Questioning the fetal microbiome illustrates pitfalls of low-biomass microbial studies

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Preface

Whether the human fetus and the prenatal intrauterine environment (amniotic fluid, placenta) are stably colonized by microbes in a healthy pregnancy remains the subject of a contentious scientific debate. Here, we evaluate recent studies that characterized microbial populations in human fetuses from the perspectives of reproductive biology, microbiology, bioinformatics and data science, immunology, clinical microbiology, and gnotobiology, and assess the likely mechanisms by which the fetus could interact with microbes. Our analysis indicates that the detected microbial signals are likely the result of contamination during the clinical procedures to obtain fetal samples, DNA extraction, and DNA sequencing. Further, the existence of live and replicating microbial populations in healthy fetal tissues is not compatible with fundamental concepts of immunology, clinical microbiology, and the derivation of germ-free mammals. These conclusions are not only important to our understanding of human immune development, but also illustrate common pitfalls in the microbial analyses of many other low-biomass environments. The pursuit of a “fetal microbiome” can serve as a cautionary example of the challenges of sequence-based microbiome studies when biomass is low or absent and emphasizes the critical need for a trans-disciplinary approach that goes beyond contamination controls, also incorporating biological, ecological, and mechanistic concepts.

Introduction

Fetal immune development prepares the neonate for life in a microbial world and underpins lifelong health[1-4]. Neonates born at term are not immunologically naïve and are specifically adapted to cope with abrupt exposure to microbial, dietary, and environmental stimuli and antigens[5,6]. Several research groups have characterized immune cell development in human fetal tissues[7-9]. However, our mechanistic understanding of how and when immune priming by microbes occurs, and the factors that drive it, is incomplete.

The long-held view that the prenatal intrauterine environment (placenta, amniotic fluid, fetus) is protected from live microbes has been challenged recently[10-15], leading to the hypothesis that fetal immune development may be driven by the presence of live microbes or even entire microbiomes at intrauterine sites[16-19]. However, these results have been debated[20-26] because several concurrent studies[27-33] point to experimental contamination dominating low–microbial-biomass sequencing data[34-36] as the source of microbial DNA apparently detected in the intrauterine environment. Since 2020, four studies have characterized the microbiology of the human fetus directly and resulted in opposing and irreconcilable conclusions. Two reports described viable low-density microbial populations in human fetal intestines[37] and organs[38], and linked these microbes to fetal immune development. In contrast, two other research groups, that included several of the authors of this perspective, reported no detectable microbes in fetal meconium and intestines[28,39].

Such disagreement over a fundamental aspect of human biology poses a significant challenge for scientific progress. This is not simply a matter of controversy or a reluctance to relinquish established dogma; rather, the notion of a fetal microbiome, if proven correct, has implications for clinical medicine and would call for concepts and research not previously contemplated. It would require radical revision of our understanding of the development of the immune and other

systems in early life and the anatomical and immunological mechanisms to facilitate symbiotic host-microbe interactions within fetal tissues. Failure to resolve the issue is a potential risk of diverting resources into research that ultimately results in no advancement for fetal and maternal health and misguided attempts to therapeutically modify a putative fetal microbiome. Moreover, the dilemma has immediate relevance to the characterization of all low-biomass samples.

Therefore, we assembled a trans-disciplinary group of scientists and clinician-scientists to clarify how and when the fetus becomes prepared for life with microbes, to identify research pitfalls and mitigation strategies, and to propose specific directions for future research. A diversity of research perspectives were included:(i) reproductive biology and obstetrics; (ii) microbiology and microbial ecology; (iii), bioinformatics and data science; (iv) immunology; (v) clinical microbiology; and (vi) gnotobiology and the derivation of germ-free mammals.

Claims and counterclaims

Although the disagreement on the presence of microbes in prenatal intrauterine locations (placenta and amniotic fluid) spans dozens of studies with contradictory findings[11,13,14,21,27,29-32,35,40-42], we focus our analysis on four recent studies since they provide a direct assessment of the fetus itself[28,38,39,43]. Collection of human fetal samples is difficult and restricted to either following pregnancy termination, or immediately prior to birth by C-section. Three of the studies used samples collected after vaginally delivered, elective, second trimester pregnancy terminations[38,39,43], and one collected samples from breech C-section deliveries immediately at birth[28].

Rackaityte *et al*.[43] reported 18 bacterial taxa as enriched in intestinal contents of vaginally delivered fetuses from 2nd trimester terminations compared to negative controls using 16S rRNA

gene amplicon sequencing (V4 region). To account for contamination, the authors removed Operational Taxonomic Units (OTUs) detected in >50% of procedural controls and then identified remaining contaminants *in silico* (using the decontam R package). They found that most fetal samples were microbiologically similar to negative controls (labelled as "other meconium", n=25), but that some samples, dominated by *Lactobacillus* (6 samples) or *Micrococcaceae* (9 samples), had distinct bacterial profiles. The authors further detected low amounts of total bacteria by qPCR, Fluorescent *in situ* hybridization (FISH), Scanning Electron Microscopy (SEM), and culture (as discussed below).

Several of the study's conclusions have been challenged by de Goffau *et al.*[44], who re-analyzed the publicly available data and found no evidence for a distinct bacterial profile in the subset of samples with matched procedural controls, and concluded that the positive findings were caused by a sequencing batch effect and contamination during culture[44]. In addition, the authors' suggestion that particles detected in SEM micrographs constitute micrococci[43] was disputed as their size exceeded that of known *Micrococcaceae*[44]. Furthermore, the 16S rRNA gene sequence of the *Micrococcus luteus* cultured from the fetal samples differed from that detected by sequencing, suggesting contamination during culture (*Micrococcus luteus* is a common contaminant of clean rooms and surgical instruments[45,46]).

Mishra *et al.*[38] detected a low but consistent microbial signal across tissues of vaginally delivered fetuses from 2nd trimester terminations by 16S rRNA gene amplicon sequencing (V4-V5 region), with 7 genera enriched in fetal samples (*Lactobacillus*, *Staphylococcus*, *Pseudomonas*, *Flavobacterium*, *Afipia*, *Bradyrhizobium*, and *Brevundimonas*). The 16S rRNA gene sequencing data were accompanied by SEM, RNA-*in situ* hybridization (RNA-ISH), and culture. In recognition of the high risk of contamination, all samples were processed in isolation with negative controls collected during sample processing. In contrast to Rackaityte *et al*., Mishra *et al*. found

198 *Micrococcus* to be enriched in phosphate buffered saline (PBS) reagent controls and reported it
199 as a contaminant, with the *M. luteus* cells detected by culture being consistent with the size and
200 morphology of the coccoid structures found by SEM[38].
201
202 Both the studies by Rackaityte *et al*. and Mishra *et al*. included assays to study immune
203 development of the fetus and concluded that the microbes detected would contribute to immune
204 maturation. Rackaityte *et al*.[43] based this conclusion on differences in patterns of T cell
205 composition and epithelial transcription between fetal intestines determined by whether
206 *Micrococcaceae* were or were not the dominant species and suggested that bacterial antigens
207 may contribute to T cell activation and immunological memory *in utero*. Mishra *et al*.[38] employed
208 flow cytometry to expand on previous findings of effector (TNF-α /IFN-γ producing) memory
209 (CD45RO+) T cells in fetal tissues, including gut tissue and mesenteric lymph nodes. Bacterial
210 isolates cultured from the fetal samples, including *Staphylococcus* and *Lactobacillus* strains,
211 induced *in vitro* activation of memory T cells isolated from fetal mesenteric lymph nodes.
212
213 In contrast to these reports, Li *et al.*[39], who also investigated fetal intestinal tissue from second
214 trimester terminations, did not detect bacterial DNA by PCR (V4 region of the 16S rRNA gene, 35
215 cycles) based on visual inspection of agarose gels in any of the 101 samples tested. The authors
216 detected a diverse set of metabolites in fetal intestinal samples and hypothesized that maternal,
217 microbiota-derived metabolites may pass through the placenta to 'educate' the fetal immune
218 system. This conclusion is supported by research in mice that showed that fetal immune
219 education can be driven in the absence of direct microbial exposure by trans-placental passage
220 of microbial metabolites from the maternal gut[47,48].
221
222 Kennedy *et al.*[28] used a different approach and collected samples using rectal swabs during
223 elective C-section for breech presentation at term gestation[28]. Comparisons with environmental

and reagent-negative controls from two independent sequencing runs were included to account for contamination and stochastic noise. No microbial signal distinct from negative controls was detected, and aerobic and anaerobic bacteria (*Staphylococcus epidermidis* and *Cutibacterium acnes* [formerly *Propionibacterium acnes*]) detected by culture of fetal samples were identified by the authors as skin contaminants.

To directly compare these recently published reports, we re-analysed the publicly available unfiltered relative abundance data associated with the three publications that reported sequence data and determined the relative abundance of each detected genus. While there was good agreement between the two studies using second trimester vaginally delivered fetuses[38,43], the bacterial taxa detected in fetuses derived by C-section[28] were vastly different (Figure 1). The number of genera was much lower in C-section-derived fetuses, and entire groups of microbes, especially those generally found in the vagina, were absent. Most importantly, in the studies that claimed fetal microbial colonization[38,43], every genus detected in fetal samples was also detected in most control samples. These findings indicate that the claimed microbiology of the human fetus is dependent on the methodology of sampling. Next, we apply perspectives from different disciplines to provide context and implications for the findings.

Reproductive biology and obstetrics perspectives

The embryo and fetus develop within the uterus but not in the uterine cavity, *per se*. The early embryo invades the maternal decidua and is completely embedded by 10 days post-fertilization. The fetus grows within the amniotic cavity, which originates between the trophoblast and inner cells mass in the second week post fertilization, surrounded by two layers of reproductive membranes as well as amniotic fluid. Hence, even if microbes were present in the uterine cavity[49], they would have to pass through to the amniotic cavity and reside within amniotic fluid to colonize the fetus. Of note, amniotic fluid has antimicrobial properties, being enriched for example in

Lysozyme[50], Human beta-defensin 2[51], and Gp340/Dmbt1[52] (binds and agglutinates a broad spectrum of both Gram-negative and Gram-positive bacteria).

The placenta mediates communication between the fetus and the mother and is a potent immune organ that protects the fetus. Historically, the placenta has been considered sterile (defined here as free from living microorganisms), but in 2014 a complex but low-biomass placental microbiome was detected by DNA sequencing, that showed some similarity with sequence data (Human Microbiome Project) of microbial communities of the oral cavity[14]. Contamination controls were not included in this early study, and subsequent evaluation of the work found that most genera detected are also common contaminants[24,34,36,53]. Several detected taxa, such as *Gloeobacter*, a genus of photosynthetic cyanobacteria, appeared biologically implausible as a component of a putative placental microbiome[22,54]. Irrespective of whether placental samples are collected by biopsy per vagina, clinically by chorionic villus sampling, or after delivery (most published studies to date have investigated the microbial communities in the placenta after delivery), it is always necessary to control for contamination, particularly from the tissues through which a placenta must pass prior to sampling. Accordingly, de Goffau *et al.*[27] detected a range of species known to dominate the vaginal microbiota[55], such as *Lactobacillus iners*, *L. jensenii*, *L. crispatus*, *L. gasseri*, and *Gardnerella vaginalis*. It is also noteworthy that when the presence of vaginal microbes and those in the laboratory reagents (the “kitome”) were accounted for, no placenta microbiome was detected in several recent studies[21,27,29-32,35].

Infection of the placenta by viral or bacterial pathogens is a well-recognized clinical phenomenon that contributes to preterm birth and neonatal sepsis. As noted by de Goffau *et al*.[27], *Streptococcus agalactiae* can be detected in around 5% of cases as the only verified bacterial signal in placentas obtained by C-section deliveries. The presence of this species is plausible as it colonizes the genital tract of about 20% of women and has invasive potential, being an important

cause of both maternal and neonatal sepsis[56]. However, the ability of specific pathogens to colonize and/or infect the placenta is not tantamount to more widespread placental microbial colonization or even the presence of an indigenous microbiome (a prevalently occurring, stable, non-pathogenic, complex microbial community).

Research claiming the presence of viable low-density microbial communities in the fetal intestine[43] and fetal organs[38] likewise calls for an evaluation of the sampling process. Mishra *et al*. obtained fetal tissues after medical termination of pregnancy in the 2nd trimester with prostaglandins[38]. This procedure typically involves the individual going through hours of labor and often leads to the rupture of the fetal membranes hours prior to vaginal delivery. Even with a standardized approach, labor may be prolonged and may be accompanied by infection and fever, which are common with 2nd trimester terminations[57,58]. Both Li *et al*.[39] and Rackaityte *et al*.[43] also used 2nd trimester terminations but obtained the fetal tissues from core facilities. The tissues used by Li *et al*. were from surgical terminations (14-23 weeks) performed with mechanical dilation. Unfortunately, Rackaityte *et al*.[37] did not provide sufficient information to determine if fetuses were obtained through surgical procedures or medical inductions. While the latter increases the risk of the fetus being exposed to vaginal microbes during labour, both procedures involve delivering the fetus through the vaginal canal. As outlined later, the reported microbiology of these fetuses reflects the sources of microbes to which they are exposed.

Microbiology and microbial ecology perspectives

Host-microbe relationships range from benign mutualism (a prolonged symbiotic association from which both benefit) and commensalism (host is unaffected), to one in which the microbe harms the host (pathogen). Although claims for fetal microbial exposure[38,43] have not established the nature of the host-microbe interaction, and the duration of exposure or colonization, they have suggested a beneficial role for live organisms in fetal immune development, thereby implying a

symbiosis. The microbiological approaches applied by Rackaityte *et al.*[43] and Mishra *et al.*[38] are, in large part, robust, and well suited to study symbiotic microbial populations. The combination of 16S rRNA gene sequencing, quantitative PCR (qPCR), microscopy, FISH, and culture is laudable, as the approaches are complementary. Next-generation sequencing of 16S rRNA gene amplicons provides a broad community overview and can detect microbes that escape cultivation, while qPCR, microscopy, and bacterial cultures have a high dynamic range, very low detection limits, and reasonable specificity. The DNA sequence-based microbiota composition data in both studies is quite consistent (Figure 1), suggesting that several of the bacterial taxa detected were present in the samples and not artifacts derived from laboratory reagents or DNA-isolation kit contamination. However, although the microbiological analyses of samples were sound, the sampling procedures do not preclude the introduction of contaminant species at the sample collection stage, and critical controls to determine if contamination occurred were missing.

In agreement with the unavoidable vaginal exposure of fetuses obtained by 2nd trimester abortions (see above), both Rackaityte *et al.*[43] and Mishra *et al.*[38] found the genera *Lactobacillus* and *Gardnerella*, which dominate the vaginal microbiota[55], among their most consistent findings (Figure 1). The species cultured by Mishra *et al.*, *G. vaginalis*, *L. iners* and *L. jensenii*, are highly specific to the human vagina[59]. Other microbes detected such as *Staphylococcus* species and *Cutibacterium acnes*, are skin commensals. As shown in Figure 1, abundances of *Lactobacillus*, *Gardnerella*, and *Staphylococcus* found by Mishra *et al*. showed gradients with high population levels in fetal samples exposed to sources of contaminants (placenta and skin) and lower levels in internal samples (gut, lung, spleen, thymus). The omission of vaginal controls by both Rackaityte *et al*. and Mishra *et al*. to determine the microbiota of vaginally delivered fetuses is an unfortunate flaw that casts doubt on the authors' conclusion that the microbes originate from the womb. Indeed, Li *et al*.[39], who used samples from 2nd trimester surgical terminations performed with mechanical dilatation, which decreases the bacterial exposure of the fetus, did not report

positive bacterial PCR results in their study, further raising suspicion that sampling contamination was a serious confounder in the work of Rackaityte *et al.* and Mishra *et al.*.

Although vaginal controls were not included by Rackaityte *et al.*[43] and Mishra *et al.*[38], direct comparisons of their findings with those by Kennedy *et al*.[28] also provide clear evidence for vaginal contamination of terminated fetuses (Figure 1). The C-section derived fetal samples in Kennedy *et al.*, which were not exposed to the vagina, carried no *Gardnerella* or *Lactobacillus* but instead contained skin and reagent contaminants[28,53]. Despite attempts to reduce contamination, C-section derived fetal meconium had at least one positive culture[28]. Kennedy *et al*. did not consider these microbes of fetal origin, as they were skin commensals, and half of the samples as well as many culture replicates did not show growth. The authors concluded that such inconsistencies point to stochastic contamination and not colonization by a stable functional microbial community.

Despite vaginal contamination, the bacterial load found in terminated fetuses was extremely low[38,43]. Signals derived from qPCRs were only marginally higher than those of controls, with Mishra *et al*. reporting cycle thresholds (Ct) of >30 cycles, with Ct values for negative controls around 31-32 cycles. Cell counts as detected by both microscopy and culture were also low. Mishra *et al*. reported fewer than 100 colonies on average per entire fetus, with many fetuses and tissues being negative for the specific microbes (see Table S6 in the original publication[38]). Such inconsistent patterns are not logical based on ecological principles and do not resemble natural microbial populations, which should be consistently detectable, especially in sample replicates. Given that they are close to the detection limits of the technical approaches used, such findings should raise concerns of contamination rather than suggesting colonization.

Further indirect insights regarding the microbiological state of the fetus may be inferred from the infant gut microbiota very early in life. Neonatal meconium samples have been studied for a

century by culture-based methods and more recently by DNA sequencing; this has also sometimes yielded contradictory findings[10,41,42,60] due to contamination and because postnatal colonization may occur before a meconium is delivered[24]. However, when meconium appears early, culturable bacteria are seldom detected (as reviewed by Perez-Munoz *et al*.[24]). In agreement with this, an analysis of meconium samples collected from extremely premature infants[61] showed that taxa identified as contaminants[34,36] make up a large proportion of sequences in meconium collected within the first 3 days after delivery and then drop to almost zero in most samples at days 4-6 (Figure 2), suggesting that the genuine bacterial signal is low in early meconium.

Relatedly, members of a putative fetal microbiome should be, in theory, detectable independent of birth mode. There is indeed some overlap between the reported fetal microbial taxa[38,43], e.g. staphylococci, enterococci, lactobacilli, and enterobacteria, and the microbiota detected in infant fecal samples in the first week[62-64]. However, there have been few attempts to track species and strains to confirm fetal origin. One study investigated gastric aspirates of newborn infants collected immediately after birth[65], which should contain microbes reported *in utero* as the fetus swallows amniotic fluid. However, aspirates from vaginally-born infants contained the specific *Lactobacillus* species (*L. iners* and *L. crispatus*) that also dominate the microbiota of the vagina, while most samples from C-section deliveries clustered with negative controls[65]. This finding is consistent with vaginal transfer of microbes to a sterile fetus during delivery. In addition, many of the genuine bacterial signals that were detected in early meconium[61] were typical maternal skin representatives (*Staphylococcus* & *Corynebacterium*) and were strongly associated with C-section, or were maternal fecal microbiota representatives (*Escherichia* & *Bacteroides*) associated with vaginal delivery (Figure 2), indicating that these genuine signals were derived from microbes acquired *ex-utero*.

Research is beginning to determine the origin of post-partum neonatal microbial colonizers and has shown a delay in appearance of bacterial species presumed to originate from the mother's gut (e.g. *Bifidobacterium* and *Bacteroides* species) in early fecal samples of infants born by C-sections[62,63,66-68]. A substantial proportion of strains acquired by infants postnatally can be traced back to their mothers[68-70], and fecal microbiota transplant (FMT) restores the microbiome in C-section delivered infants[71]. Thus, the published evidence, although still incomplete, suggests that the early life microbiome in humans is acquired through the vertical and horizontal transfer of microbes whose origin is fecal or environmental (from outside) rather than fetal (from inside).

Bioinformatic and data science perspectives

Characterization of low-biomass samples by 16S rRNA gene amplicon sequencing is challenging as DNA contamination can occur from the microbial DNA present in reagents, tools, instruments, and DNA isolation kits[34-36] and through cross-contamination between PCR tubes/wells, sequencing runs, or sequencing lanes[35]. A common misconception in the field of low microbial biomass samples is that the use of negative controls is sufficient to account for all kinds of contaminants. Commonly, imperfect negative controls are used that account only for a limited number of the sample processing steps or are not spread evenly amongst all batches (thus not accounting for processing days, reagent batches, different sequencing runs), leading to batch effects which may be mistaken for genuine signals[44]. Overreliance on or under analysis of such negative controls in combination with the misuse of contamination removal programs like Decontam[72], specifically by not having negative controls in all batches, frequently results in false retention of contaminants[44]. Even with appropriate controls, it is challenging to separate genuine signals from low abundance contaminants due to the law of small numbers, which means that contaminant signals may appear sporadically in samples and negative controls[73]. Thus, suboptimal handling of sequencing control samples may not reveal the full spectrum of contaminants because only the most abundant contaminant species are consistently

406 detected. On the other hand, potentially genuine sample-associated signals sometimes also
407 erroneously end up in negative controls through cross-contamination during PCR or sequencing
408 (machine contamination)[35].
409
410 Unfortunately, both Rackaityte *et al.*[43] and Mishra *et al.*[38] reported taxa as legitimate findings that
411 are typical contaminants (Figure 1). The most obvious case is *Bradyrhizobium*, which is one of
412 the most dominant and consistent contaminants found in sequencing studies[36,74]. Rackaityte *et*
413 *al.* reported *Micrococcus* and *Lactobacillus* as genuine fetal inhabitants, but a re-analysis of the
414 data revealed that this finding was driven by a batch effect[44]. Although the authors rejected this
415 conclusion[37], this batch effect is clearly visible if the findings of the two batches are plotted
416 together (Figure 3). In addition, Mishra *et al.* considered their signal for *Micrococcus* to be derived
417 from contamination[38]. *Afipia*, *Flavobacterium*, *Pseudomonas*, and *Brevundimonas* are genera
418 reported by Mishra *et al.*[38] that are commonly detected as kit or laboratory reagent
419 contaminants[34,36].
420
421 Mishra *et al*. and Rackaityte *et al.* also reported marginally higher total bacterial load in fetal
422 samples as compared to controls, using qPCR[38,43]. However, eukaryotic DNA in tissue samples
423 (which is absent in negative controls) might have a DNA carrier effect leading to a more efficient
424 DNA precipitation of prokaryotic reagent contaminants. In addition, bacterial PCR primers also
425 amplify mitochondrial DNA, which is evolutionarily of bacterial origin. Together these factors may
426 explain why samples from low-biomass studies are often reported as having more bacterial DNA
427 than controls and show that this cannot always be relied upon as evidence for the presence of
428 microbes. Rackaityte *et al*. depleted human mitochondrial DNA (mtDNA) from their 16S rRNA
429 gene sequence set that co-amplified in the PCR, but neither study accounted for mtDNA in their
430 qPCR analysis, although their primers targeted the 16S rRNA gene and were therefore potentially
431 susceptible to cross-reactivity[38,43].

Immunological perspective

The enteric microbiota in general, and some microbial taxa in particular, undoubtedly act as potent drivers of adaptive mucosal immune maturation and priming in the adult host[75-78]. Besides their intrinsic immunogenic nature, microorganisms also generate metabolites that critically promote and shape immune maturation and priming[79-81]. Although the early fetal immune system is immature, recent research demonstrates migration of fetal dendritic cells (DCs) to the mesenteric lymph nodes; somatic hypermutation in fetal B cells; and increasing T cell receptor repertoire diversity, evenness and activation during late fetal development[7,82,83].

The existence of metabolically active microbes in the fetus could, in principle, provide one possible explanation for these findings. Mishra *et al.*[38] used an autologous T cell expansion assay to show that fetal DCs loaded with antigen from bacteria that had been isolated from fetal tissues stimulated proliferation of CD45RO+ and CD69+ T cells. T cell proliferation was reduced but still detectable in the absence of DC-derived cytokine release suggesting an activated memory response[38]. Demonstration that the fetal T cell memory response is specific for the bacteria present in one individual fetus would be necessary to strengthen the interpretation that specific immune responses are routinely driven by fetal bacterial colonization. There are alternative explanations for fetal immune responses apart from *bona fide* microbial colonization. Maternal antigen-IgG complexes have been detected in cord blood and transplacental immune priming of the fetal immune system in early gestation has been demonstrated[84,85] Cross-reactivity, as observed for microbiota reactive enteric secretory immunoglobulin A, would support fetal priming by maternal microbial antigens[80]. Similarly, maternal microbiota-derived microbial molecules partly bound to IgG stimulated innate immune maturation of the murine fetal gut[47], and maternal intestinal carriage of *Prevotella* protected the offspring from food allergy in humans[86]. Thus,

maternal microbiota-derived microbial antigens and metabolites may pass the placental filter directly or bound to IgG and evoke the observed primary fetal immune response[87].

If a significant biomass of microbes in fetal tissues is not rapidly cleared, it implies either overt infection and inflammation, or mechanisms of immune or microbial adaptation for symbiosis. At present, we have no clear evidence for such a symbiosis. Bacteria detected in fetal tissues from the genera *Staphylococcus, Escherichia, Enterococcus* or *Pseudomonas* represent important causative agents of infection in human preterm neonates (see section below on clinical microbiology). These can withstand the host's innate defence system at least to some extent and provoke an inflammatory response[88]. Such bacteria are also capable of very rapid replication, as they expand several million-fold during microbiota assembly after birth[89]. Their presence in placental tissue in the absence of an inflammatory tissue response or colonization of fetal mucosal surfaces would require highly efficient host mechanisms of immune control and bacterial growth restriction, which are unlikely considering the immature state of the fetal immune system. On the other hand, bacteria such as *Micrococcus*, which were detected in fetal intestines by Rackaityte *et al.*[37], rarely cause invasive infection in humans. Their prolonged presence within healthy tissues such as the placenta would require bacterial mechanisms of resistance against antimicrobial effector molecules of the host innate immune system such as complement. Such mechanisms have not been described for the genus *Micrococcus*, which is an environmental organism found in water, dust, and soil, and is also a common contaminant[45,46].

From an immunological perspective, the hypothesis of a fetal microbiome therefore requires the identification of mechanisms that control and tolerate bacterial populations and prevent overt inflammation and inflammation-driven tissue destruction in the presence of viable and metabolically active microorganisms, many of which are opportunistic pathogens (see below). Alongside this, mechanisms by which the commensal or symbiotic microbes survive the immune

response would also have to be identified, and it is unclear how the fetal immune system would differentiate between pathogens and symbionts once protective barriers are breached. Given that such immunological and anatomical mechanisms have not been identified or even proposed[26], the observed immune maturation and priming during fetal development is most likely not induced through colonization of the fetus with live microbes but rather through maternal immune components or microbial fragments and metabolites crossing the placental barrier.

Clinical microbiology perspective

No part of the human body is impregnable to bacterial invasion. Transient bloodstream bacteraemia is associated with something as innocuous as tooth brushing[90], and most host tissues can tolerate occasional ingress by microbes. However, to avoid serious pathology bacteraemia must be rapidly cleared by innate immune mechanisms and inflammation. Some pathogens establish persistent infections that may be asymptomatic either by evading the immune system or by forming persister cells in response to antibiotic treatment[91]. The claims for non-pathogenic fetal microbial exposure[38,43] have not established whether host-microbe interactions reflect small scale translocation, asymptomatic infection, persistent symbiosis or mutualism, and how microbes might persist at low levels without immune elimination and without harming the host.

The 'fetal-enriched taxa' reported by Mishra *et al*. include *Flavobacterium, Lactobacillus, Staphylococcus, Afipia, Pseudomonas, Bradyrhizobium*, and *Brevundimonas*[38]*.* They also report successful culturing of lactobacilli and staphylococci from fetal tissue, but the lack of unambiguous species-level taxonomic identification of the cultured organisms is an unfortunate and significant technical limitation. Lactobacilli are usually of low pathogenic potential, they inhabit external mucosal surfaces of healthy humans, including the nose[92] and vagina[55], and they are often used as probiotics[93]. However, some strains and species lactobacilli do express potential virulence

509 factors such as fibrinogen-binding, platelet-aggregation[94] and inerolysin[95] and have the ability to
510 adhere to biotic surfaces with pili[96]. Furthermore, their ability to resist oxidative stress[97] and grow
511 in the absence of iron[98], allows them to cause serious infections such as endocarditis when
512 provided with the opportunity to access the bloodstream[99,100]. Such systemic infections can be
513 life-threatening with mortality rates as high as 30%[100]. This casts doubt on the interpretation of
514 lactobacilli being asymptomatic colonizers of fetal tissue rather than contaminants that are picked
515 up during vaginal delivery.
516
517 A greater challenge arises when species of the genus *Staphylococcus* are considered, particularly
518 strains that were cultured from fetal tissue and that exhibit high-level 16S rRNA gene sequence
519 identity (99-100%) to *Staphylococcus aureus* and several closely related coagulase-negative
520 *Staphylococcus* species (CoNS)[38]. These organisms can be long-term colonizers of external
521 mucosal surfaces of humans [101,102], do not typically cause disease unless the mucosal barrier is
522 breached. However, once they bypass mucosal barriers, they can deploy a more extensive
523 repertoire of virulence factors to invade tissues by degrading connective tissues and, in the case
524 of *S. aureus*, a repertoire of over a dozen cytolytic toxins genes that kill human cells[103,104]. CoNS,
525 on the other hand, are ubiquitous skin colonizers, and their detection in clinical diagnostic
526 laboratories (which is so common that it is considered a major diagnostic challenge[105,106]) is
527 usually assumed to reflect contamination from the patient and occasionally the healthcare worker,
528 in the absence of other reasons to suspect a CoNS infection[77-79]. There are, however, distinct
529 clinical scenarios where the presence of CoNS and their pathogenic capacity are considered
530 critical. For example, in patients with indwelling devices and in preterm neonates, where they are
531 the most common cause of late-onset neonatal sepsis[107]. Therefore, given that they are either
532 contaminants or overt pathogens, the detection of staphylococci, no matter whether *S. aureus* or
533 CoNS, is difficult to accept as evidence for *in utero* colonization of a healthy fetus.
534

Other bacteria identified as part of a notional “fetal microbiome”, such as *Enterococcus faecalis* and *Klebsiella pneumoniae*, are equally problematic. These belong to a group known as “ESKAPE pathogens”, which include *Enterococcus faecium*, *Staphylococcus aureus*, *Klebsiella pneumoniae*, *Acinetobacter baumannii*, *Pseudomonas aeruginosa*, and *Enterobacter* species. The lethality of tissue colonization with ESKAPE pathogens is well documented in mouse models, and these microbes are leading causes of healthcare-acquired infections worldwide with significant mortality and morbidity, even when treated with antibiotics[108]. Several ESKAPE pathogens readily survive in adverse conditions outside of vertebrate hosts, including drying, oxidative stress, and exposure to heat or sanitation chemicals[109]. They are likely to persist on inanimate surfaces including utensils or clinical fabrics[110,111], thereby increasing their likelihood of being contaminants. While these microorganisms were not reported at the species level[38], it is noteworthy that closely related organisms can also cause neonatal sepsis[112-114] which makes them unlikely colonizers of a healthy fetus.

A consideration prompted by a notional fetal microbiome is the possibility that the fetus might cope better with nosocomial pathogens than neonates or even adults. However, there is ample evidence to show that amniotic fluid, the placenta and fetal tissues are highly susceptible to bacterial infection, and the outcomes of infections with *Streptococcus agalactiae* or *L. monocytogenes* are often catastrophic[115,116]. Importantly, in *L. monocytogenes* infections that occur during the third trimester of pregnancy, fetal infection progresses while the mother’s infection can be cleared, indicating that the placenta and fetus do not have greater resistance to infection than an adult human. Therefore, from a clinical perspective, most interpretations brought forward in recent publications[38,43] on the presence of microbes in fetuses seem to be biologically difficult to reconcile as it is highly plausible that they would result in harm or death of the fetus. In agreement with this conclusion, in a series of well-controlled studies in various clinical settings,

DiGiulio and co-workers found no evidence for microbes in amniotic fluid except when associated with neonatal morbidity and mortality[117-120].

Gnotobiology perspective

The traditional assumption that the human fetus is free from other life forms *in utero* is based primarily on the observation that, with few exceptions, bacterial and viral pathogens that infect the mother are incapable of crossing the placental barrier to infect the fetus[121-123]. Additionally, the amnio-chorionic membranes enclosing the fetus in the uterine cavity, as well as the cervical mucus plug, protect the fetus from external microbes. Sterility of the fetus is the basis for the derivation by hysterectomy of germ-free mammals (mainly mice and rats, but also pigs and other species[24]), which have long been used to study the biochemical, metabolic, and immunological influences of microbes on their mammalian hosts[124-126]. The primary consideration is whether germ-free animals are truly 'free of all demonstrable forms of microbial life'[127]. If they lack microbial associates, there cannot be a fetal microbiome. Testing germ-free animals for contaminating microbes uses microscopic observation of stained fecal smears, culture of feces in nutrient media under various conditions of temperature and gaseous atmosphere[122,127-129], PCR using 'universal bacterial' primers[128,130], and serological assays for viral infections[131]. These tests consistently demonstrate an absence of microbial associates. Therefore, gnotobiology provides strong evidence that the fetus *in utero* is sterile.

Summary - the experimental evidence indicates that a healthy human fetus is effectively sterile

In this perspective, we have applied a trans-disciplinary approach focused on scrutiny of existing evidence and mechanistic explanations and conclude that the evidence is strongly in favour of the sterile womb hypothesis. Although it is impossible to disprove the occasional presence of live

585 microbes in a typical human fetus, the available data does not support stable, functional,
586 nontrivially abundant colonizers under normal, non-pathogenic circumstances. We are aware that
587 our position conflicts with dozens of publications that claim evidence for *in utero* microbial
588 populations, but we feel confident about the validity of our multi-layered approach. Our aim was
589 to bring additional clarity to the debate and suggest re-focussing scientific effort towards other
590 concepts that will provide solid scientific foundations, enable translation, and improve maternal-
591 fetal and child health through appropriate research priorities and use of resources.
592
593 The processes by which the fetus matures and becomes immunologically equipped for life in a
594 microbial world have life-long implications and is one of the most important areas in biology and
595 medicine. This research calls for scientific minds that are open to fresh thinking and willing to
596 change, and no dogma, no matter how well established, is exempt from scrutiny. Notwithstanding
597 the caution and safeguards recommended in this perspective, scientists should not be dissuaded
598 from exploring the microbial drivers of fetal immune development. Paradoxically, we contend that
599 sterile tissues are both immunologically and microbiologically fascinating. How does the fetus
600 mature and become immunologically equipped for life in a microbial world in the absence of direct
601 exposure to live microbes? Are maternal-derived microbial metabolites sufficient for fetal immune
602 education? Future research could include exploration of how maternal microbial-derived
603 metabolites and small molecules, as well as maternal immune components, prepare the fetus for
604 the microbial challenges of post-natal life[87].
605
606 **Considerations for the critical evaluation of low- or no biomass samples**
607 Contamination has always been a confounder in microbiology but is of particular concern for those
608 studying low- or no biomass samples.[34,36]. The issue has been highlighted by recent reports of
609 human tissues, such as blood, brain, and cancers (Box 1), previously thought to contain no, or
610 very little, bacterial biomass, to harbour diverse microbial communities. As with intrauterine

611 studies described above, these microbial populations are generally discussed in light of their
612 importance for human diseases and health. In instances of contamination, a tissue may be
613 misjudged as non-sterile, whereas in others, a real microbiological signal may be obfuscated by
614 contamination.
615
616 As Saffarian et al[132] point out, one is faced in studies on low biomass samples with the difficult
617 exercise of extracting relevant signals from among contaminating noise that cannot be rationally
618 eliminated. The removal of all sequences present in negative-control samples or that have been
619 previously identified as contaminants in the literature may result in loss of relevant biological
620 signals. Post-sequencing contamination removal using software packages such as Decontam[72]
621 or other statistical approaches[34] have been developed to remove the more abundant
622 contaminants, leading to microbiome profiles that are more likely to reflect the real community.
623 Practical examples of contamination removal in 16S rRNA gene sequence data is provided by
624 Heida et al.[61] and Saffarian et al[132], and we extend on these examples in Box 1. There is clearly
625 a need for formal standardisation of best practices in the analysis of low and putative “no biomass”
626 samples.
627
628 We draw attention to the distinction between “low biomass” and no biomass samples. This has
629 practical significance; true “low (microbial) biomass” samples are amenable to contamination-
630 removal approaches described above, but “no (microbial) biomass” samples require a different
631 approach (Box 1). For credible proof of the presence of microbes, multiple layers of evidence are
632 required, first with quantitative, sensitive (lower detection limit) approaches such as quantitative
633 PCR with strict controls before contamination-sensitive sequencing approaches are applied.
634 Since contamination removal will provide data regardless of whether microbes are present or
635 absent, the starting proposition should be the null hypothesis to avoid confirmation bias,

particularly when results are inconsistent and at the outer technical limits for detection or if results defy mechanistic plausibility.

Given the limitation of sequencing approaches, confirmation by alternative methods, such as FISH and culture, are required. However, the flaws of the recent studies on fecal microbial populations demonstrates that even a combination of approaches has the potential to produce false findings, as contamination during sampling is a considerable challenge. We posit that studies on all low biomass samples can benefit from a similar trans-disciplinary assessment as applied above for fetal samples to interpret findings considering biological and mechanistic explanations[26]. When obligately photosynthetic, psychrophilic, thermophilic, halophilic, or chemolithoautotrophic bacteria are found in human tissues which do not provide the growth conditions for such organisms[22,133], or if the detected genera are known contaminants of laboratory kits/reagents that should not have escaped decades of culture studies, such as Proteobacteria (*Pseudomonas and E.coli* for example)[134-136], the authenticity of such signals must be questioned.

Box 1: Experimental considerations for biological samples containing different levels of biomass.

High biomass samples

***Examples*:** Faeces, dental plaque, wastewater treatment plant samples.

***Impact of contamination*:** Very low. The high microbial biomass derived from the sample dominates the signal derived from background contamination, meaning most observations are robust.

***Mitigations*:** Experimental design seldom needs to be significantly adjusted to account for contamination, beyond monitoring "blank" negative control samples that reveal which contaminating species are present and basic post sequencing analysis. Sequencing controls and removing samples with significant contamination levels is nevertheless prudent.

Low biomass

***Examples*:** Skin Swabs, nasal tract swabs, breastmilk, most respiratory tract samples, tissue biopsies & mucosal samples, including intestinal crypts.

***Impact of contamination*:** Ranges from low to high. Contaminated samples are progressively affected with reducing input microbial biomass[36].

***Mitigations*:** Inclusion of multiple controls facilitate contamination recognition. When possible, samples should be concentrated prior to processing to increase input biomass. Advance consideration of potential sources of contamination during the sample acquisition stage is always recommended. After sample collection, processing should be carried out in a clean-room environment, preferably with all surfaces bleached and UV-treated. The extraction step may benefit from use of non-kit-based methods (e.g. phenol-chloroform extractions) where plasticware and individual reagents are UV-treated prior to use. Contamination from DNA isolation and PCR kits is usually identifiable, particularly if well-defined and controlled batch effects are created using

678 different lot numbers of particular kits. Regardless of the DNA extraction method, the presence of
679 contaminants should be monitored by including “blank” negative controls. The inclusion of controls
680 generated by serial dilution of DNA of known composition (e.g. mock community) will indicate the
681 biomass level at which contamination becomes a dominant feature of sequencing results.
682 Contamination may also be estimated prior to sequencing by qPCR using serially diluted known
683 quantities of spiked DNA. Post-sequencing analyses, using programs like Decontam, and
684 analysis steps as described by de Goffau et al.[34] and used by Heida et al.[61] will usually identify
685 contaminants. To elucidate the source of contaminants introduced during the sample collection
686 stage, sufficient numbers of samples acquired with different methods should be included.
687
688 **Samples in which the existence of microbes is not established (potential “No-biomass”**
689 **samples)**
690 ***Examples*****:** Placental and fetal tissues, amniotic fluid, brain tissue and cerebrospinal fluid, blood,
691 bone, and internal cancer tissues.
692 ***Impact of contamination*****:** High and potentially up to 100%, unless infection, injury is present.
693 ***Mitigations*****:** Experimental design should be robust and directed specifically against
694 contamination. An initial assessment using quantitative methods (e.g. qPCR) with low detection
695 limit and microscopic visualisation (e.g. Gram staining/labelling by FISH) is required to determine
696 if microbes are present, before embarking on a sequence-based approach. Note such
697 approaches are still susceptible to sample contamination and other artefacts (e.g. non-specific
698 staining or auto-fluorescence from mucins, can sometimes appear “microbe-like” in size and
699 shape)[44]. All mitigations outlined for “Low biomass” samples above should be adopted.
700 Furthermore, repeat sample analysis with different DNA extraction kits/methods[30] and/or at
701 different days[137]. These will track the presence of particular species in sequencing profiles
702 associated with specific kits/reagents or environment. Species that are repeatedly detected
703 regardless of technical approach used are more likely to be genuine signals, unless they were

introduced during the sample collection. Binary statistics (absence/presence) are recommended. Ideally, the presence of microbes identified by sequencing should be verified with a different technique such as cultivation, another sequencing technique with sufficient taxonomic resolution, and a species-specific qPCR or FISH using high magnification to visualize the size and morphology of individual microbial cells.

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References

1 Macpherson, A. J., de Aguero, M. G. & Ganal-Vonarburg, S. C. How nutrition and the maternal microbiota shape the neonatal immune system. *Nat Rev Immunol* **17**, 508-517, doi:10.1038/nri.2017.58 (2017).

2 Kalbermatter, C., Fernandez Trigo, N., Christensen, S. & Ganal-Vonarburg, S. C. Maternal Microbiota, Early Life Colonization and Breast Milk Drive Immune Development in the Newborn. *Front Immunol* **12**, 683022, doi:10.3389/fimmu.2021.683022 (2021).

3 Gensollen, T., Iyer, S. S., Kasper, D. L. & Blumberg, R. S. How colonization by microbiota in early life shapes the immune system. *Science* **352**, 539-544, doi:10.1126/science.aad9378 (2016).

4 Jain, N. The early life education of the immune system: Moms, microbes and (missed) opportunities. *Gut Microbes* **12**, 1824564, doi:10.1080/19490976.2020.1824564 (2020).

5 Hornef, M. W. & Torow, N. 'Layered immunity' and the 'neonatal window of opportunity' - timed succession of non-redundant phases to establish mucosal host-microbial homeostasis after birth. *Immunology* **159**, 15-25, doi:10.1111/imm.13149 (2020).

6 Torow, N., Marsland, B. J., Hornef, M. W. & Gollwitzer, E. S. Neonatal mucosal immunology. *Mucosal Immunol* **10**, 5-17, doi:10.1038/mi.2016.81 (2017).

7 Schreurs, R. *et al.* Human Fetal TNF-alpha-Cytokine-Producing CD4(+) Effector Memory T Cells Promote Intestinal Development and Mediate Inflammation Early in Life. *Immunity* **50**, 462-476 e468, doi:10.1016/j.immuni.2018.12.010 (2019).

8 Stras, S. F. *et al.* Maturation of the Human Intestinal Immune System Occurs Early in Fetal Development. *Dev Cell* **51**, 357-373 e355, doi:10.1016/j.devcel.2019.09.008 (2019).

9 Zhang, X. *et al.* CD4 T cells with effector memory phenotype and function develop in the sterile environment of the fetus. *Sci Transl Med* **6**, 238ra272, doi:10.1126/scitranslmed.3008748 (2014).

10 He, Q. *et al.* The meconium microbiota shares more features with the amniotic fluid microbiota than the maternal fecal and vaginal microbiota. *Gut Microbes* **12**, 1794266, doi:10.1080/19490976.2020.1794266 (2020).

11 Stinson, L. *et al.* Comparison of Bacterial DNA Profiles in Mid-Trimester Amniotic Fluid Samples From Preterm and Term Deliveries. *Front Microbiol* **11**, 415, doi:10.3389/fmicb.2020.00415 (2020).

12 Younge, N. *et al.* Fetal exposure to the maternal microbiota in humans and mice. *JCI Insight* **4**, doi:10.1172/jci.insight.127806 (2019).

13 Stinson, L. F., Boyce, M. C., Payne, M. S. & Keelan, J. A. The Not-so-Sterile Womb: Evidence That the Human Fetus Is Exposed to Bacteria Prior to Birth. *Front Microbiol* **10**, 1124, doi:10.3389/fmicb.2019.01124 (2019).

14 Aagaard, K. *et al.* The placenta harbors a unique microbiome. *Sci Transl Med* **6**, 237ra265, doi:10.1126/scitranslmed.3008599 (2014).

15 Tissier, H. *Recherches sur la flore intestinale des nourrissons (état normal et pathologique)*. (1900).

16 D'Argenio, V. The Prenatal Microbiome: A New Player for Human Health. *High Throughput* **7**, doi:10.3390/ht7040038 (2018).

17 Funkhouser, L. J. & Bordenstein, S. R. Mom knows best: the universality of maternal microbial transmission. *PLoS Biol* **11**, e1001631, doi:10.1371/journal.pbio.1001631 (2013).

18 Stinson, L. F., Payne, M. S. & Keelan, J. A. Planting the seed: Origins, composition, and postnatal health significance of the fetal gastrointestinal microbiota. *Crit Rev Microbiol* **43**, 352-369, doi:10.1080/1040841X.2016.1211088 (2017).

19 Walker, R. W., Clemente, J. C., Peter, I. & Loos, R. J. F. The prenatal gut microbiome: are we colonized with bacteria in utero? *Pediatr Obes* **12 Suppl 1**, 3-17, doi:10.1111/ijpo.12217 (2017).

20 Blaser, M. J. *et al.* Lessons learned from the prenatal microbiome controversy. *Microbiome* **9**, 8, doi:10.1186/s40168-020-00946-2 (2021).

21 Bushman, F. D. De-Discovery of the Placenta Microbiome. *Am J Obstet Gynecol* **220**, 213-214, doi:10.1016/j.ajog.2018.11.1093 (2019).

22 Editorial. Microbiome studies and "blue whales in the Himalayas". *Lancet Infect Dis* **18**, 925, doi:10.1016/S1473-3099(18)30503-6 (2018).

23 Hornef, M. & Penders, J. Does a prenatal bacterial microbiota exist? *Mucosal Immunol* **10**, 598-601, doi:10.1038/mi.2016.141 (2017).

24 Perez-Munoz, M. E., Arrieta, M. C., Ramer-Tait, A. E. & Walter, J. A critical assessment of the "sterile womb" and "in utero colonization" hypotheses: implications for research on the pioneer infant microbiome. *Microbiome* **5**, 48, doi:10.1186/s40168-017-0268-4 (2017).

25 Segata, N. No bacteria found in healthy placentas. *Nature* **572**, 317-318, doi:10.1038/d41586-019-02262-8 (2019).

26 Walter, J. & Hornef, M. W. A philosophical perspective on the prenatal in utero microbiome debate. *Microbiome* **9**, 5, doi:10.1186/s40168-020-00979-7 (2021).

27 de Goffau, M. C. *et al.* Human placenta has no microbiome but can contain potential pathogens. *Nature* **572**, 329-334, doi:10.1038/s41586-019-1451-5 (2019).

28 Kennedy, K. M. *et al.* Fetal meconium does not have a detectable microbiota before birth. *Nat Microbiol*, doi:10.1038/s41564-021-00904-0 (2021).

29 Kuperman, A. A. *et al.* Deep microbial analysis of multiple placentas shows no evidence for a placental microbiome. *BJOG* **127**, 159-169, doi:10.1111/1471-0528.15896 (2020).

30 Lauder, A. P. *et al.* Comparison of placenta samples with contamination controls does not provide evidence for a distinct placenta microbiota. *Microbiome* **4**, 29, doi:10.1186/s40168-016-0172-3 (2016).

31 Leiby, J. S. *et al.* Lack of detection of a human placenta microbiome in samples from preterm and term deliveries. *Microbiome* **6**, 196, doi:10.1186/s40168-018-0575-4 (2018).

32 Theis, K. R. *et al.* Does the human placenta delivered at term have a microbiota? Results of cultivation, quantitative real-time PCR, 16S rRNA gene sequencing, and metagenomics. *Am J Obstet Gynecol* **220**, 267 e261-267 e239, doi:10.1016/j.ajog.2018.10.018 (2019).

33 Sterpu, I. *et al.* No evidence for a placental microbiome in human pregnancies at term. *Am J Obstet Gynecol* **224**, 296 e291-296 e223, doi:10.1016/j.ajog.2020.08.103 (2021).

34 de Goffau, M. C. *et al.* Recognizing the reagent microbiome. *Nat Microbiol* **3**, 851-853, doi:10.1038/s41564-018-0202-y (2018).

35 Olomu, I. N. *et al.* Elimination of "kitome" and "splashome" contamination results in lack of detection of a unique placental microbiome. *BMC Microbiol* **20**, 157, doi:10.1186/s12866-020-01839-y (2020).

36 Salter, S. J. *et al.* Reagent and laboratory contamination can critically impact sequence-based microbiome analyses. *BMC Biol* **12**, 87, doi:10.1186/s12915-014-0087-z (2014).

37 Rackaityte, E. *et al.* Corroborating evidence refutes batch effect as explanation for fetal bacteria. *Microbiome* **9**, 10, doi:10.1186/s40168-020-00948-0 (2021).

38 Mishra, A. *et al.* Microbial exposure during early human development primes fetal immune cells. *Cell*, doi:10.1016/j.cell.2021.04.039 (2021).

39 Li, Y. *et al.* In utero human intestine harbors unique metabolomic features including bacterial metabolites. *JCI Insight*, doi:10.1172/jci.insight.138751 (2020).

40 Lim, E. S., Rodriguez, C. & Holtz, L. R. Amniotic fluid from healthy term pregnancies does not harbor a detectable microbial community. *Microbiome* **6**, 87, doi:10.1186/s40168-018-0475-7 (2018).

41 Liu, Y. *et al.* Midtrimester amniotic fluid from healthy pregnancies has no microorganisms using multiple methods of microbiologic inquiry. *Am J Obstet Gynecol* **223**, 248 e241-248 e221, doi:10.1016/j.ajog.2020.01.056 (2020).

42 Rehbinder, E. M. *et al.* Is amniotic fluid of women with uncomplicated term pregnancies free of bacteria? *Am J Obstet Gynecol* **219**, 289 e281-289 e212, doi:10.1016/j.ajog.2018.05.028 (2018).

43 Rackaityte, E. *et al.* Viable bacterial colonization is highly limited in the human intestine in utero. *Nat Med* **26**, 599-607, doi:10.1038/s41591-020-0761-3 (2020).

44 de Goffau, M. C., Charnock-Jones, D. S., Smith, G. C. S. & Parkhill, J. Batch effects account for the main findings of an in utero human intestinal bacterial colonization study. *Microbiome* **9**, 6, doi:10.1186/s40168-020-00949-z (2021).

45 Powell, S., Perry, J. & Meikle, D. Microbial contamination of non-disposable instruments in otolaryngology out-patients. *J Laryngol Otol* **117**, 122-125, doi:10.1258/002221503762624567 (2003).

46 Wistrand, C., Soderquist, B. & Sundqvist, A. S. Time-dependent bacterial air contamination of sterile fields in a controlled operating room environment: an experimental intervention study. *J Hosp Infect* **110**, 97-102, doi:10.1016/j.jhin.2021.01.016 (2021).

47 Gomez de Aguero, M. *et al.* The maternal microbiota drives early postnatal innate immune development. *Science* **351**, 1296-1302, doi:10.1126/science.aad2571 (2016).

48 Vuong, H. E. *et al.* The maternal microbiome modulates fetal neurodevelopment in mice. *Nature* **586**, 281-286, doi:10.1038/s41586-020-2745-3 (2020).

49 Baker, J. M., Chase, D. M. & Herbst-Kralovetz, M. M. Uterine Microbiota: Residents, Tourists, or Invaders? *Front Immunol* **9**, 208, doi:10.3389/fimmu.2018.00208 (2018).

50 Cherry, S. H., Filler, M. & Harvey, H. Lysozyme content of amniotic fluid. *Am J Obstet Gynecol* **116**, 639-642, doi:10.1016/s0002-9378(15)33127-6 (1973).

51 Soto, E. *et al.* Human beta-defensin-2: a natural antimicrobial peptide present in amniotic fluid participates in the host response to microbial invasion of the amniotic cavity. *J Matern Fetal Neonatal Med* **20**, 15-22, doi:10.1080/14767050601036212 (2007).

52 Reichhardt, M. P. *et al.* The salivary scavenger and agglutinin in early life: diverse roles in amniotic fluid and in the infant intestine. *J Immunol* **193**, 5240-5248, doi:10.4049/jimmunol.1401631 (2014).

53 Sinha, R. *et al.* Assessment of variation in microbial community amplicon sequencing by the Microbiome Quality Control (MBQC) project consortium. *Nat Biotechnol* **35**, 1077-1086, doi:10.1038/nbt.3981 (2017).

54 Grettenberger, C. L. Novel Gloeobacterales spp. from Diverse Environments across the Globe. *mSphere* **6**, e0006121, doi:10.1128/mSphere.00061-21 (2021).

55 Ravel, J. *et al.* Vaginal microbiome of reproductive-age women. *Proc Natl Acad Sci U S A* **108 Suppl 1**, 4680-4687, doi:10.1073/pnas.1002611107 (2011).

56 Armistead, B., Oler, E., Adams Waldorf, K. & Rajagopal, L. The Double Life of Group B Streptococcus: Asymptomatic Colonizer and Potent Pathogen. *J Mol Biol* **431**, 2914-2931, doi:10.1016/j.jmb.2019.01.035 (2019).

57 Dodd, J. M. & Crowther, C. A. Misoprostol for induction of labour to terminate pregnancy in the second or third trimester for women with a fetal anomaly or after intrauterine fetal death. *Cochrane Database Syst Rev*, CD004901, doi:10.1002/14651858.CD004901.pub2 (2010).

58 Nijman, T. A. *et al.* Association between infection and fever in terminations of pregnancy using misoprostol: a retrospective cohort study. *BMC Pregnancy Childbirth* **17**, 7, doi:10.1186/s12884-016-1188-1 (2017).

59 Duar, R. M. *et al.* Lifestyles in transition: evolution and natural history of the genus Lactobacillus. *FEMS Microbiol Rev* **41**, S27-S48, doi:10.1093/femsre/fux030 (2017).

60 Dominguez-Bello, M. G. *et al.* Delivery mode shapes the acquisition and structure of the initial microbiota across multiple body habitats in newborns. *Proc Natl Acad Sci U S A* **107**, 11971-11975, doi:10.1073/pnas.1002601107 (2010).

61 Heida, F. H. *et al.* Weight shapes the intestinal microbiome in preterm infants: results of a prospective observational study. *BMC Microbiol* **21**, 219, doi:10.1186/s12866-021-02279-y (2021).

62 Backhed, F. *et al.* Dynamics and Stabilization of the Human Gut Microbiome during the First Year of Life. *Cell Host Microbe* **17**, 690-703, doi:10.1016/j.chom.2015.04.004 (2015).

63 Shao, Y. *et al.* Stunted microbiota and opportunistic pathogen colonization in caesarean-section birth. *Nature* **574**, 117-121, doi:10.1038/s41586-019-1560-1 (2019).

64 Podlesny, D. & Fricke, W. F. Strain inheritance and neonatal gut microbiota development: A meta-analysis. *Int J Med Microbiol* **311**, 151483, doi:10.1016/j.ijmm.2021.151483 (2021).

65 Bajorek, S. *et al.* Initial microbial community of the neonatal stomach immediately after birth. *Gut Microbes* **10**, 289-297, doi:10.1080/19490976.2018.1520578 (2019).

66 Martin, R. *et al.* Early-Life Events, Including Mode of Delivery and Type of Feeding, Siblings and Gender, Shape the Developing Gut Microbiota. *PLoS One* **11**, e0158498, doi:10.1371/journal.pone.0158498 (2016).

67 Yassour, M. *et al.* Natural history of the infant gut microbiome and impact of antibiotic treatment on bacterial strain diversity and stability. *Sci Transl Med* **8**, 343ra381, doi:10.1126/scitranslmed.aad0917 (2016).

68 Mitchell, C. M. *et al.* Delivery Mode Affects Stability of Early Infant Gut Microbiota. *Cell Rep Med* **1**, 100156, doi:10.1016/j.xcrm.2020.100156 (2020).

69 Ferretti, P. *et al.* Mother-to-Infant Microbial Transmission from Different Body Sites Shapes the Developing Infant Gut Microbiome. *Cell Host Microbe* **24**, 133-145 e135, doi:10.1016/j.chom.2018.06.005 (2018).

70 Yassour, M. *et al.* Strain-Level Analysis of Mother-to-Child Bacterial Transmission during the First Few Months of Life. *Cell Host Microbe* **24**, 146-154 e144, doi:10.1016/j.chom.2018.06.007 (2018).

71 Korpela, K. *et al.* Maternal Fecal Microbiota Transplantation in Cesarean-Born Infants Rapidly Restores Normal Gut Microbial Development: A Proof-of-Concept Study. *Cell* **183**, 324-334 e325, doi:10.1016/j.cell.2020.08.047 (2020).

72 Davis, N. M., Proctor, D. M., Holmes, S. P., Relman, D. A. & Callahan, B. J. Simple statistical identification and removal of contaminant sequences in marker-gene and metagenomics data. *Microbiome* **6**, 226, doi:10.1186/s40168-018-0605-2 (2018).

73 Dyrhovden, R. *et al.* Managing Contamination and Diverse Bacterial Loads in 16S rRNA Deep Sequencing of Clinical Samples: Implications of the Law of Small Numbers. *mBio* **12**, e0059821, doi:10.1128/mBio.00598-21 (2021).

74 Laurence, M., Hatzis, C. & Brash, D. E. Common contaminants in next-generation sequencing that hinder discovery of low-abundance microbes. *PLoS One* **9**, e97876, doi:10.1371/journal.pone.0097876 (2014).

75 Cebra, J. J., Periwal, S. B., Lee, G., Lee, F. & Shroff, K. E. Development and maintenance of the gut-associated lymphoid tissue (GALT): the roles of enteric bacteria and viruses. *Dev Immunol* **6**, 13-18, doi:10.1155/1998/68382 (1998).

76 Gaboriau-Routhiau, V. *et al.* The key role of segmented filamentous bacteria in the coordinated maturation of gut helper T cell responses. *Immunity* **31**, 677-689, doi:10.1016/j.immuni.2009.08.020 (2009).

77 Wesemann, D. R. *et al.* Microbial colonization influences early B-lineage development in the gut lamina propria. *Nature* **501**, 112-115, doi:10.1038/nature12496 (2013).

78 Li, H. *et al.* Mucosal or systemic microbiota exposures shape the B cell repertoire. *Nature* **584**, 274-278, doi:10.1038/s41586-020-2564-6 (2020).

79 Bacher, P. *et al.* Human Anti-fungal Th17 Immunity and Pathology Rely on Cross-Reactivity against Candida albicans. *Cell* **176**, 1340-1355 e1315, doi:10.1016/j.cell.2019.01.041 (2019).

80 Kabbert, J. *et al.* High microbiota reactivity of adult human intestinal IgA requires somatic mutations. *J Exp Med* **217**, doi:10.1084/jem.20200275 (2020).

81 Arpaia, N. *et al.* Metabolites produced by commensal bacteria promote peripheral regulatory T-cell generation. *Nature* **504**, 451-455, doi:10.1038/nature12726 (2013).

82 McGovern, N. *et al.* Human fetal dendritic cells promote prenatal T-cell immune suppression through arginase-2. *Nature* **546**, 662-666, doi:10.1038/nature22795 (2017).

83 Rechavi, E. *et al.* Timely and spatially regulated maturation of B and T cell repertoire during human fetal development. *Sci Transl Med* **7**, 276ra225, doi:10.1126/scitranslmed.aaa0072 (2015).

84 Casas, R. & Bjorksten, B. Detection of Fel d 1-immunoglobulin G immune complexes in cord blood and sera from allergic and non-allergic mothers. *Pediatr Allergy Immunol* **12**, 59-64, doi:10.1034/j.1399-3038.2001.012002059.x (2001).

85 Szepfalusi, Z. *et al.* Transplacental priming of the human immune system with environmental allergens can occur early in gestation. *J Allergy Clin Immunol* **106**, 530-536, doi:10.1067/mai.2000.108710 (2000).

86 Vuillermin, P. J. *et al.* Maternal carriage of Prevotella during pregnancy associates with protection against food allergy in the offspring. *Nat Commun* **11**, 1452, doi:10.1038/s41467-020-14552-1 (2020).

87 Ganal-Vonarburg, S. C., Hornef, M. W. & Macpherson, A. J. Microbial-host molecular exchange and its functional consequences in early mammalian life. *Science* **368**, 604-607, doi:10.1126/science.aba0478 (2020).

88 Henneke, P., Kierdorf, K., Hall, L. J., Sperandio, M. & Hornef, M. Perinatal development of innate immune topology. *Elife* **10**, doi:10.7554/eLife.67793 (2021).

89 van Best, N. *et al.* Bile acids drive the newborn's gut microbiota maturation. *Nat Commun* **11**, 3692, doi:10.1038/s41467-020-17183-8 (2020).

90 Lockhart, P. B. *et al.* Bacteremia associated with toothbrushing and dental extraction. *Circulation* **117**, 3118-3125, doi:10.1161/CIRCULATIONAHA.107.758524 (2008).

91 Fisher, R. A., Gollan, B. & Helaine, S. Persistent bacterial infections and persister cells. *Nat Rev Microbiol* **15**, 453-464, doi:10.1038/nrmicro.2017.42 (2017).

92 De Boeck, I. *et al.* Lactobacilli Have a Niche in the Human Nose. *Cell Rep* **31**, 107674, doi:10.1016/j.celrep.2020.107674 (2020).

93 Lebeer, S., Vanderleyden, J. & De Keersmaecker, S. C. Host interactions of probiotic bacterial surface molecules: comparison with commensals and pathogens. *Nat Rev Microbiol* **8**, 171-184, doi:10.1038/nrmicro2297 (2010).

94 Collins, J. *et al.* Fibrinogen-binding and platelet-aggregation activities of a Lactobacillus salivarius septicaemia isolate are mediated by a novel fibrinogen-binding protein. *Mol Microbiol* **85**, 862-877, doi:10.1111/j.1365-2958.2012.08148.x (2012).

95 Rampersaud, R. *et al.* Inerolysin, a cholesterol-dependent cytolysin produced by Lactobacillus iners. *J Bacteriol* **193**, 1034-1041, doi:10.1128/JB.00694-10 (2011).

96 Kankainen, M. *et al.* Comparative genomic analysis of Lactobacillus rhamnosus GG reveals pili containing a human- mucus binding protein. *Proc Natl Acad Sci U S A* **106**, 17193-17198, doi:10.1073/pnas.0908876106 (2009).

97 Wuyts, S. *et al.* Large-Scale Phylogenomics of the Lactobacillus casei Group Highlights Taxonomic Inconsistencies and Reveals Novel Clade-Associated Features. *mSystems* **2**, doi:10.1128/mSystems.00061-17 (2017).

98 Weinberg, E. D. The Lactobacillus anomaly: total iron abstinence. *Perspect Biol Med* **40**, 578-583, doi:10.1353/pbm.1997.0072 (1997).

99 Hazards, E. P. o. B. *et al.* Update of the list of QPS-recommended biological agents intentionally added to food or feed as notified to EFSA 7: suitability of taxonomic units notified to EFSA until September 2017. *EFSA J* **16**, e05131, doi:10.2903/j.efsa.2018.5131 (2018).

100 Cannon, J. P., Lee, T. A., Bolanos, J. T. & Danziger, L. H. Pathogenic relevance of Lactobacillus: a retrospective review of over 200 cases. *Eur J Clin Microbiol Infect Dis* **24**, 31-40, doi:10.1007/s10096-004-1253-y (2005).

101 Richardson, E. J. *et al.* Gene exchange drives the ecological success of a multi-host bacterial pathogen. *Nat Ecol Evol* **2**, 1468-1478, doi:10.1038/s41559-018-0617-0 (2018).

102 Gordon, R. J. & Lowy, F. D. Pathogenesis of methicillin-resistant Staphylococcus aureus infection. *Clin Infect Dis* **46 Suppl 5**, S350-359, doi:10.1086/533591 (2008).

103 Otto, M. Staphylococcus aureus toxins. *Curr Opin Microbiol* **17**, 32-37, doi:10.1016/j.mib.2013.11.004 (2014).

104 Powers, M. E. & Bubeck Wardenburg, J. Igniting the fire: Staphylococcus aureus virulence factors in the pathogenesis of sepsis. *PLoS Pathog* **10**, e1003871, doi:10.1371/journal.ppat.1003871 (2014).

105 Healy, C. M., Baker, C. J., Palazzi, D. L., Campbell, J. R. & Edwards, M. S. Distinguishing true coagulase-negative Staphylococcus infections from contaminants in the neonatal intensive care unit. *J Perinatol* **33**, 52-58, doi:10.1038/jp.2012.36 (2013).

106 Michels, R., Last, K., Becker, S. L. & Papan, C. Update on Coagulase-Negative Staphylococci-What the Clinician Should Know. *Microorganisms* **9**, doi:10.3390/microorganisms9040830 (2021).

107 Marchant, E. A., Boyce, G. K., Sadarangani, M. & Lavoie, P. M. Neonatal sepsis due to coagulase-negative staphylococci. *Clin Dev Immunol* **2013**, 586076, doi:10.1155/2013/586076 (2013).

108 Zhen, X., Lundborg, C. S., Sun, X., Hu, X. & Dong, H. Economic burden of antibiotic resistance in ESKAPE organisms: a systematic review. *Antimicrob Resist Infect Control* **8**, 137, doi:10.1186/s13756-019-0590-7 (2019).

109 Kamal, S. M., Simpson, D. J., Wang, Z., Ganzle, M. & Romling, U. Horizontal Transmission of Stress Resistance Genes Shape the Ecology of Beta- and Gamma-Proteobacteria. *Front Microbiol* **12**, 696522, doi:10.3389/fmicb.2021.696522 (2021).

110 Kramer, A., Schwebke, I. & Kampf, G. How long do nosocomial pathogens persist on inanimate surfaces? A systematic review. *BMC Infect Dis* **6**, 130, doi:10.1186/1471-2334-6-130 (2006).

111 Neely, A. N. & Maley, M. P. Survival of enterococci and staphylococci on hospital fabrics and plastic. *J Clin Microbiol* **38**, 724-726, doi:10.1128/JCM.38.2.724-726.2000 (2000).

112 Bizzarro, M. J. *et al.* Neonatal sepsis 2004-2013: the rise and fall of coagulase-negative staphylococci. *J Pediatr* **166**, 1193-1199, doi:10.1016/j.jpeds.2015.02.009 (2015).

113 Dong, Y., Speer, C. P. & Glaser, K. Beyond sepsis: Staphylococcus epidermidis is an underestimated but significant contributor to neonatal morbidity. *Virulence* **9**, 621-633, doi:10.1080/21505594.2017.1419117 (2018).

114 Glaser, M. A., Hughes, L. M., Jnah, A. & Newberry, D. Neonatal Sepsis: A Review of Pathophysiology and Current Management Strategies. *Adv Neonatal Care* **21**, 49-60, doi:10.1097/ANC.0000000000000769 (2021).

115 Nan, C. *et al.* Maternal group B Streptococcus-related stillbirth: a systematic review. *BJOG* **122**, 1437-1445, doi:10.1111/1471-0528.13527 (2015).

116 Vazquez-Boland, J. A., Krypotou, E. & Scortti, M. Listeria Placental Infection. *mBio* **8**, doi:10.1128/mBio.00949-17 (2017).

117 DiGiulio, D. B. *et al.* Microbial invasion of the amniotic cavity in preeclampsia as assessed by cultivation and sequence-based methods. *J Perinat Med* **38**, 503-513, doi:10.1515/jpm.2010.078 (2010).

118 DiGiulio, D. B. *et al.* Microbial prevalence, diversity and abundance in amniotic fluid during preterm labor: a molecular and culture-based investigation. *PLoS One* **3**, e3056, doi:10.1371/journal.pone.0003056 (2008).

119 DiGiulio, D. B. *et al.* Prevalence and diversity of microbes in the amniotic fluid, the fetal inflammatory response, and pregnancy outcome in women with preterm pre-labor rupture of membranes. *Am J Reprod Immunol* **64**, 38-57, doi:10.1111/j.1600-0897.2010.00830.x (2010).

120 DiGiulio, D. B. *et al.* Microbial invasion of the amniotic cavity in pregnancies with small-for-gestational-age fetuses. *J Perinat Med* **38**, 495-502, doi:10.1515/jpm.2010.076 (2010).
121 Enders, G., Daiminger, A., Bader, U., Exler, S. & Enders, M. Intrauterine transmission and clinical outcome of 248 pregnancies with primary cytomegalovirus infection in relation to gestational age. *J Clin Virol* **52**, 244-246, doi:10.1016/j.jcv.2011.07.005 (2011).
122 Luckey, T. D. *Germfree Life and Gnotobiology*. (Academic Press, 1963).
123 Rasmussen, S. A., Jamieson, D. J., Honein, M. A. & Petersen, L. R. Zika Virus and Birth Defects--Reviewing the Evidence for Causality. *N Engl J Med* **374**, 1981-1987, doi:10.1056/NEJMsr1604338 (2016).
124 Falk, P. G., Hooper, L. V., Midtvedt, T. & Gordon, J. I. Creating and maintaining the gastrointestinal ecosystem: what we know and need to know from gnotobiology. *Microbiol Mol Biol Rev* **62**, 1157-1170, doi:10.1128/MMBR.62.4.1157-1170.1998 (1998).
125 Gordon, H. A. & Pesti, L. The gnotobiotic animal as a tool in the study of host microbial relationships. *Bacteriol Rev* **35**, 390-429 (1971).
126 Hooper, L. V. *et al.* Molecular analysis of commensal host-microbial relationships in the intestine. *Science* **291**, 881-884, doi:10.1126/science.291.5505.881 (2001).
127 Wostman, B. S. *Germfree and Gnotobiotic Animal Models. Background and Applications*. (CRC Press, 1996).
128 Arvidsson, C., Hallen, A. & Backhed, F. Generating and Analyzing Germ-Free Mice. *Curr Protoc Mouse Biol* **2**, 307-316, doi:10.1002/9780470942390.mo120064 (2012).
129 Carter, P. B., Norin, E. & Swennes, A. G. Gnotobiotics and the Microbiome. *The Laboratory Rat*, 827–848, doi:10.1016/B978-0-12-814338-4.00021-0 (2020).
130 Qv, L. *et al.* Methods for Establishment and Maintenance of Germ-Free Rat Models. *Front Microbiol* **11**, 1148, doi:10.3389/fmicb.2020.01148 (2020).
131 Schoeb, T. R. & Eaton, K. A. *Gnotobiotics.* . (Academic Press (Elsevier), 2017).
132 Saffarian, A. *et al.* Crypt- and Mucosa-Associated Core Microbiotas in Humans and Their Alteration in Colon Cancer Patients. *mBio* **10**, doi:10.1128/mBio.01315-19 (2019).
133 Salzberg, S. in *Forbes* (2020).
134 Jost, T., Lacroix, C., Braegger, C. & Chassard, C. Assessment of bacterial diversity in breast milk using culture-dependent and culture-independent approaches. *Br J Nutr* **110**, 1253-1262, doi:10.1017/S0007114513000597 (2013).
135 Treven, P. *et al.* Evaluation of Human Milk Microbiota by 16S rRNA Gene Next-Generation Sequencing (NGS) and Cultivation/MALDI-TOF Mass Spectrometry Identification. *Front Microbiol* **10**, 2612, doi:10.3389/fmicb.2019.02612 (2019).
136 Bihl, S. *et al.* When to suspect contamination rather than colonization - lessons from a putative fetal sheep microbiome. *Gut Microbes* **14**, 2005751, doi:10.1080/19490976.2021.2005751 (2022).
137 Eisenhofer, R. *et al.* Contamination in Low Microbial Biomass Microbiome Studies: Issues and Recommendations. *Trends Microbiol* **27**, 105-117, doi:10.1016/j.tim.2018.11.003 (2019).

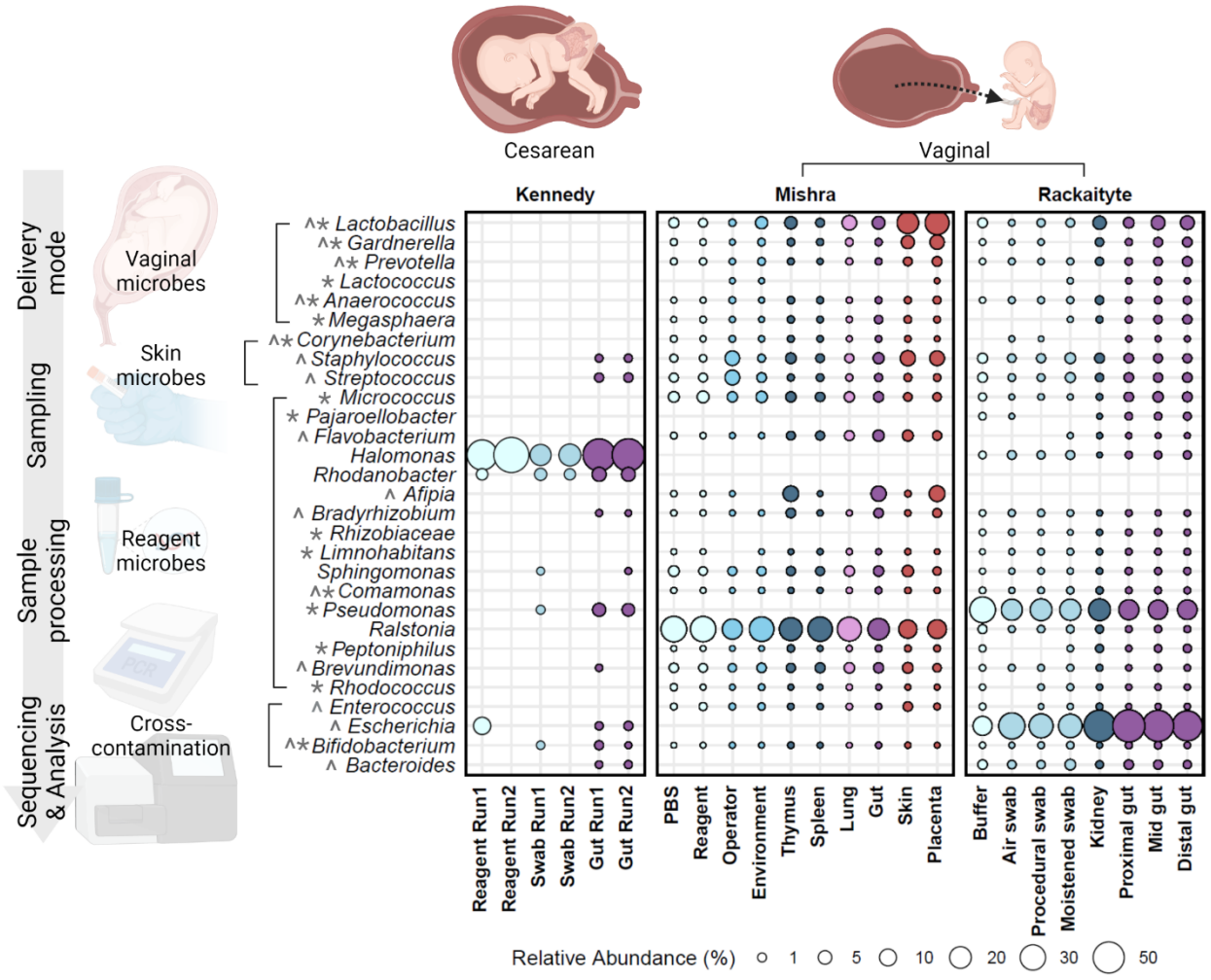

Figure 1. Distribution and mean relative abundance (%) of genera present in fetal samples from three recent studies[28,38,43] investigating the fetal microbiome and their corresponding abundance in control samples. Taxa were selected based on the following criteria: Genera that were cultured from or enriched in fetal samples as described by Mishra *et al.*[38] (indicated by ^) or by Rackaityte *et al.*[43] (indicated by *); all genera detected in fetal samples from Kennedy *et al.*[28]; and the PBS-enriched genus *Ralstonia*[38]. Taxa were grouped by potential source of contamination in agreement with the origin of genera (for skin microbes) and previous studies that characterized sources of contamination[34-36]. For taxonomic data from Rackaityte *et al.*, OTU10 (family *Micrococcaceae*) was manually assigned to the genus *Micrococcus* as in the original publication. Publicly available unfiltered relative abundance data associated with each publication were

merged into a single phyloseq object (RRID:SCR_01380). Amplicon Sequence Variants (ASVs) were grouped at the genus level. The mean relative abundance of each genus was calculated for each sample type within each study and plotted in R (tidyverse, ggplot2; RRID:SCR_014601). Dot size corresponds to the mean relative abundance of each genus by sample type and study (mean relative abundances <0.0001% were excluded). Dots are colored by sample type: reagent controls in lightest blue (Mishra: PBS n=42, Reagent n=23; Rackaityte: Buffer n=11; Kennedy Reagent n=2); sampling negatives in light blue (Kennedy: Swab n=1; Rackaityte: Air swab n=19; Procedural swab n=16; Moistened swab n=17) and environmental negatives in sky blue (Mishra: Environment n=47, Operator n=12), internal controls in dark blue (Mishra: Thymus n=27, Spleen n=12; Rackaityte: Kidney n=16), fetal lung in pink (Mishra, n=25), fetal gut in purple (Kennedy: n=20; Mishra: n=44; Rackaityte: Proximal n=41, Mid n=45, Distal n=42), and external tissues in red (Mishra: Skin n=35, Placenta n=16).

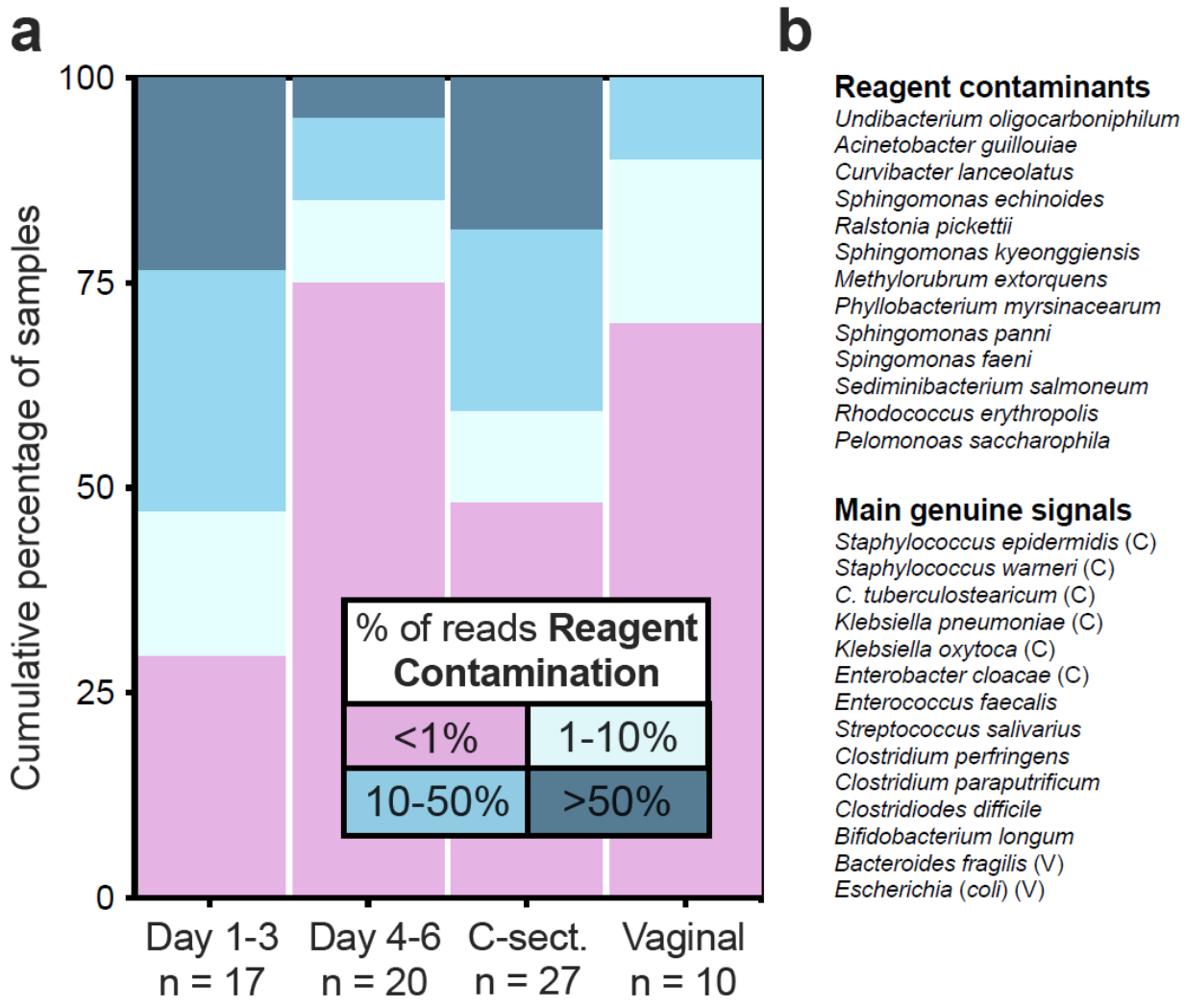

Figure 2. Reagent contamination in meconium samples of extremely premature infants. a) Representation of the % of reagent contamination in the first meconium of extremely premature infants in relation to the day of procurement of said samples (Day 1-3 or Day 4-6) or in regard to the mode of delivery (C-section or Vaginal). Colors indicate the percentage of reagent contamination reads (legend on top). The day of procurement is significantly correlated with the % of reagent contamination reads ($p = 0.005$ MW-U test or $p = 0.01$ Spearman rho test) and the mode of delivery shows a trend ($p = 0.07$ MW-U test). The number of samples is noted below each category (n). **b**) Lists of reagent contaminants shown together in **Figure 2a** (top) and of the most abundant sample-associated-signals and their association (or lack thereof due to limited size of cohort) with vaginal (V) or C-section (C) delivery (bottom).

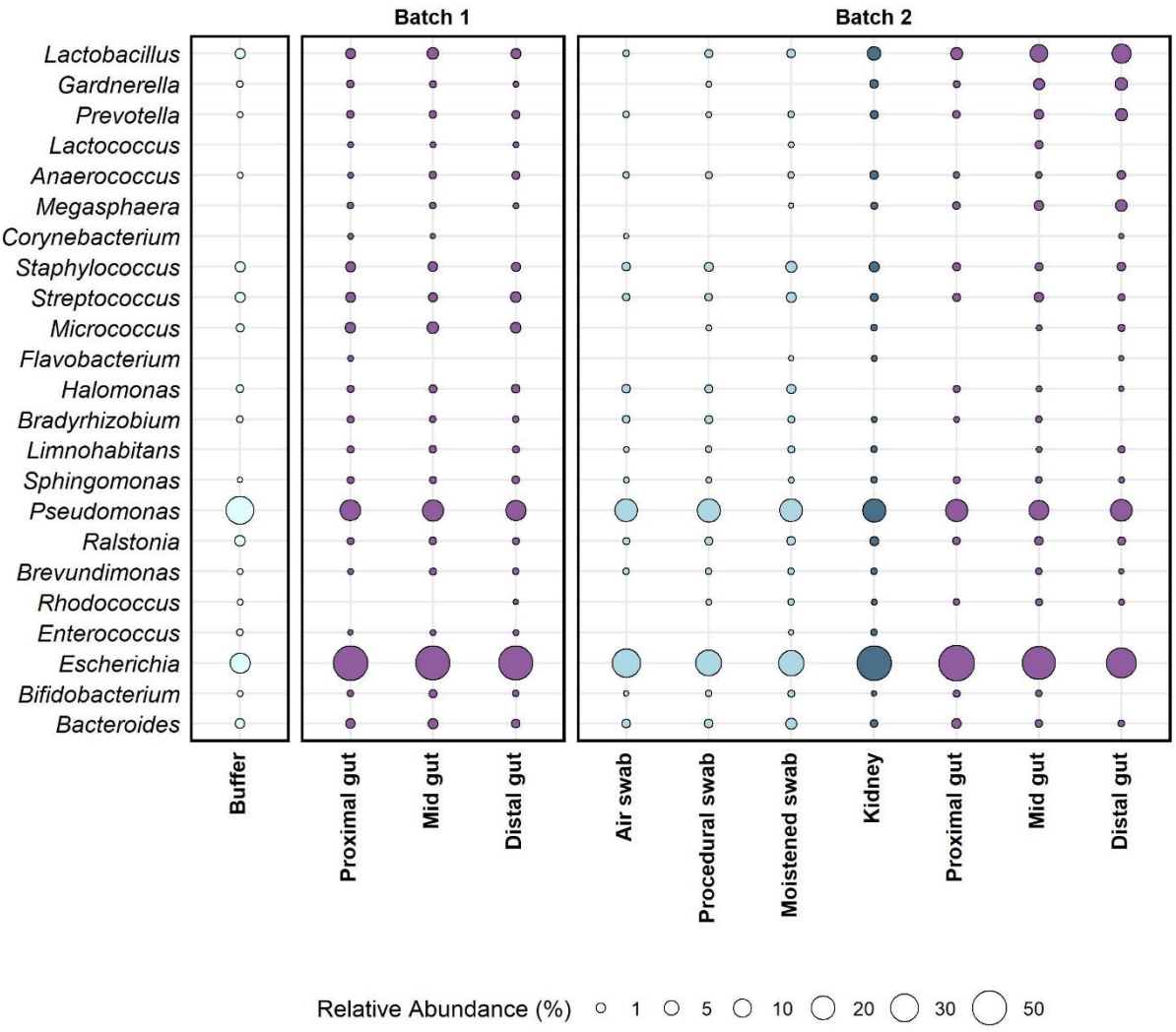

Figure 3. Distribution and mean relative abundance (%) of genera present in fetal and control samples from Rackaityte *et al.*[43] by batch as defined by *Rackaityte et al.*[37]. Dominant taxa were manually selected as described in Fig. 1. For taxonomic data OTU10 (family *Micrococcaceae*) was manually assigned to the genus *Micrococcus* as in the original publication[43]. Publicly available unfiltered relative abundance data associated with each publication were merged into a single phyloseq object (RRID:SCR_01380). ASVs were grouped at the genus level. The mean relative abundance of each genus was calculated for each sample type within each batch and plotted in R (tidyverse, ggplot2; RRID:SCR_014601). Dot size corresponds to the mean relative

abundance of each Genus by sample type and batch. Dots are coloured by sample type: reagent controls in lightest blue (Buffer), sampling negatives in light blue, internal controls in dark blue (Kidney), and fetal gut in purple.