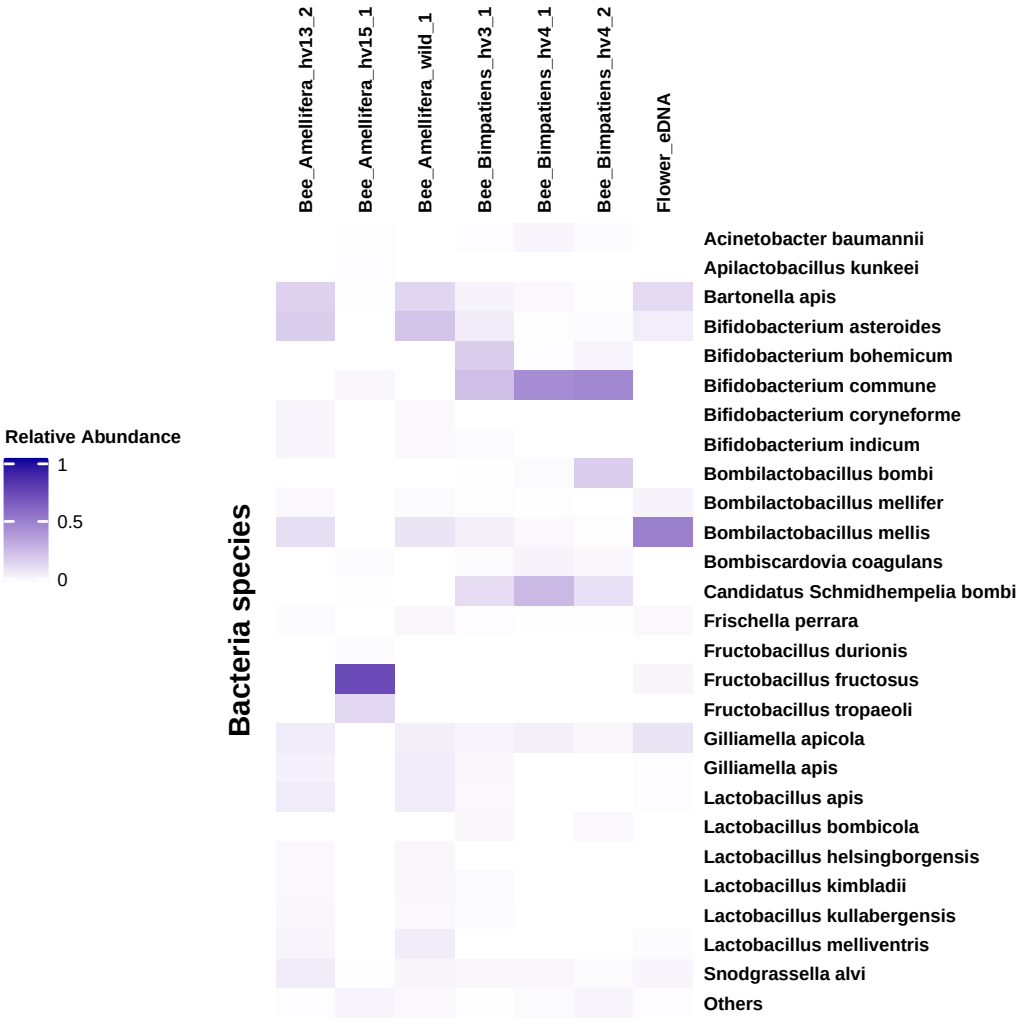

Figure 3: Heatmaps for bacterial species abundance distribution in all samples. The relative abundance takes reads assigned to bacterial species as background. Species with relative abundance smaller than 1% in all samples are collapsed as "others".

The integrated pipeline also provides information on FGC content, which shows function potentiality of metagenome. Here FGC content was represented by identified KOs, which were used for KEGG pathway inference in order to illustrate metabolic potentially of metagenomic samples. Reads assigned to plants and arthropods were not involved in pathway inference since they are unlikely to represent living organisms. The coverage of a pathway was calculated by the ratio between number of annotated KOs and total number of KOs involved in that pathway.

Here concern is given to metabolism pathways of carbonhydrates and amino acids, which are crucial for bee health. Inferred pathways indicate potential capability of metabolism of sugars including fructose, sucrose, mannose and galactose (Figure S6), and all ten essential amino acids for honey bees (*i.e.* arginine, histidine, isoleucine, leucine, lysine, methionine, phenylalanine, threonine, tryptophan and valine) (Figure S7) (Groot, 1953).

3.3 Evaluation of performance of integrated pipeline in species identification

The performance of integrated pipeline in species identification was evaluated by comparing it with assembly-dependent method, using rarefied datasets. In integrated pipeline (Figure 1), clean non-host reads are first assembled to into contigs and assigned to taxa. Then a reference database composed of genomes of assembly-represented species is constructed. Reads not aligned to the reference database are subjected to assembly-independent taxon search. The reference database and assembly-independent search helped improve species identification in all three sample types, especially in simulations of low sequencing depth (Figure 4).

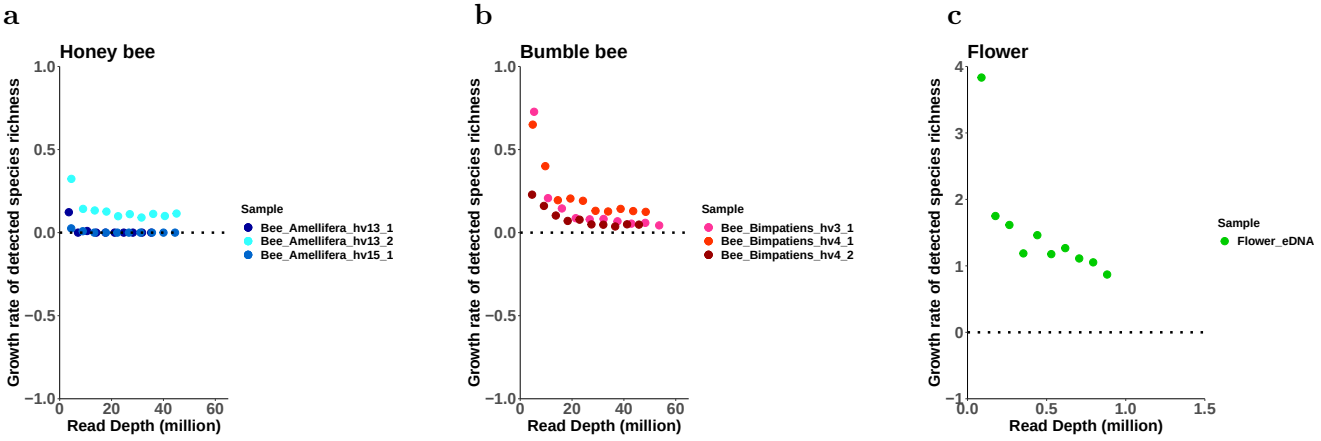

Figure 4: Integrated pipeline improves the detection of species richness. The horizonal axis represents sequencing depth, and the vertical axis represents growth rate of detected species richness comparing the integrated pipeline and assembly-dependent species identification. Different sequencing depth was simulated by rarefaction. Sample type is shown in the top left of each subfigure.

3.4 Optimal sequencing depth required for detection of species and function diversity

In order to determine the influence of sequencing depth and thus optimize sequencing depth for analysing taxonomic and functional diversity, rarefactiona anlysis was conducted. Different sequencing depth was simulated by randomly subsampling original datasets at proportions from 10% to 90% at an interval of 10%. The relationship between sequencing depth (clean read pair number) and species/KO diversity (Hill numbers of order 0, 1, 2) was quantified by fitting and averaging asymptotic species accumulation models. The slope of the model reflects the increase rate of diversity. The point at which it drops to a cut-off value provides an estimation of optimal sequencing depth. Besides, the asymptote of the model as sequencing depth tends to infinity provides an estimation of total diversity.

Rarefaction assumes that the original dataset provides an almost complete inventory, which can be verified by final slope of rarefaction curve for Hill number of order 0 (richness). Figure 5 shows rarefaction curves for species/KO richness and Table 1 summarizes their final slopes. For species diversity rarefaction, all bumble bee samples are sufficient, with final slopes < 0.1 and completeness (ratio between final richness and asymptote) > 0.98. As for honey bees, Bee_Amellifera_hv15_1 and *Bee_Amellifera_wild_1* are sufficient, while *Bee_Amellifera_hv13_2* is insufficient, with final slope > 1 and completeness < 0.8. For the flower eDNA sample, the final slope of species richness rarefaction curve is 10.8380 and its completeness is 1.32%, indicating more sequencing effort is needed for species profiling. As for function diversity rarefaction, the final slopes of all KO richness rarefaction curves are higher than 15, indicating no dataset can providing an almost complete inventory of KOs. Thus, estimation of optimal sequencing depth was conducted for the combinations of two sample types (honey bee and bumble bee) and one study type (species profiling), based on five datasets (two honey bees and three bumble bees).

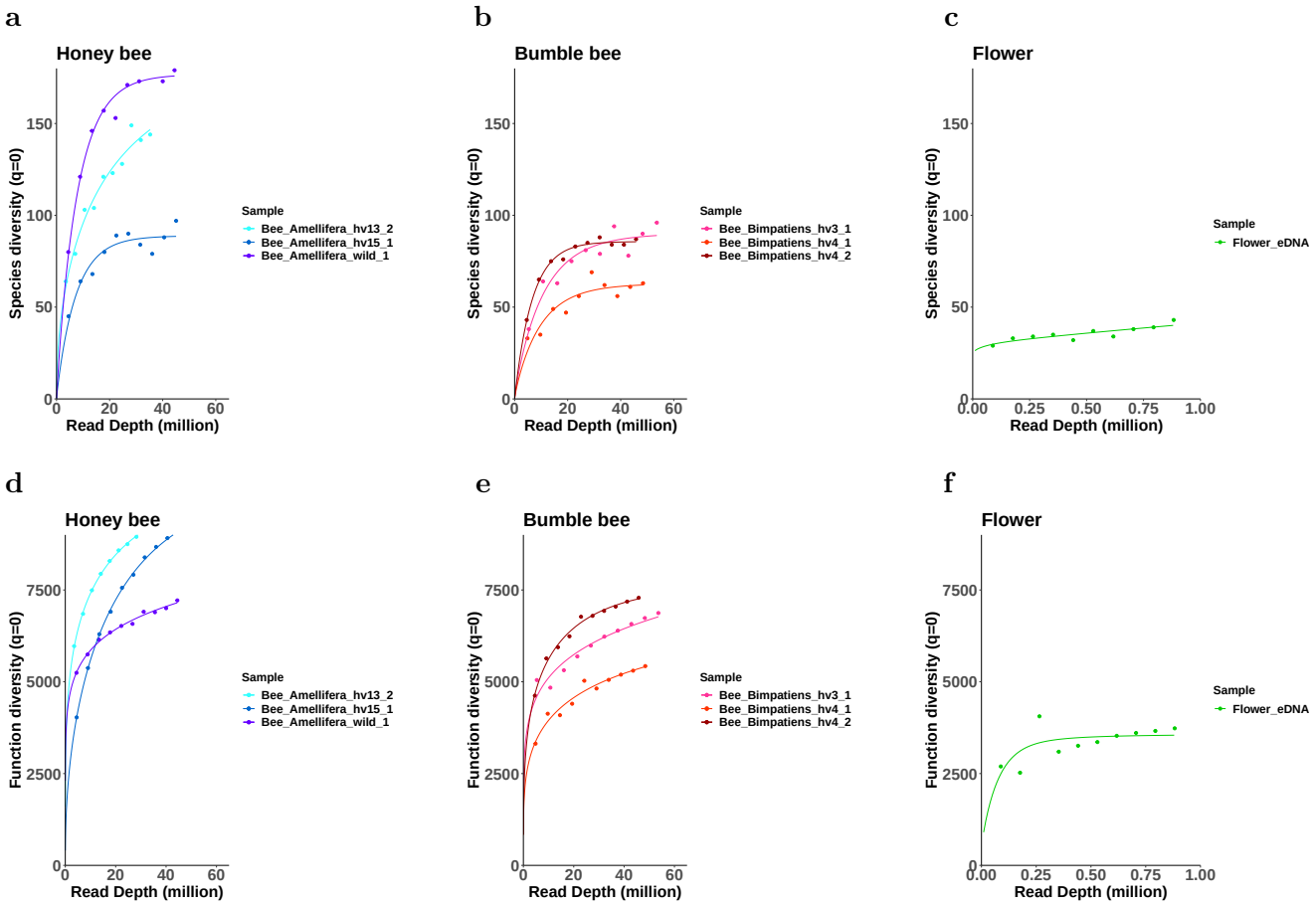

Figure 5: Rarefaction curves for species (a, b, c) or KO (d, e, f) richness (Hill number of order 0). The horizonal axis represents sequencing depth, and the vertical axis represents richness. Sample type is shown in the top left of each subfigure. Note that the scale of horizonal axis in subfigure c and f is much smaller than that in other subfigures.

Table 1: Summary of final point of rarefaction curve for species/KO richness (Hill number of order 0). Type: indicates whether this row reports rarefaction curve for species or KO richness. Depth: sequencing depth taking million read pairs as unit. OR: observed species/KO richness. ER: expected species/KO richness predicted by modeling rarefaction curve. FS: final slope of rarefaction curve. Asym: asymptote calculated by modeling rarefaction curve. Comp: completeness, represented by ratio between ER and Asym.

Sample	Type	Depth	OR	ER	FS	Asym	Comp
Bee_Amellifera_hv13_2	species	35.28	144	146.69	1.0958	184.94	0.7931
Bee_Amellifera_hv15_1	species	45.05	97	88.53	0.0344	88.84	0.9966
Bee_Amellifera_wild_1	species	44.46	179	175.89	0.0991	176.84	0.9946
Bee_Bimpatiens_hv3_1	species	53.61	96	89.00	0.0897	90.79	0.9803
Bee_Bimpatiens_hv4_1	species	48.42	63	62.06	0.0626	62.92	0.9862
Bee_Bimpatiens_hv4_2	species	45.80	87	85.59	0.0146	85.69	0.9988
Flower_eDNA	species	0.88	43	40.04	10.8380	3034.88	0.0132
Bee_Amellifera_hv13_2	KO	35.28	9289	9284.45	38.6844	10641.27	0.8725
Bee_Amellifera_hv15_1	KO	45.05	9046	9102.78	43.8717	10453.98	0.8707
Bee_Amellifera_wild_1	KO	44.46	7216	7151.72	21.4368	14779.89	0.4839
Bee_Bimpatiens_hv3_1	KO	53.61	6872	6769.80	21.1588	28111.79	0.2408
Bee_Bimpatiens_hv4_1	KO	48.42	5427	5410.71	19.6932	7665.58	0.7058
Bee_Bimpatiens_hv4_2	KO	45.8	7288	7259.56	15.6309	7873.64	0.9220
Flower_eDNA	KO	0.88	3732	3544.54	60.4367	3571.11	0.9926

Optimal sequencing depth is estimated by the point at which the slope of rarefaction curve drops to a cut-off value. Table 2 summarizes estimations of optimal sequencing depth for detection of species diversity, using 0.5, 0.1, 0.05 and 0.01 as cut-off values. When order q of Hill number equals 0, *i.e.* species abundances are not considered, cut-off value of 0.1 for slope of rarefaction curve is sufficient for providing completeness $> 97\%$ in all samples. The average optimal sequencing depth are 40.33 million for honey bees and 42.49 million for bumble bees. When species abundances are considered (order q equals 1 or 2), cut-off value of 0.01 can provide completeness $> 95\%$ in most pairs of sample and q value. For honey bees, the average optimal sequencing depth are 18.57 million ($q = 1$) and 17.45 million ($q = 2$). For bumble bee samples, the average optimal sequencing depth are 40.33 million ($q = 1$) and 24.77 million ($q = 2$).

Table 2: Summary of minimal sequencing depth estimated from different cut-off values of slope. Minimal sequencing depth was estimated by the point at which the slope of rarefaction curve drops to a cut-off value (0.5, 0.1, 0.05 or 0.01), which is indicated in names of columns. For example, MinD_0.5 represents estimated minimal sequencing depth taking 0.5 as cut-off value, and ED_0.5 is expected Hill number from MinD_0.5. Comp_0.5 is the ratio between OptDiv_0.5 and the asymptote (represented by Asym). Unit of sequencing depth is million read pairs. q refers to order of Hill number determining sensitivity to species abundance distribution.

Sample	q	Asym	MinD_0.5	ED_0.5	Comp_0.5	MinD_0.1	ED_0.1	Comp_0.1	MinD_0.05	ED_0.05	Comp_0.05	MinD_0.01	ED_0.01	Comp_0.01
Bee_Amellifera_hv15_1	0	88.84	23.54	84.85	0.9551	36.27	87.99	0.9905	41.95	88.40	0.9951	55.75	88.74	0.9989
Bee_Amellifera_wild_1	0	176.84	30.53	172.46	0.9752	44.39	175.88	0.9946	50.57	176.33	0.9971	65.86	176.70	0.9992
Bee_Amellifera_hv15_1	1	12.28	3.18	10.67	0.8699	5.83	11.33	0.9224	7.21	11.43	0.9304	15.21	11.58	0.9429
Bee_Amellifera_wild_1	1	14.77	0.69	13.49	0.9137	3.30	14.03	0.9498	6.20	14.23	0.9635	21.93	14.58	0.9868
Bee_Amellifera_hv15_1	2	8.40	3.10	6.35	0.7561	8.17	7.49	0.8909	11.97	7.75	0.9229	27.99	8.11	0.9655
Bee_Amellifera_wild_1	2	9.88	0.18	9.49	0.9602	0.90	9.63	0.9747	1.73	9.69	0.9805	6.91	9.80	0.9916
Bee_Bimpatiens_hv3_1	0	90.79	30.80	83.69	0.9218	52.03	88.85	0.9786	62.56	89.61	0.9870	92.35	90.32	0.9948
Bee_Bimpatiens_hv4_1	0	62.92	24.83	57.22	0.9093	42.76	61.61	0.9791	51.24	62.22	0.9887	73.55	62.76	0.9973
Bee_Bimpatiens_hv4_2	0	85.69	21.75	82.30	0.9604	32.67	85.01	0.9920	37.39	85.35	0.9960	48.40	85.62	0.9992
Bee_Bimpatiens_hv3_1	1	21.59	3.63	10.50	0.4862	9.47	11.76	0.5449	15.37	12.18	0.5639	58.09	13.04	0.6040
Bee_Bimpatiens_hv4_1	1	11.85	3.80	9.45	0.7973	10.15	10.87	0.9174	14.68	11.19	0.9445	31.22	11.57	0.9766
Bee_Bimpatiens_hv4_2	1	12.66	4.17	10.24	0.8087	10.47	11.67	0.9213	14.85	11.98	0.9458	31.68	12.36	0.9760
Bee_Bimpatiens_hv3_1	2	8.73	2.20	6.16	0.7051	6.28	7.06	0.8088	9.45	7.28	0.8344	23.89	7.60	0.8708
Bee_Bimpatiens_hv4_1	2	8.33	3.12	6.28	0.7540	8.52	7.50	0.9002	12.30	7.76	0.9323	26.68	8.09	0.9718
Bee_Bimpatiens_hv4_2	2	7.12	2.90	5.34	0.7493	7.47	6.38	0.8961	10.58	6.60	0.9270	23.73	6.90	0.9683

4 Discussion

Here, I constructed an integrated pipeline combining assembly-dependent and -independent methods for taxonomic and functional profiling of shotgun metagenomics, and applied it to analysis of metagenomes from honey bees, bumble bees and flower washes. The profiling results showed that the integrated pipeline is able to capture taxonomic diversity by identifying species from multiple clades and infer metabolic potentiality of metagenome by providing information on FGC content. It can be more sensitive in species identification compared with standard assembly-dependent methods. Then I computed expected species/FGC diversity represented by given sequencing depth through rarefaction analysis, in order to optimize sequencing depth to balance cost and reliability of analysis results. Optimal sequencing depth differs by types of samples and investigations. For estimation of species diversity, sequencing depth can be optimized especially when low leverage is given to rare species. As for functional profiling, deeper sequencing depth is required.

The integrated pipeline (Figure 1) for taxonomic and functional profiling of metagenome provides several advantages. First, through combination with assembly-free taxon search, it helps solve high false negative rate associated with assembly-dependent species identification, which is caused by reads left unassembled. This was shown by analyzing real metagenomic datasets from pollination system and simulating differ-

ent sequencing depth by rarefaction (Figure 4). Second, the modularity of integrated pipeline provides flexibility for incorporation of alternative tools. For example, BWA aligner (Li and Durbin, 2009) serves as an alternative for Bowtie2, and SPAdes can be replaced by other metagenomic assemblers such as Megahit (Li et al., 2015) and IDBA-UD (Peng et al., 2012). Third, the output files generated by each step are recorded and can be inspected easily, which provides transparence for troubleshooting. However, the performance of integrated pipeline need to be evaluated more comprehensively by analysis of mock datasets and comparison with other profiling strategies. Standardized mock dataset can be used to benchmark sensitivity and accuracy of integrated pipeline since it is an artificial metagenome with predefined diversity, generated by combining known sequences from different species (Vollmers et al., 2017, **?**, Becker et al., 2020). Besides, the performance of integrated pipeline need to be compared with other widely used analysis strategies, such as MG-RAST (Meyer et al., 2008), SqueezeMeta (Tamames and Puente-Sánchez, 2019) and Kraken (Wood and Salzberg, 2014).

The integrated pipeline was used to analyze metagenomes of bees and flower washes, illustrating their taxon composition and metabolic potentiality. Bee-associated microorganisms were identified in flower washes, including both pathogens (*e.g. Nosema ceranae* and *Apis mellifera filamentous virus*) and symbionts (*e.g. Bifidobacterium asteroides*, *Bombilactobacillus mellis* and *Gilliamella apicola*) (Figure 3, S4 and S5). Flowers have been indicated as hubs for transmission of bee pathogens Durrer and Schmid-Hempel (1994), Koch et al. (2017). Through shared flower use, multiple pathogens transmit between pollinators, including *Nosema ceranae* (Fürst et al., 2014) and *Crithidia bombi* (Figueroa et al., 2019). There is also growing evidence for present of bee symbionts on flower (McFREDERICK et al., 2012, McFrederick et al., 2017, Keller et al., 2020, Vannette, 2020). However, its role in bee microbiome assembly and functional importance in pollination remains an open question (Keller et al., 2020, Vannette, 2020). As for function profiling, it is indicated that bee-associated microbiome is capable of metabolizing carbohydrates such as glucose, fructose, sucrose and mannose (Figure S6). Carbohydrates are main component of bee diet and microbiome-mediated carbohydrate-processing have been vastly investigated (Engel et al., 2012, Lee et al., 2015, 2018, Taylor et al., 2019). Besides, pathways for metabolism of all ten essential amino acids for honey bees (Groot, 1953) were inferred (Figure S7). Essential amino acids are crucial for bee health (Simcock et al., 2014, Paoli et al., 2014, Stabler et al., 2015, Hendriksma et al., 2019) and influence feeding preference due to their potential deficiency in single pollen source (Cook et al., 2003, Hendriksma et al., 2014, Hendriksma and Shafir, 2016). Whether bee-associated microbiome influences host health by providing amino acids need to be further investigated.

Optimization of sequencing depth is important for shotgun metagenomics since insufficient sequencing causes underestimation of taxonomic/functional diversity (Cattonaro et al., 2018, Zaheer et al., 2018,

Gweon et al., 2019, Pereira-Marques et al., 2019), while deep sequencing is of high cost. Here, expected species/KO diversity provided by given sequencing depth was estimated by rarefaction and model fitting, using datasets from honey bees, bumble bees and flower washes. It was shown that increasing sequencing depth boosted identification of species/KO (Figure 5, Table 1), highlighting value of deep metagenomic sequencing. For function profiling, no dataset involved here is big enough to provide an almost complete inventory of KOs (Table 1) even though most samples were deeply sequenced (Table S1), indicating such task is demanding about sequencing depth. Therefore, when dealing with function potentiality of microbiome associated with bees or flowers, retrieving as many reads as possible would be recommended. As for species profiling, although deep sequencing is still valuable, sequencing depth can be optimized when the budget is limited (Table 2), especially when low leverage is given to rare species. For honey bees, approximate 40 million sequencing depth (12 Gbp) can be sufficient for representing species richness. When assessing biodiversity with reduced emphasis on rare species, 17-19 million sequencing depth (5.1-5.7 Gbp) can provide robust estimation. As for bumble bees, about 40-43 million sequencing depth (12-12.9 Gbp) can provide reliable estimation for species richness and diversity index with emphasis on species of general abundance, and about 25 (7.5 Gbp) million sequencing depth can be sufficient for estimation of biodiversity with high leverage on abundant species. It should be noted that the efficacy of optimal sequencing depth estimation is reduced by limited number of samples. Here, honey bee and bumble bee associated microbiome was represented by only three samples, and there was only one sample of flower eDNA. However, microbiomes of pollination system are highly dynamic and variable in diversity. To generate reliable guideline for sequencing depth optimization, more samples need to be evolved. Besides, there is a lack of repeat in sequencing depth subsampling. Repeated subsampling boosts precise computation of rarefaction curve and increases accuracy of estimation of optimal sequencing depth.

5 Conclusion

Shotgun metagenomics is capable of illustrating diversity of multiple taxonomic clades and gene content, and thus provides unique advantages over vastly used amplicon sequencing, particularly for investigations of highly diverse microbial communities. However, utilization of shotgun metagenomics is hindered by challenges in data analysis and high cost of sequencing. Here, I constructed an integrated pipeline for analysis of shotgun metagenomic data. It provides benefits in terms of results, flexibility and transparence. I also constructed a framework for optimizing depth of shotgun metagenomic sequencing in order to balance high cost of sequencing and reliability of analysis results. The pipeline and the framework were used for analysis of real datasets from pollination system, providing guidelines for cost-effective metagenomic investigations of bee- and flower-associated microbiomes.

6 Data and Code Availability

Scripts used for the analyses are available at github. Metagenomic datasets involved in this project cannot be made available publicly since they are yet to be formally published.

7 Supplementary

7.1 Parameter settings of integrated pipeline

7.1.1 Quality control and host removing

Raw data quality control was conducted to reduce compromise from low quality reads. Raw data quality was checked using FastQC v.0.11.5 (Andrews et al., 2010) before filtering. FastQC reports of raw reads showed the following aspects need to be covered in quality control: (1) low base quality in 3'-end (Figure S1a); (2) uneven base content in 5'-end (Figure S1c) and (3) the present of Nextera adaptors (Figure S1e). Therefore, raw reads were filtered using Trimmomatic v.0.39 (Bolger et al., 2014), which (1) trimmed adaptors; (2) cutted 15 bases from the 5'-ends of reads; (3) cutted bases off from 3'-ends of reads if Phred-33 quality is below 20; (4) dropped reads shorter than 50 bp; (5) dropped reads if average Phred-33 quality is below 20. Then unpaired reads were removed and quality of clean data was checked using FastQC (Figure S1d, S1e, S1f).

After quality control, host read pairs were removed. First, clean read pairs were mapped to host genome (GCA_003254395.2 for *Apis mellifera* and GCA_000188095 for *Bombus impatiens*, downloaded from NCBI) using Bowtie2 v.2.4.2 (Langmead and Salzberg, 2012) with flags *–end-to-end* and *–sensitive*. With flag *–end-to-end*, Bowtie2 requires the read aligned without any clipping from neither end, and *–sensitive* maintains a trade-off between speed and sensitivity. SAM files generated by Bowtie2 were converted to BAM format using SAMtools v.1.11 (Li et al., 2009). Then non-host read pairs were extracted from BAM files by SAMtools according to the present of SAM flag 12 (neither forward nor reverse read in a pair of reads is mapped).

7.1.2 Assembly-dependent species identification

In order to identify taxa of metagenome, *de novo* assembly was conducted. Non-host read pairs were assembled using SPAdes v.3.15.2 (Prjibelski et al., 2020). Values of k-mer ranged from 21 to 101 at interval of 10. Flags *–only-assembler* and *–meta* were used. Through flag *–meta*, SPAdes runs metaSPAdes which is developed for metagenomic assembly (Nurk et al., 2017). The *–only-assembler* flag skips read error correction and runs assembly only. Its utilization is justified by the following facts. First, when *–only-*

assembler is not used, SPAdes conducts error correction before assembly. It is conducted by BayesHamme, which is optimized for single cell sequencing instead of shotgun metagenomics (Nikolenko et al., 2013). Besides, reads used for *de novo* assembly had been filtered to ensure their quality.

After assembly, taxon identification was conducted using DIAMOND v.2.0.7.145 (Buchfink et al., 2015) and MEGAN6 (Huson et al., 2007). Assembled contigs with a length above 500 bp were aligned to nr database using DIAMOND v.2.0.7.145 with *–long-reads* flag. This flag triggers frame-shift aware alignment mode, which is optimized for long sequence alignment. Therefore, short contigs (length < 500 bp) were not retained. Besides, alignments with an E-value $< 1e-5$ or identity $< 50\%$ were removed, and for each contig, only alignments above 10% of the best local bit score were retained. The output of DIAMOND was analysized by the *blast2rma* tool of MEGAN6 with *-lg* flag, which runs lowest-common-ancestor (LCA)-based algorithm developed for long contigs and assigns each contig to a taxon (Huson et al., 2018). The parameter *-supp* was 0, which means the present of a taxon would be identified as long as at least one contig was assigned. This value was used because a contig is assembled from multiple short reads and represents a strong signal for the present of a taxon.

7.1.3 Fragment recruitment

To integrate individual genomic data of species identified by assembly-dependent search, a reference database comprising reference genome dataset, *i.e.* genomic sequences in FASTA format and corresponding gff file, was constructed. For each species represented by assembly, its reference genome dataset, if available, was downloaded from NCBI using its *datasets* command-line tool and added to the reference database.

Then fragment recruitment was conducted. The non-host read pairs were mapped to genomic sequences in the reference database using Bowtie2. Read pairs that were not recruited were extracted using SAMtools. Settings for Bowtie2 and SAMtools were the same as that described in 7.1.1. Read pairs recruited by the reference database were assigned to corresponding species, while the others were subjected to assembly-independent search.

7.1.4 Assembly-independent species identification

In order to detect species not represented by assembly (Sharon et al., 2015, Vollmers et al., 2017), assembly-independent search was conducted, taking read pairs not recruited by the reference database as input. These reads were aligned to nr database through DIAMOND without using *–long-reads* flag, which triggers computing alignments for short metagenomic reads. Other settings were the same as described in 7.1.2. Then the output of DIAMOND was analysed by MEGAN6 (*blast2rma* tool), which assigns read pairs to

taxa through LCA algorithm. Here the parameter *-supp* was 0.1, which means a taxon is reported after being represented by at least 0.1% of all assigned read pairs. It was used in order to avoid false positive results.

7.1.5 Functional annotation

Functional annotation was conducted by EggNOG-mapper v.2.1.2 (Huerta-Cepas et al., 2017). Sequences were searched against eggNOG database (Huerta-Cepas et al., 2019) for best seed orthologs using DIAMOND and fine-grained orthology assignments were retrieved from pre-computed eggNOG phylogenetic trees. Then functional descriptions of retrieved orthologs including Gene Ontology (GO) terms (Consortium, 2004), KOs, Enzyme Commission (EC) numbers (Webb et al., 1992), Carbohydrate-Active Enzymes (CAZy) terms (Cantarel et al., 2009) and Clusters of Orthologous Groups (COG) functional categories (Tatusov et al., 2000) were transferred to query sequences.

7.2 Candidate models for fitting rarefaction curves

Table S1: Candidate species accumulation models. Dependent variable $D^{(q)}$ is Hill number of order q and independent variable x is sequencing depth. a, b, c, d are fitted coefficients.

Model	Parameter(k)	Derivative	Asymptote	Reference
$D^{(q)} = \frac{ax}{bx+1}$	2	$\frac{dD^q}{dx} = \frac{a}{(bx+1)^2}$	$\frac{a}{b}$	Clench (1979)
$D^{(q)} = a(1-e^{-bx})$	2	$\frac{dD^q}{dx} = abe^{-bx}$	a	Miller and Wiegert (1989)
$D^{(q)} = a - bc^x$	3	$\frac{dD^q}{dx} = -bc^x log(c)$	a	Ratkowsky (1983)
$D^{(q)} = a(1-e^{-bx})^c$	3	$\frac{dD^q}{dx} = abce^{-bx}(1-e^{-bx})^{c-1}$	a	Ratkowsky and Giles (1990)
$D^{(q)} = a(1-(1+(\frac{x}{c})^d)^{-b})$	4	$\frac{dD^q}{dx} = \frac{abd}{c}(\frac{x}{c})^{d-1}(1+(\frac{x}{c})^d)^{-b-1}$	a	Mielke Jr and Johnson (1974)

7.3 Exemplification of effect of quality control

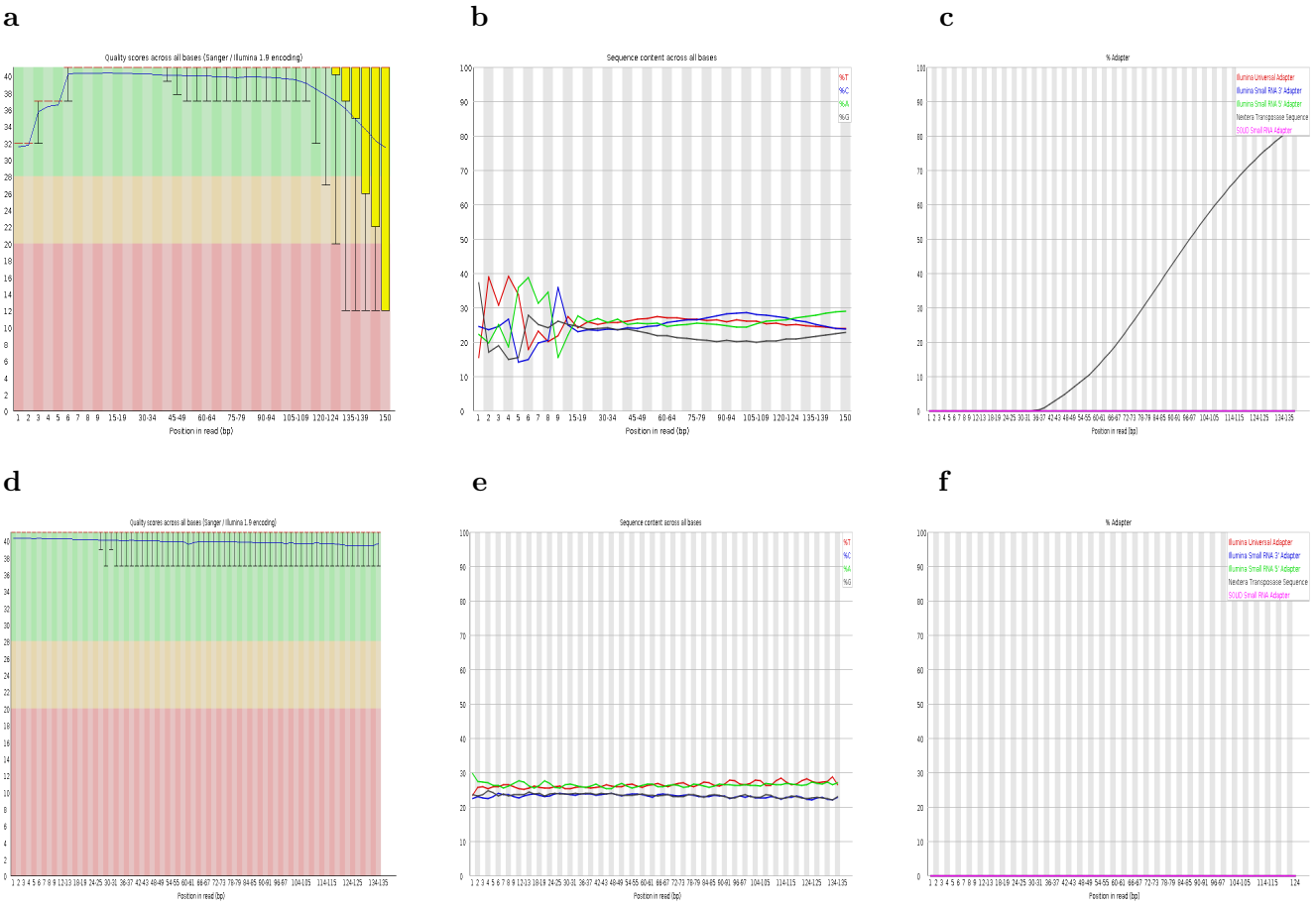

Figure S1: Data quality report of forward reads from bumble bee sample *Bee_Bimpatiens_hv4_1*. a, b and c shows low base quality in 3'-end, uneven base content in 5'-end and present of Nextera adaptors in raw data, respectively. d, e and f shows the same results from clean data.

7.4 Numbers of raw, clean and non-host read pairs

Table S2: Statistics of sequenced read pairs.

Sample	Host	Raw read pair	Clean read pair [*percentage of raw read pair*]	Non-host read pair [*percentage of clean read pair*]
Bee_Amellifera_hv15_1	*Apis mellifera*	63159968	45059211 [71.34%]	38837759 [86.19%]
Bee_Amellifera_wild_1	*Apis mellifera*	58113227	44466776 [76.52%]	17014144 [38.26%]
Bee_Amellifera_hv13_2	*Apis mellifera*	56836899	35282101 [62.08%]	10413704 [29.52%]
Bee_Amellifera_hv13_1	*Apis mellifera*	1104861	842095 [76.22%]	665507 [79.03%]
Bee_Bimpatiens_hv3_1	*Bombus impatiens*	63973750	53612702 [83.80%]	5300592 [9.89%]
Bee_Bimpatiens_hv4_1	*Bombus impatiens*	58988182	48426748 [82.10%]	3557052 [7.35%]
Bee_Bimpatiens_hv4_2	*Bombus impatiens*	54955553	45805759 [83.35%]	4618023 [10.08%]
Flower_eDNA	None	1443107	882436 [61.15%]	882436 [100%]

7.5 Species composition of arthropods, plants, fungi and viruses

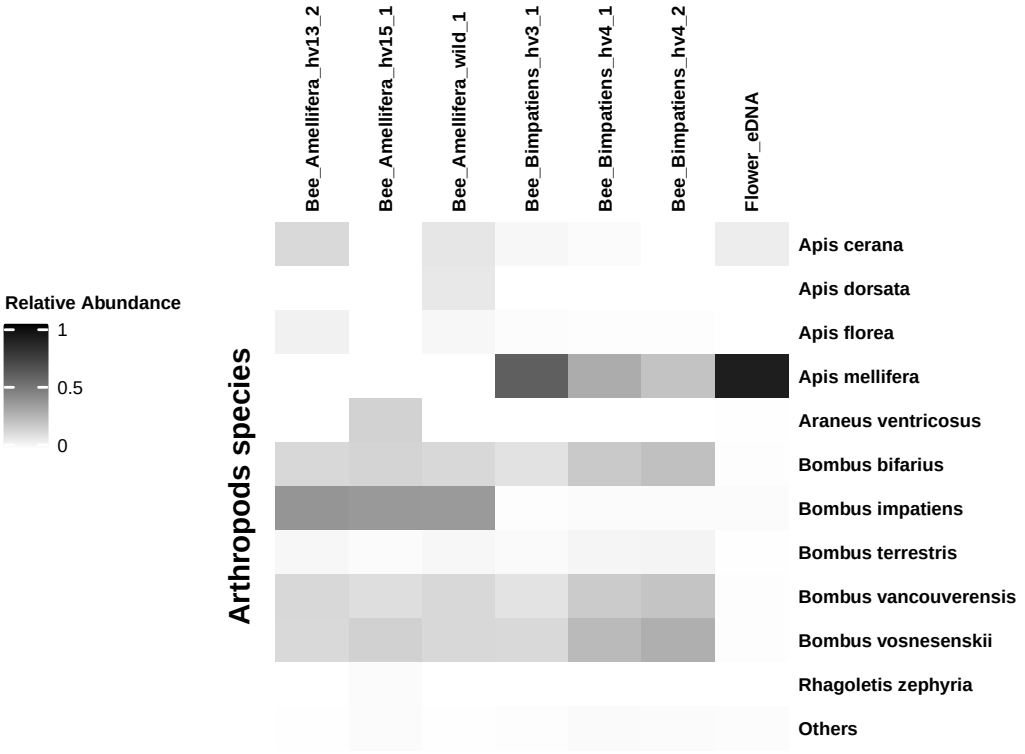

Figure S2: Heatmaps for arthropod species abundance distribution in all samples. The relative abundance takes reads assigned to arthropod species as background. Species with relative abundance smaller than 1% in all samples are collapsed as "others". It should be noted that for bee samples, host contamination was removed before taxon profiling. As a result, the relative abundances of honey bees are extremely low in three honey bee samples, and the same for bumble bees in three bumble bee samples.

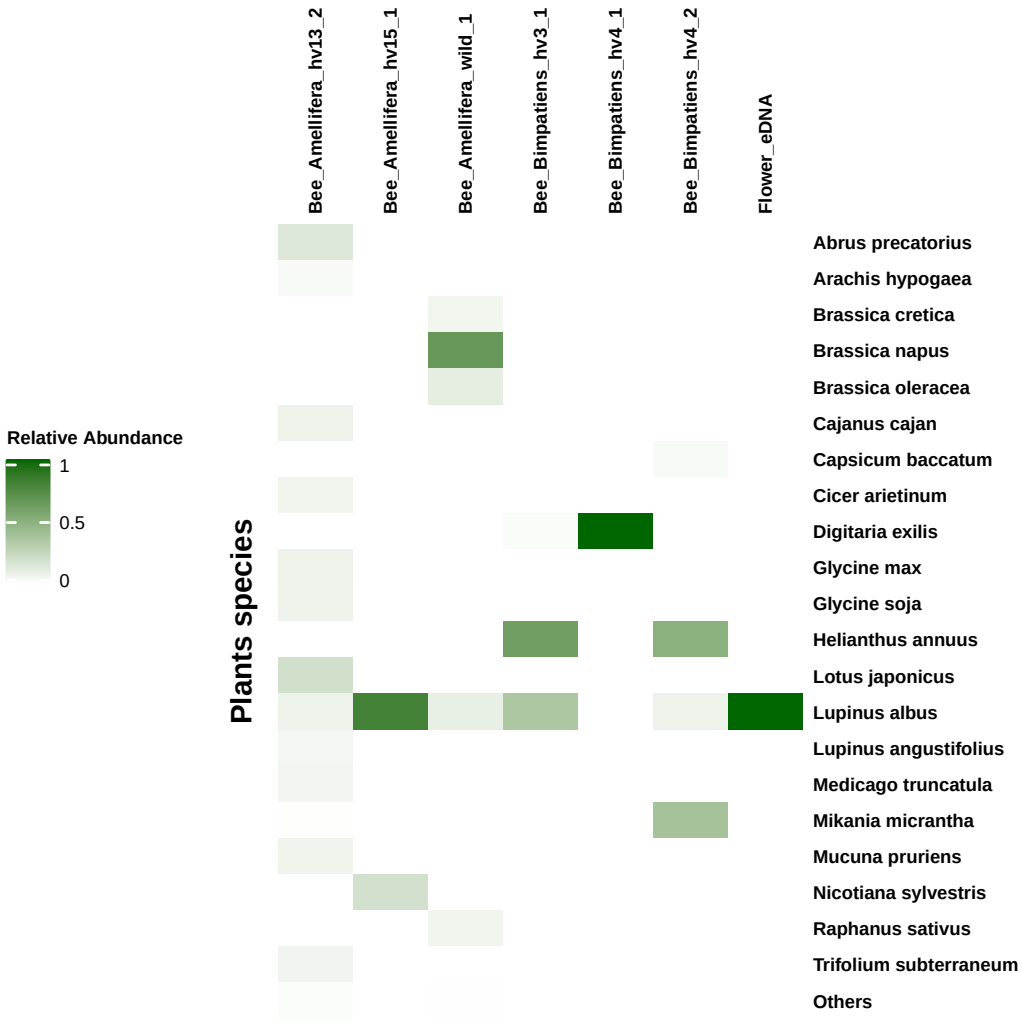

Figure S3: Heatmaps for plant species abundance distribution in all samples. The relative abundance takes reads assigned to plant species as background. Species with relative abundance smaller than 1% in all samples are collapsed as "others".

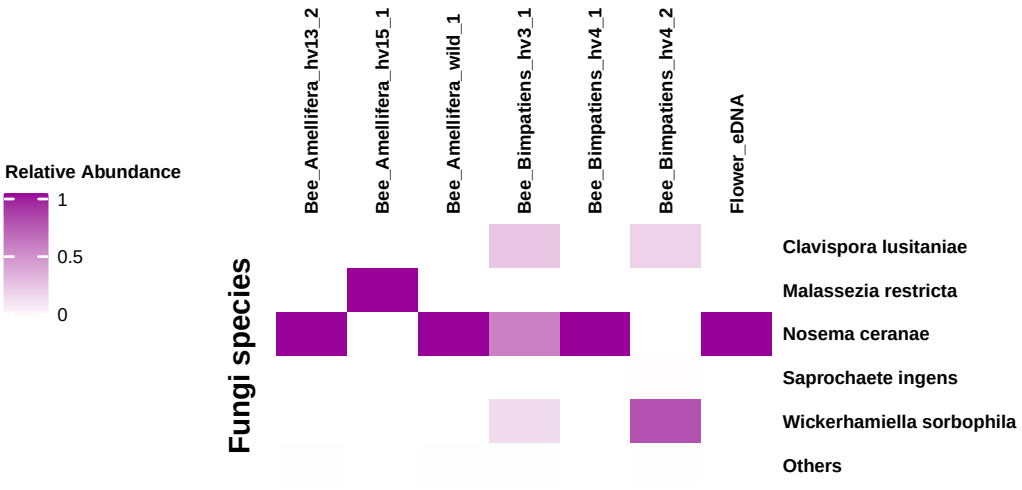

Figure S4: Heatmaps for fungal species abundance distribution in all samples. The relative abundance takes reads assigned to fungal species as background. Species with relative abundance smaller than 1% in all samples are collapsed as "others".

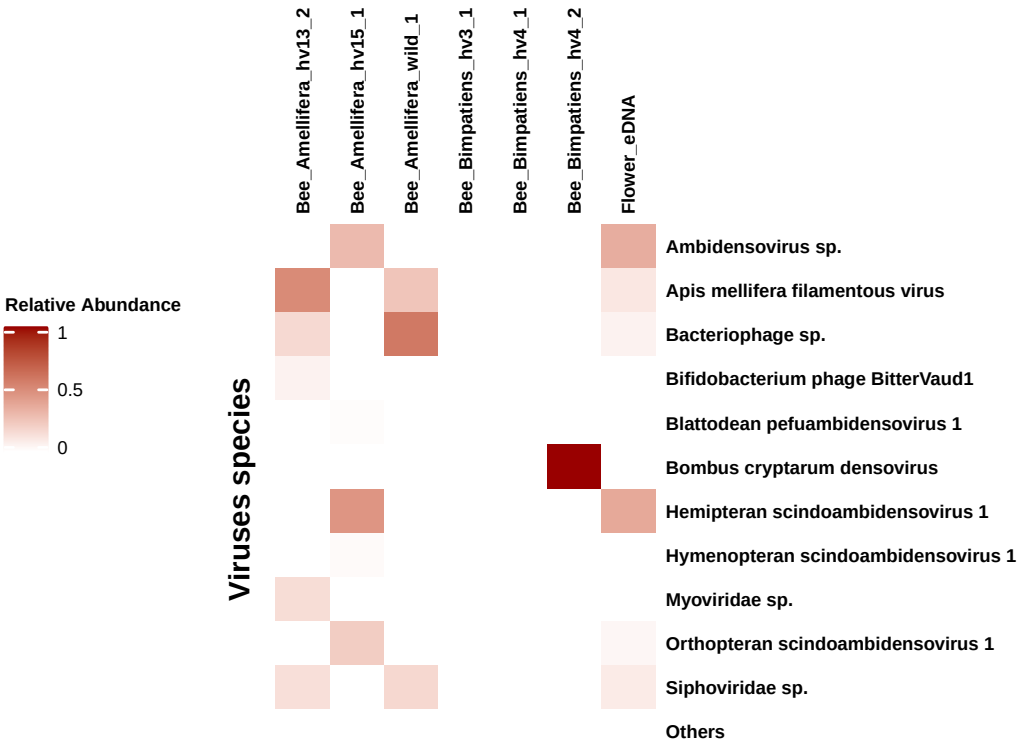

Figure S5: Heatmaps for viruse species abundance distribution in all samples. The relative abundance takes reads assigned to viruse species as background. Species with relative abundance smaller than 1% in all samples are collapsed as "others".

7.6 Inferred pathways of carbohydrate and amino acid metabolism

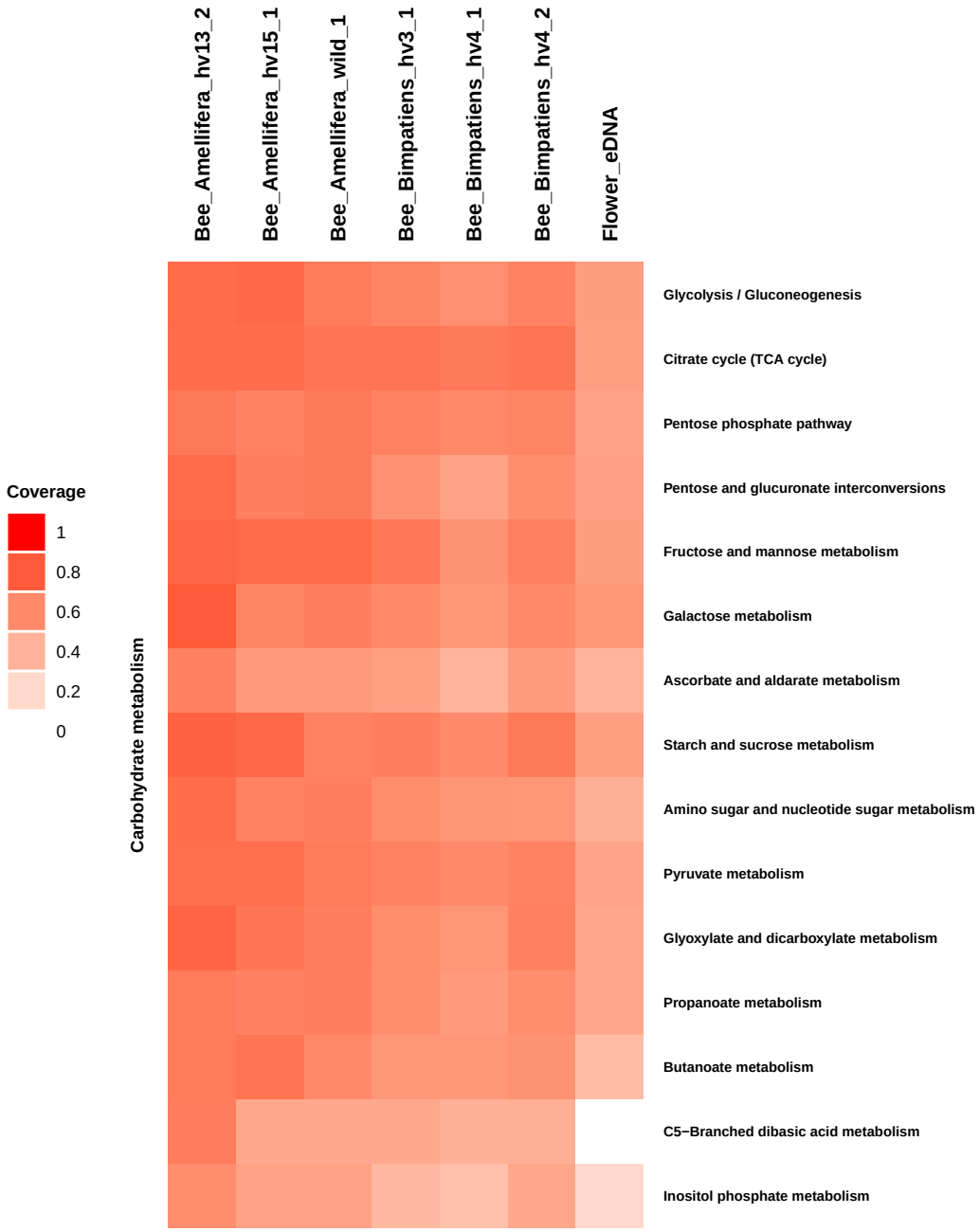

Figure S6: Heatmaps for pathways of carbohydrate metabolism.

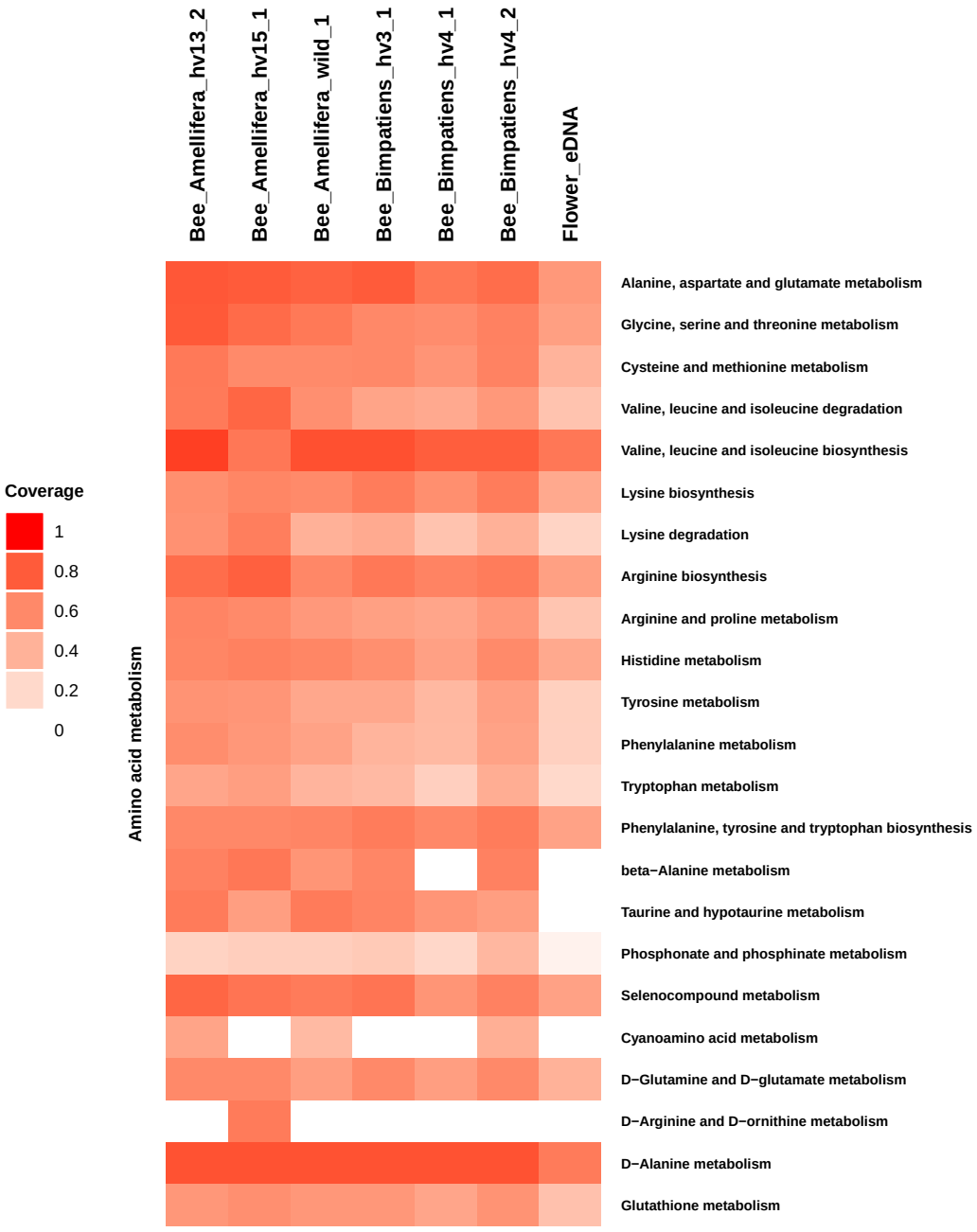

Figure S7: Heatmaps for pathways of amino acid metabolism.

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