

High-throughput Experimental and Computational Studies of Bacterial Evolution



Lars Barquist
Queens' College
University of Cambridge

A thesis submitted for the degree of

Doctor of Philosophy

XX August 2013

Declaration

HIGH-THROUGHPUT EXPERIMENTAL AND COMPUTATIONAL STUDIES OF BACTERIAL EVOLUTION

The work presented in this dissertation was carried out at the Wellcome Trust Sanger Institute between October 2009 and August 2013. This dissertation is the result of my own work and includes nothing which is the outcome of work done in collaboration except where specifically indicated in the text. This dissertation does not exceed the limit of 60,000 words as specified by the Faculty of Biology Degree Committee. This dissertation has been typeset in 12pt Computer Modern font using L^AT_EX according to the specifications set by the Board of Graduate Studies and the Faculty of Biology Degree Committee. No part of this dissertation or anything substantially similar has been or is being submitted for any other qualification at any other university.

Contents

Declaration	i
Contents	iii
List of Figures	v
List of Tables	vii
List of Symbols	xii
1 Querying bacterial genomes with transposon-insertion sequencing	1
1.1 Introduction	1
1.2 Protocols	4
1.2.1 Transposon mutagenesis	5
1.2.2 Pool construction	6
1.2.3 Enrichment of transposon-insertion junctions	6
1.2.4 Sequencing	7
1.3 Reproducibility, accuracy, and concordance with previous methods	8
1.4 Identifying gene requirements	10
1.4.1 Gene essentiality	10
1.4.2 Applications of transposon-insertion sequencing	11
1.5 Determining conditional gene requirements	12
1.6 Monitoring ncRNA contributions to fitness	14
1.7 Limitations	17
1.8 The future of transposon-insertion sequencing	17

2 A comparison of dense transposon insertion libraries in the <i>Salmonella</i> serovars Typhi and Typhimurium	19
2.1 Introduction	19
2.1.1 The genus <i>Salmonella</i>	20
2.1.2 Host adaptation and restriction	22
2.1.3 Serovars Typhi and Typhimurium	23
2.2 Materials and Methods	26
2.2.1 Strains	26
2.2.2 Annotation	26
2.2.3 Creation of <i>S. Typhimurium</i> transposon mutant library	26
2.2.4 DNA manipulations and sequencing	27
2.2.5 Sequence analysis	27
2.2.6 Statistical analysis of required genes	27
2.3 Results and Discussion	28
2.3.1 TraDIS assay of every <i>Salmonella</i> Typhimurium protein-coding gene	28
2.3.2 Cross-species comparison of genes required for growth	30
2.3.3 Serovar-specific genes required for growth	34
2.3.4 TraDIS provides resolution sufficient to evaluate ncRNA contributions to fitness	45
2.3.5 sRNAs required for competitive growth	49
2.4 Conclusions	51
Published Works	55
References	57

List of Figures

1.1	Transposon-insertion sequencing protocols	4
1.2	Applications of transposon-insertion sequencing to non-coding RNAs	16
2.1	Genomic acquisitions in the evolution of the <i>Salmonellae</i>	21
2.2	The distribution of gene-wise insertion indexes in <i>S. Typhi</i>	29
2.3	Genome-wide transposon mutagenesis of <i>S. Typhimurium</i>	31
2.4	Comparison of required genes	32
2.5	Comparison of cell surface operon structure and requirements	40
2.6	H-NS enrichment across the SPI-2 locus	41
2.7	Proposed differences in sRNA utilization	50

List of Tables

1.1	Summary of transposon-insertion sequencing studies to date	3
2.1	Core genome functions in <i>S. Typhimurium</i>	34
2.2	Phage elements in <i>S. Typhimurium</i>	35
2.3	Phage elements in <i>S. Typhi</i>	36
2.4	Genes uniquely required in <i>S. Typhimurium</i>	38
2.5	Candidate required genes affected by H-NS binding in <i>S. Typhimurium</i> .	42
2.6	Genes uniquely required in <i>S. Typhi</i>	44
2.7	Candidate required ncRNAs	48

List of Symbols

Roman Symbols

A, C, G, T, U	Adenine, Cytosine, Guanine, Thymine, Uracil
Fe(II)	Ferrous iron
Fe(III)	Ferric iron

Greek Symbols

λ	Phage lambda
σ^E	σ^{24} , extracytoplasmic stress sigma factor
σ^S	σ^{38} , starvation/stationary phase sigma factor

Amino Acids

Ala, A	Alanine
Arg, R	Arginine
Asn, N	Asparagine
Asp, D	Aspartic acid (Aspartate)
Cys, C	Cystine
Gln, Q	Glutamine
Glu, E	Glutamic acid (Glutamate)

Gly, G	Glycine
His, H	Histidine
Ile, I	Isoleucine
Leu, L	Leucine
Lys, K	Lysine
Met, M	Methionine
Phe, F	Phenylalanine
Pro, P	Proline
Ser, S	Serine
Thr, T	Threonine
Trp, W	Tryptophan
Tyr, Y	Tyrosine
Val, V	Valine

Acronyms and Abbreviations

BALB	Bagg albino (mouse)
bp	Base pair
CCD	Charge-coupled device
CDP	Cytidine diphosphate glucose
cI	Clear 1 (λ repressor protein)
ddNTP	dideoxynucleotide
DeADMAn	Designer microarrays for defined mutant analysis
DNA	Deoxyribonucleic acid

dNTP	deoxynucleotide
EHEC	Enterohemorrhagic <i>Escherichia coli</i>
FASTA	Fast alignment
FMN	Flavin mononucleotide
FPR	False positive rate
GEBA	Genomic encyclopedia of bacteria and archaea
GTM	Global transposon mutagenesis
HIRAN	HIP116, Rad5p N-terminal
HITS	High-throughput insertion tracking by deep sequencing
INSeq	Insertion sequencing
LEE	Locus of enterocyte effacement
LLR	Log_2 -likelihood ratios
MATT	Microarray tracking of transposon mutants
ncRNA	non-coding RNA
NOGD	Nonorthologous gene displacement
OMP	Outer membrane protein
ORF	Open reading frame
PAGE	Polyacrylamide gel electrophoresis
PCR	Polymerase chain reaction
PPV	Positive predictive value
RNA	Ribonucleic acid
RNA-seq	RNA sequencing

RNase	Ribonuclease
SPI	<i>Salmonella</i> pathogenicity island
SPV	<i>Salmonella</i> plasmid virulence (genes)
sRNA	Bacterial small RNA
SRP	Signal recognition particle
STM	Signature-tagged mutagenesis
T3SS	Type III secretion system
tmRNA	Transfer-messenger RNA
Tn-seq	Transposon mutagenesis and sequencing
TraDIS	Transposon directed insertion sequencing
TraSH	Transposon site hybridization
tRNA	Transfer RNA

Chapter 1

Querying bacterial genomes with transposon-insertion sequencing

This chapter is an expansion of the previously published article “Approaches to querying bacterial genomes using transposon-insertion sequencing” (Barquist, Boinett, and Cain, 2013). Amy K. Cain and Christine J. Boinett (Pathogen Genomics, Wellcome Trust Sanger Institute) contributed to the research of the original article. All final language is my own.

1.1 Introduction

A common approach to identifying genomic regions involved in survival under a particular set of conditions is to screen large pools of mutants simultaneously. This can be done with defined mutants (Baba et al., 2006; Hobbs, Astarita, and Storz, 2010); however, the construction of defined mutant libraries is labor-intensive and requires accurate genomic annotation, which can be particularly difficult to define for non-coding regions. An alternative to defined libraries is the construction and analysis of random transposon-insertion libraries. The original application of this method used DNA hybridization to track uniquely tagged transposon-insertions in *Salmonella enterica* serovar Typhimurium over the course of BALB/c mouse infection (Hensel, Shea, Gleeson, et al., 1995). DNA hybridization was eventually superseded by methods that used microarray detection of the genomic DNA flanking insertion sites, variously known as TraSH, MATT, and DeADMan

(reviewed in Mazurkiewicz et al., 2006). However, these methods suffered from many of the problems microarrays generally suffer from: difficulty detecting low-abundance transcripts, mis-hybridization, probe saturation, and difficulty identifying insertion sites precisely.

The application of high-throughput sequencing to the challenge of determining insertion location and prevalence solves many of these problems. Interestingly, the first application of transposon-insertion sequencing, developed by Hutchison et al. (1999), actually predates the development of microarray-based methods. However, this was applied to libraries of only approximately 1000 transposon mutants in highly reduced *Mycoplasma* genomes, and the difficulty of sequencing at the time prevented wide spread adoption or high resolution. Modern high-throughput sequencing technology allows the methods discussed in this chapter to routinely monitor as many as one million mutants simultaneously in virtually any genetically tractable microorganism.

Table 1.1: Summary of transposon-insertion sequencing studies to date. Columns: 1) study reference, 2) organism mutagenized, 3) number of mutants generated, 4) insertion density, 5) brief description of the application, 6) transposon used, 7) method name coined, if any.

Study	Organism	Total Mutants	Density	Application	Tn used	Name Coined
Hutchison et al. (1999)	<i>M. genitalium</i> <i>M. pneumoniae</i>	1291 918	1/850 bp 1/850 bp	Gene requirements	Tn4001	GTM
Goodman, McNulty, et al. (2009)	<i>B. thetaiotaomicron</i>	2 X 35,000	1/182 bp	Establishment in a murine model of the human gut	Mariner	INSeq
Gawronski et al. (2009)	<i>H. influenzae</i>	75,000	1/32 bp	Prolonged survival in murine lung	Mariner	HITS
Opijken, Bodl, and Camilli (2009)	<i>S. pneumoniae</i>	6 x 25,000	1/91 bp	Transcriptional regulation and carbohydrate transport	Mariner	Tn-seq
Langridge et al. (2009)	<i>S. Typhi</i>	1.1 million	1/13 bp	Gene requirements, bile tolerance	Tn5	TraDIS
Gallagher, Shendure, and Manoil (2011)	<i>P. aeruginosa</i>	100,000	1/65 bp	Tobramycin resistance	Mariner	Tn-seq (circle method)
Eckert et al. (2011)	<i>E. coli</i>	19 x 95	N/A	Colonization of bovine intestinal tract; retrospective re-evaluation of a STM study	Tn5	-
Christen et al. (2011)	<i>C. crescentus</i>	800,000	1/8 bp	Genomic requirements	Tn5	-
Griffin et al. (2011)	<i>M. tuberculosis</i>	2 X 100,000	1/120 bp	Gene requirements and cholesterol utilization	Mariner	-
Khatiwara et al. (2012)	<i>S. Typhimurium</i>	16,000	1/610 bp	Bile, starvation, and heat tolerance	Tn5	-
Mann et al. (2012)	<i>S. pneumoniae</i>	9,000-24,000	Varying	Determining roles of sRNAs in pathogenesis	Mariner	-
Opijken and Camilli (2012)	<i>S. pneumoniae</i>	4,000 - 30,000	Varying	Stress response and metabolism <i>in vitro</i> and murine <i>in vivo</i> colonization	Mariner	-
Brutinel and Gralnick (2012)	<i>S. oneidensis</i>	50,000	1/191 bp	Gene requirements and metabolism	Mariner	-
Zhang et al. (2012)	<i>M. tuberculosis</i>	2 x 100,000	1/120 bp	Genomic requirements	Mariner	-
Klein et al. (2012)	<i>P. gingivalis</i>	N/A	1/43 bp	Gene requirements	Mariner	-
Pickard, Kingsley, et al. (2013)	<i>S. Typhi</i>	1.1 million	1/13 bp	Bacteriophage infection	Tn5	-
Barquist, Langridge, et al. (2013)	<i>S. Typhi</i>	1.1million	1/13 bp	Comparison of genomic requirements between two <i>Salmonella</i> serovars	Tn5	-

1.2 Protocols

Several methods were developed concurrently for high-throughput sequencing of transposon-insertion sites: TraDIS (Langridge et al., 2009), INSeq (Goodman, McNulty, et al., 2009), HITS (Gawronski et al., 2009), and Tn-seq (Opijken, Bodi, and Camilli, 2009) followed by Tn-seq Circle (Gallagher, Shendure, and Manoil, 2011) and refinements to the INSeq protocol (Goodman, M. Wu, and Gordon, 2011). All of these protocols follow the same basic workflow with minor variations (see Figure 1.1; Table 1.1): transposon mutagenesis and construction of pools of single insertion mutants; enrichment of transposon-insertion junctions; and finally, in some protocols a purification step either precedes or follows PCR enrichment before sequencing.

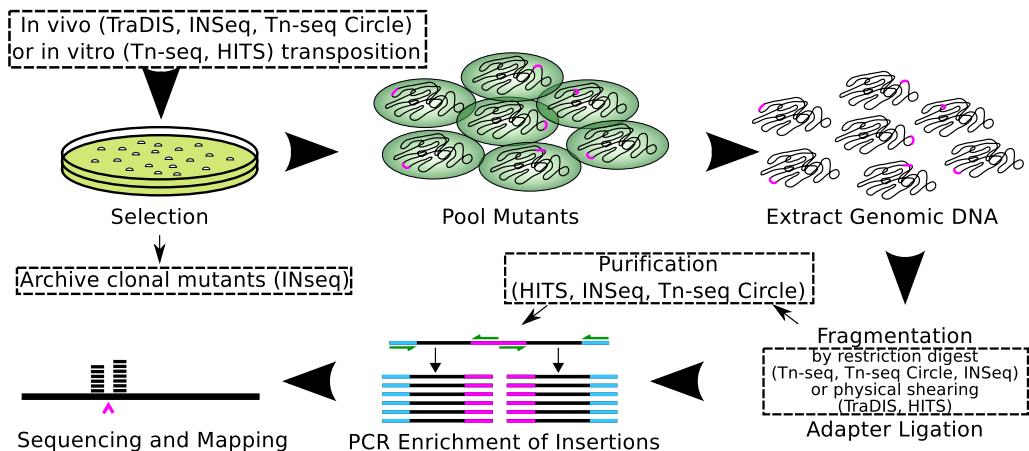


Figure 1.1: Transposon-insertion sequencing protocols. An illustration of the workflow typical of transposon-insertion sequencing protocols. Transposons are represented by pink lines, sequencing adaptors by blue, genomic DNA by black, and PCR primers by green. Mutants are generated through either *in vivo* or *in vitro* transposition and subsequent selection for antibiotic resistance. These mutants are pooled, and optionally competed in test conditions, then genomic DNA is extracted and fragmented by restriction digest or physical shearing. Sequencing adaptors are ligated, some protocols then perform a step to purify fragments containing transposon insertions, and PCR with transposon- and adapter-specific primers is used to specifically enrich for transposon-containing fragments. The fragments are then sequenced and mapped back to a reference genome to uniquely identify insertion sites with nucleotide-resolution. Dashed boxes indicate steps which differ between protocols.

1.2.1 Transposon mutagenesis

Most studies have used either Tn5 or Mariner transposon derivatives. Tn5 originated as a bacterial transposon which has been adapted for laboratory use. Large-scale studies have shown that Tn5, while not showing any strong preference for regional GC-content, does have a weak preference for a particular insertion motif (Shevchenko et al., 2002; Adey et al., 2010; Green et al., 2012). Transposon-insertion sequencing studies performed with Tn5 transposons in *S. enterica* serovars have reported a slight bias towards AT-rich sequence regions (Langridge et al., 2009; Barquist, Langridge, et al., 2013). However, this preference does not appear to be a major obstacle to analysis given the extremely high insertion densities obtained with this transposon (Langridge et al., 2009; Christen et al., 2011; Barquist, Langridge, et al., 2013) (see Table 1.1). Additionally, Tn5 has been shown to be active in a wide range of bacterial species, though the number of transformants obtained can vary significantly depending on the transformation efficiency of the host.

Mariner *Himar1* transposons on the other hand originate from eukaryotic hosts and have an absolute requirement for TA bases at their integration site (Lampe, Grant, and Robertson, 1998; Rubin et al., 1999), with no other known bias besides a possible preference for bent DNA (Lampe, Grant, and Robertson, 1998). This can be a disadvantage in that it limits the number of potential insertion sites, particularly in GC-rich sequence. However, this specificity can also be used in the prediction of gene essentiality in near-saturated libraries: as every potential integration site is known and the probability of integration at any particular site can be assumed to be roughly equal, it is straight-forward to calculate the probability that any particular region lacks insertions by chance. *Himar1* transposition can also be conducted *in vitro* in the absence of any host factors (Lampe, Churchill, and Robertson, 1996), and inserted transposons can then be transferred to the genomes of naturally transformable bacteria through homologous recombination (Johnsborg, Eldholm, and Havarstein, 2007). This can be advantageous when working with naturally transformable bacteria with poor electroporation efficiency (Gawronski et al., 2009; Opijnen, Bodi, and Camilli, 2009). It is worth noting that Tn5 is also capable of transposition *in vitro* (Goryshin and Reznikoff, 1998), and could potentially be used to increase insertion density and hence the resolution of the assay, particularly in GC-rich genomic regions.

1.2.2 Pool construction

Once mutants have been constructed, they are plated on an appropriate selective media for the transposon chosen, and colonies are counted, picked, and pooled. A disadvantage of this is that the mutants must be recreated for follow up or validation studies. Goodman et al. introduced a clever way around this in the INSeq protocol: by individually archiving mutants, then sequencing combinatorial mutant pools it is possible to uniquely characterize $2n$ insertion mutants by sequencing only n pools (Goodman, McNulty, et al., 2009). Each mutant is labelled with a unique binary string that indicates which pools it has been added to. These binary strings can then be reconstructed for each insertion observed in these pools by recording their presence or absence in sequencing data, providing a unique pattern relating insertions to archived mutants. The authors control false identifications due to errors in sequencing by requiring that each binary label have a minimum edit distance to every other label, allowing for a robust association of labels with insertions despite sometimes noisy sequencing data. As a proof of concept, the authors were able to identify over 7,000 *Bacteroides thetaiotaomicron* mutants from only 24 sequenced pools. This effectively uses methods for the generation of random transposon pools to rapidly generate defined mutant arrays, though it is heavily dependent on liquid-handling robotics.

1.2.3 Enrichment of transposon-insertion junctions

Once pools have been constructed they are grown in either selective or permissive conditions, depending on the experiment, and then genomic DNA is extracted. Fragmentation proceeds either through restriction digestion in the case of transposons modified to contain appropriate sites (Goodman, McNulty, et al., 2009; Opijken, Bodi, and Camilli, 2009; Gallagher, Shendure, and Manoil, 2011) or via physical shearing (Langridge et al., 2009; Gawronski et al., 2009), then sequencing adapters are ligated to the resulting fragments. PCR is performed on these fragments using a transposon-specific primer and a sequencing adapter-specific primer to enrich for fragments spanning the transposon-genomic DNA junction.

Some protocols purify fragments containing transposon insertions using biotinylated primers (Gallagher, Shendure, and Manoil, 2011; Goodman, M. Wu, and Gordon, 2011) or PAGE (Goodman, McNulty, et al., 2009) before and/or after PCR enrichment. The

purification step from the Tn-seq Circle protocol is particularly unusual in that restriction digested fragments containing transposon sequence are circularized before being treated with an exonuclease that digests all fragments without transposon insertions, theoretically completely eliminating background (Gallagher, Shendure, and Manoil, 2011). Given the success of protocols that do not include a purification step and the lack of systematic comparisons, it is currently unclear whether including one provides any major advantages.

1.2.4 Sequencing

The protocol steps described so far are broadly similar to those used in microarray-based studies of transposon mutant pools. The major advancement that has driven transposon-insertion sequencing has been the recent development of second generation DNA sequencing technologies. For 30 years, DNA sequencing was dominated by dideoxynucleotide, or Sanger, sequencing, first described by Sanger, Nicklen, and Coulson (1977). Sanger sequencing requires a clonal population of template DNA molecules, to which a primer and a full complement of four deoxynucleotides (dNTPs) and a single species of dideoxynucleotide (ddNTP) are added. DNA polymerase is then used to perform rounds of DNA extension, with ddNTPs stochastically terminating the reaction, before the resulting fragments are denatured and separated with gel electrophoresis. By running four such reactions with each species of ddNTP, the sequence of the template molecule can be determined by reading off bands on the gel. A number of advancements progressively improved the throughput and decreased the cost of Sanger sequencing, including the substitution of capillary electrophoresis for gel electrophoresis and the use of fluorescently labelled ddNTP (fluorescent dye-terminator sequencing) enabling sequencing in a single reaction. However, even with these advances the throughput of Sanger sequencing remained in the range of kilobases of sequence per hour, and costs remained high due to requirements for template cloning and inherent limitations in the technology (Morozova and Marra, 2008).

The development of second generation sequencing technologies in the early-mid 2000's broke these barriers to the adoption of sequencing as a routine experimental technique. These technologies include Roche 454 pyrosequencing, Illumina/Solexa reversible terminator sequencing, and ABI SOLiD parallel sequencing by ligation. While in principle any of these technologies could be applicable to transposon-insertion sequencing, all studies

to date have used Solexa sequencing. Solexa sequencing is similar in principle to Sanger sequencing, with two major innovations: the ability to generate arrayed clonal clusters of template molecules on a glass flow cell (described by Fedurco et al. (2006)) allowing for hundreds of thousands of simultaneous sequencing reactions, and the adoption of reversible dye terminator chemistry (described by Bentley et al. (2008)) which allows for fluorescently labelled terminators to be rapidly stripped of their fluorophore, their termination reversed, and extension continued. By monitoring successive rounds of these hundreds of thousands of parallel sequencing reactions with a CCD camera, the sequence of a large population of template molecules can be determined quickly and simultaneously, leading to current throughputs of megabases to gigabases of sequence per hour. As each resulting read corresponds to a single template molecule, this technology is ideally suited to monitoring populations of transposon mutants, providing an accurate digital count of insertion prevalence.

1.3 Reproducibility, accuracy, and concordance with previous methods

A number of studies have looked at the reproducibility of transposon-insertion sequencing. Multiple studies using different protocol variations have repeatedly shown extremely high reproducibility in the number of insertions per gene (correlations of 90%) in replicates of the same library grown and sequenced independently (Goodman, McNulty, et al., 2009; Opijnen, Bodi, and Camilli, 2009; Gallagher, Shendure, and Manoil, 2011), and good reproducibility (correlations between 70-90%) in independently constructed unsaturated libraries (Opijnen, Bodi, and Camilli, 2009; Opijnen and Camilli, 2012). Opijnen and Camilli (2012) compared traditional 1 X 1 competition experiments between wild-type and mutant *Streptococcus pneumoniae* to results obtained by transposon-insertion sequencing and showed that there was no significant difference in results over a range of tested conditions. The accuracy of transposon-insertion sequencing in determining library composition has also been assessed. Zhang et al. (2012) constructed a library of identified transposon-insertion mutants in known relative quantities, and then were able to recover the relative mutant prevalence with transposon-insertion sequencing. Additionally, by estimating the number of PCR templates prior to enrichment, this study showed that

there is a high correlation between enrichment input and sequencing output.

Two studies have evaluated concordance between results obtained with transposon-insertion sequencing and microarray monitoring of transposon insertions in order to demonstrate the enhanced accuracy and dynamic range of sequencing over previous methods. In the first, 19 libraries of 95 enterohemorrhagic *Escherichia coli* (EHEC) transposon mutants that had previously been screened in cattle using signature-tagged mutagenesis (STM) were pooled and re-evaluated using the TraDIS protocol (Eckert et al., 2011). The original STM study had identified 13 insertions in 11 genes attenuating intestinal colonization in a type III secretion system located in the locus of enterocyte effacement (LEE) (Dziva et al., 2004). By applying sequencing to the same samples, an additional 41 mutations in the LEE were identified, spanning a total of 21 genes. Additional loci outside the LEE which have been previously implicated in intestinal colonization but had not been detected by STM were also reported by TraDIS.

The second study re-evaluated genes required for optimal growth determined by TrASH in *Mycobacterium tuberculosis* (Sassetti, Boyd, and Rubin, 2003; Griffin et al., 2011). The greater dynamic range of sequencing as compared to microarrays allowed easier discrimination between insertions that were truly unviable and those that were only significantly underrepresented. The authors estimate that genes called as required by sequencing in their study are at least 100-fold underrepresented in the pool. In comparison, the threshold in the previous microarray experiment reported genes that had log probe ratios at least 5-fold lower than average between transposon-flanking DNA hybridization and whole genomic DNA hybridization. Additionally, the nucleotide-resolution of insertion sequencing allowed the authors to identify genes which had required regions, likely corresponding to required protein domains (Zhang et al., 2012), but which tolerated insertions in other regions. Altogether the authors increase the set of genes predicted to be required for growth in laboratory conditions in *M. tuberculosis* by more than 25% (from 614 to 774).

1.4 Identifying gene requirements

1.4.1 Gene essentiality

The study of gene essentiality has its roots in evolutionary theory, systems biology, and comparative genomics, and has been instrumental in the development of the emerging discipline of synthetic biology. Koonin summarizes the major scientific motivation behind this line of research succinctly: “When reverse-engineering a complex machine, one basic goal is to draw up a list of essential parts” (Koonin, 2003). The earliest attempt at constructing such a minimal gene set involved a comparison between the first two complete genomes sequenced: *M. genitalium* and *Haemophilus influenzae* (Mushegian and Koonin, 1996). Both of these organisms are pathogens with highly reduced genomes; however, they are derived from distant branches of the bacterial phylogeny being Gram-positive and -negative, respectively. Orthology prediction based on sequence similarity identified 240 genes shared between the two organisms. However, a number of essential pathways were found to be incomplete in this set due to non-orthologous gene displacement (NOGD), and a true minimal gene set was estimated to contain 256 genes. NOGD apparently occurs when an unrelated but functionally analogous gene is introduced in a lineage, and subsequently the ancestral gene is lost. The sequencing of complete genomes has shown that this phenomena is surprisingly wide-spread, and only \sim 60 genes appear to be universally conserved (Koonin, 2003). Rather obviously in hindsight, it appears that gene essentiality is highly dependent on the evolutionary and systems context in which the gene occurs - our essential parts list depends on the machine we wish to build.

Large-scale experimental studies seem to confirm this. A range of approaches have been taken to experimentally determining the ‘essential’ genes of a diverse array of organisms. These include plasmid-insertion mutagenesis in *Bacillus subtilis* (Kobayashi et al., 2003), antisense-mediated gene inactivation in *H. influenzae* (Akerley et al., 2002), transposon mutagenesis in *Pseudomonas aeruginosa* (Jacobs et al., 2003), and insertion-duplication mutagenesis in *S. enterica* (Knuth et al., 2004). However, the “gold standard” for the determination of gene essentiality is repeated failure to generate targeted single gene deletions. Comprehensive single gene deletion libraries have been created for the γ -proteobacteria *E. coli* and *Acinetobacter baylyi* (Baba et al., 2006; Berardinis et al., 2008) where λ -red mediated recombineering has simplified the

generation of defined deletions (Datsenko and Wanner, 2000), though the process is still extremely labor-intensive. Typical estimates for essential gene sets determined by these various techniques range from less than 300 to 600 genes, depending on the organism. This variability is likely dependent on a variety of factors, including false positives and negatives due to experimental techniques, the growth conditions of the experiment, intrinsic properties of the cell being manipulated, and accidents of evolution. Now that it has become feasible to synthesize a viable bacterial chromosome (Gibson et al., 2010), a deeper understanding of the factors affecting gene essentiality is the next hurdle on the road to engineering truly synthetic life.

1.4.2 Applications of transposon-insertion sequencing

The earliest application of transposon-insertion sequencing, and indeed the earliest genome-wide experimental study of gene essentiality, was to determine the minimal set of genes necessary for the survival of *Mycoplasma* (Hutchison et al., 1999). This essential genome is of great interest in synthetic and systems biology where it is seen as a foundation for engineering cell metabolism as described previously, and also in infection biology and medicine where it is seen as a promising target for therapies. However, it is important to remember that essentiality is always relative to growth conditions: a biosynthetic gene that is non-essential in a growth medium supplying a particular nutrient may become essential in a medium that lacks it. Traditionally, gene essentiality has been determined in clonal populations (Baba et al., 2006; Jacobs et al., 2003; Glass et al., 2006); since the high-throughput transposon sequencing protocols described here necessarily contain a short period of competitive growth before DNA extraction, many of these studies prefer to refer to the required genome for the particular conditions under evaluation.

Because of this short period of competitive growth, and because many otherwise required genes tolerate insertions in their terminus (Goodman, McNulty, et al., 2009; Griffin et al., 2011; Zomer et al., 2012) or outside essential domains (Zhang et al., 2012) the determination of required genomic regions is not completely straight-forward and a number of approaches have been taken to counter this. These include only calling genes completely lacking insertions as required (Opijken, Bodi, and Camilli, 2009), or determining a cut-off based on the empirical or theoretical distribution of gene-wise

insertion densities (Langridge et al., 2009; Barquist, Langridge, et al., 2013; Griffin et al., 2011; Zomer et al., 2012). Additionally, windowed methods have been developed which can be used to identify essential regions in the absence of gene annotation (Zhang et al., 2012; DeJesus et al., 2013), and have had success in identifying required protein domains, promoter regions, and non-coding RNAs (ncRNAs). The organisms that have been evaluated for gene requirements under standard laboratory conditions are summarized in Table 1.1. In agreement with previous studies (Baba et al., 2006; Jacobs et al., 2003), many required genes identified by transposon-insertion sequencing are involved in fundamental biological processes such as cell division, DNA replication, transcription and translation (Langridge et al., 2009; Goodman, McNulty, et al., 2009; Barquist, Langridge, et al., 2013; Griffin et al., 2011), and many of these requirements appear to be conserved between genera and classes (Barquist, Langridge, et al., 2013; Christen et al., 2011).

However, a recent study defining required gene sets in *Salmonella* serovars (described in detail in the next chapter) has found that phage repressors, necessary for maintaining the lysogenic state of the prophage, are also required (Barquist, Langridge, et al., 2013), even though mobile genetic elements such as phage are usually considered part of the accessory genome. This study also highlights the need for temperance when interpreting the results of high-throughput assays of gene requirements. For example, many genes in *Salmonella* Pathogenicity Island 2 (SPI-2) did not exhibit transposon-insertions, despite clear evidence from directed knockouts showing that these genes are non-essential for viability or growth. Under laboratory conditions, SPI-2 is silenced by the nucleoid-forming protein H-NS (Lucchini et al., 2006; Navarre et al., 2006), which acts by oligomerizing along silenced regions of DNA blocking RNA polymerase access. A previous study has shown that transposon insertion cold spots can be caused by competition between high-density proteins and transposases for DNA (Manna et al., 2007). This suggests that H-NS may be restricting transposase access to DNA, though this has not previously been observed in transposon-insertion sequencing data, and will require additional work to confirm.

1.5 Determining conditional gene requirements

One of the most valuable applications of the transposon-insertion sequencing method is the ability to identify genes important in a condition of interest, by comparing differences

in the numbers of sequencing reads from input (control) mutant pools to output (test) pools that have been subject to passaging in a certain growth condition. Insertion counts are compared from cells in the input pool and those after passage, thereby identifying genes that either enhance or detract from survival and/or growth in the given condition, defined by decreased or increased insertion frequency, respectively. I describe a pipeline for analyzing such experiments in chapter **XX TraDIS PIPELINE CHAPTER HERE**. A further application of this method involves comparing insertions between biologically linked conditions, such as cellular stresses or different stages of a murine infection, to gain insight into complex systems (Opijnen and Camilli, 2012).

So far, transposon-insertion sequencing has been used to investigate a number of interesting biologically relevant conditions: bile tolerance in *S. Typhi* (Langridge et al., 2009) and *S. Typhimurium* (Khatiwara et al., 2012), bacteriophage infection of *S. Typhi* (Pickard, Kingsley, et al., 2013), antibiotic resistance in *P. aeruginosa* (Gallagher, Shendure, and Manoil, 2011), cholesterol utilization in *M. tuberculosis* (Griffin et al., 2011) and survival in number of stress and nutrient conditions in *S. pneumoniae* (Opijnen and Camilli, 2012). Transposon-insertion sequencing of populations passed through murine models have been used to assess genes required to establish the gut commensal *B. thetaiotaomicron* in its niche (Goodman, McNulty, et al., 2009), for *H. influenzae* infection (Gawronski et al., 2009), as well as *S. pneumoniae* responses to two *in vivo* niches - the lung and nasopharynx (Opijnen and Camilli, 2012). A further extension of the method examined double mutant libraries, that is transposon mutant libraries generated in a defined deletion background, to tease apart complex networks of regulatory genes (Opijnen, Bodi, and Camilli, 2009).

Two studies in particular illustrate the power of using transposon-insertion sequencing to identify conditionally required genes. In the first, Goodman, McNulty, et al. (2009) set out to determine the genes necessary for the establishment of the commensal *B. thetaiotaomicron* in a murine model. First, the growth requirements of transposon mutant populations in the cecum of germ-free mice was assessed, and genes required for growth in monoassociation with the host were found to be enriched in functions such as energy production and amino acid metabolism. By further comparing monoassociated transposon mutant libraries with those grown in the presence of three defined communities of human gut-associated bacteria, the authors identified a locus up-regulated by low levels of vitamin B12 that is only required in the absence of other bacteria capable of

synthesizing B12. This showed that the gene requirements of any particular bacterium in the gut are at least partially dependent on the metabolic capabilities of the entire community and emphasizes the importance of testing *in vivo* conditions to complement *in vitro* study.

The second study, conducted by Opijnen and Camilli (2012), aimed to map the genetic networks involved in a range of cellular stress responses in *S. pneumoniae*. Seventeen *in vitro* conditions were tested, including: pH, nutrient limitation, temperature, antibiotic, heavy metal, and hydrogen peroxide stress. Approximately 6% of disrupted genes resulted in increased fitness in some condition, suggesting that some genes are maintained despite being detrimental to the organism under particular conditions. These would be interesting candidates for further functional and evolutionary study, as the maintenance of these genes is presumably highly dependent on the conditions the bacteria faces, and may have implications for our understanding of e.g. gene loss in the process of bacterial host adaptation (Toft and Andersson, 2010). Two additional *in vivo* experiments were performed in a murine model, where cells were recovered from the lung and nasopharynx. Combining this data, over 1,800 genotype-phenotype genetic interactions were identified. These interactions were mapped and pathways identified. Between the two *in vivo* niches, certain stress responses pathways were markedly different. For example, temperature stress produced a distinct response in the lung, compared to the nasopharynx, which is perhaps to be expected as temperature varies greatly between these two sites. By further examining sub-pathways required in the two different niches and comparing them to *in vitro* requirements, the authors were able to draw conclusions regarding the condition *S. pneumoniae* faces when establishing an infection. This comprehensive mapping of genotype-phenotype relationships will serve as an important atlas for further studies of the factors affecting *S. pneumoniae* carriage and virulence.

1.6 Monitoring ncRNA contributions to fitness

To date, four studies (including one described in detail in the next chapter) have used transposon-insertion sequencing to examine the contribution of non-coding RNAs (ncRNAs) and other non-coding regions to organismal fitness (see Table 1.1). Two of these examined requirements for non-coding regions in the relatively under-explored bacterial species *Caulobacter crescentus* (Christen et al., 2011) and *M. tuberculosis* (Zhang

et al., 2012). Both utilized analytical techniques that allowed for the identification of putative required regions in the absence of genome annotation. Twenty-seven small RNAs (sRNAs) had previously been detected in *C. crescentus* (Landt et al., 2008); 6 were found to be depleted in transposon insertions indicating an important role in basic cellular processes. Additionally, the well-characterized ncRNAs tmRNA and RNase P, as well as 29 non-redundant tRNAs were found to be required. An additional 90 unannotated non-disruptable regions were identified throughout the genome, implying an abundance of unexplored functional non-coding sequence.

While the non-coding transcripts of *M. tuberculosis* have been explored more thoroughly than those of *C. crescentus*, most remain functionally uncharacterized, though there are hints that some of these may be involved in pathogenicity (Arnvig and Young, 2012). Using a Mariner transposon-based assay and a windowed statistical analysis that accounted for the distribution of potential TA integration sites, 35 intergenic regions were identified as putatively required in the *M. tuberculosis* genome (Zhang et al., 2012). In common with the *C. crescentus* study, the RNA component of RNase P, required for the maturation of tRNAs, and tmRNA, involved in the freeing of stalled ribosomes, were identified as required (Figure 1.2 A) together with 10 non-redundant tRNAs and potential promoter regions. However, due to the lower overall insertion density and lack of TA sites in some GC-rich regions, there were some regions that could not be assayed and the resolution was limited to 250 bases.

A particularly exciting study has been conducted in *S. pneumoniae* TIGR4 combining RNA-seq with transposon-insertion sequencing (Mann et al., 2012). To identify sRNA loci the authors first sequenced size-select RNA from wild type TIGR4 and three two-component system knockouts, identifying 89 putative sRNAs, 56 of which were novel. Fifteen of these candidates, selected on the basis of high expression and low predicted folding free energy, were assayed for their ability to establish invasive disease in a murine model. Of these 8 sRNA deletions showed a significant attenuation of disease. To more broadly establish the roles of sRNAs in infecting particular organs, transposon insertion libraries were administered directly to the nasopharynx, lungs, or blood of mice, and bacteria were harvested following disease progression. Twenty-six, 28, and 18 sRNAs were found to attenuate infection in the nasopharynx, lung and blood respectively. These results were then validated with targeted deletions of 11 sRNAs (Figure 1.2 B). In addition to establishing the role of sRNAs in *S. pneumoniae* virulence, this study

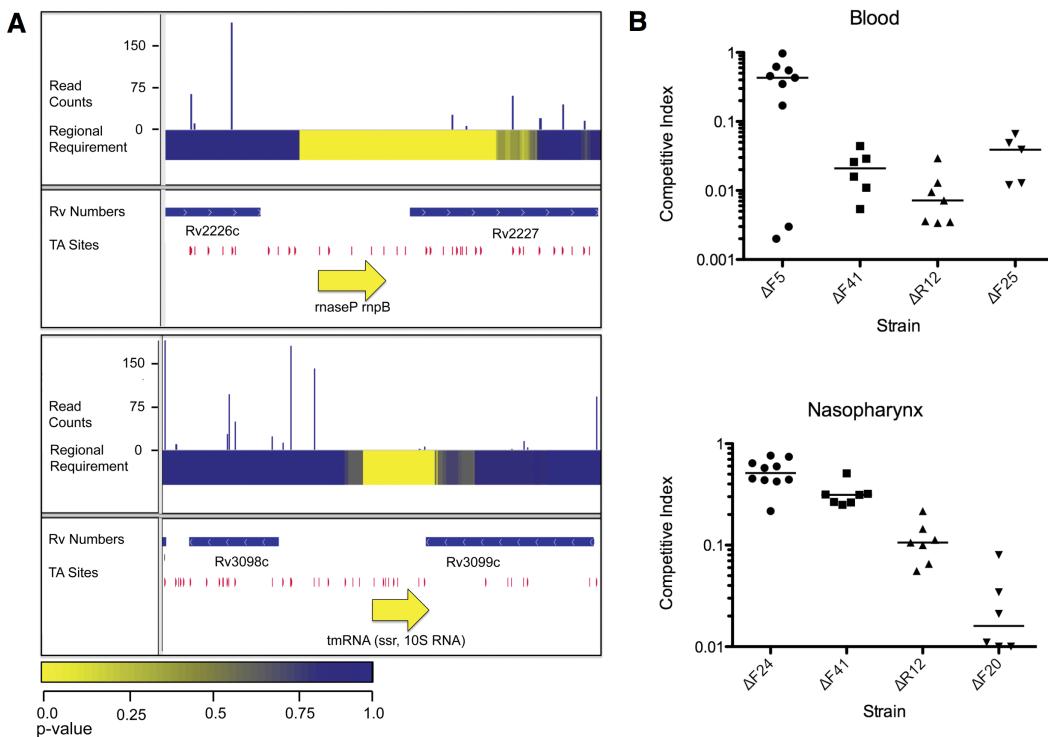


Figure 1.2: Applications of transposon-insertion sequencing to non-coding RNAs.

A) Plots of genomic regions in *Mycobacterium tuberculosis* containing the required non-coding RNAs RNase P (top) and tmRNA (bottom). Tracks, from top to bottom: 1. Histogram of insertion counts, 2. Comprehensive heat-map of requirement of 500-bp windows, 3. Position of annotated genes, 4. Position of TA dinucleotide sites, 5. Position of non-coding RNA. Reproduced from Zhang et al. (2012). B) 1 X 1 competition assays validate attenuating *Streptococcus pneumoniae* sRNA mutants identified by transposon-insertion sequencing. Mice were infected with defined deletions of sRNAs identified as attenuating by Tn-seq and wild type *S. pneumoniae* TIGR4 at the body site indicated and bacterial densities were compared 24 hours post-infection. These plots show the derived competitive index in blood (top) and the nasopharynx (bottom). Each point represents the result of a competition experiment between an sRNA deletion mutant and wild-type TIGR4. A competitive index of 1 indicates equivalent numbers of mutants and wild-type were recovered. Modified from Mann et al. (2012).

illustrated the power of combining RNA-seq and transposon-insertion sequencing to rapidly assign phenotypes to non-coding sequences.

1.7 Limitations

In this chapter, I have largely focused on the potential of transposon insertion sequencing. However, this technology does have a number of important limitations. As discussed previously, requirements for particular nucleotides at insertion sites, such as the TA required by Mariner transposons, or preference for certain sequence composition, such as the AT bias exhibited by Tn5, can limit the density of observed insertions in certain genomic regions. This may impact any down-stream analysis, and can potentially bias results, particularly the determination of gene requirements. Even if this bias has been accounted for, transposon-insertion screens will always over-predict gene requirements in comparison to targeted deletion libraries as discussed previously. However, this over-prediction can be controlled either through careful consideration of known insertion biases as in many Mariner-based studies, or by high insertion densities, such as those achieved in several Tn5-based studies (Table 1.1). Once the library has been created, only regions that have accumulated insertions in the conditions of library creation will be able to be assayed for fitness effects in further conditions. This means that regions that lead to slow growth phenotypes when disrupted in standard laboratory conditions may be difficult to assay in other conditions. Additionally, the dynamic range of fitness effects detected will depend on the complexity of the input library(s). The absence of insertions may be a particular problem for assaying small genomic elements, such as sRNAs or short ORFs. Finally, the validation of hypotheses derived from transposon-insertion sequencing will require the construction of targeted deletions, as individual mutants cannot be recovered from pools unless specialized protocols have been followed during library construction (as in Goodman, McNulty, et al., 2009).

1.8 The future of transposon-insertion sequencing

Transposon-insertion sequencing is a robust and powerful technique for the rapid connection of genotype to phenotype in a wide range of bacterial species. Already, a number

of studies have demonstrated the effectiveness of this method and the results have been far-reaching: enhancing our understanding of basic gene functions, establishing requirements for colonization and infection, mapping complex metabolic pathways, and exploring non-coding genomic dark matter. Due to the range of potential applications of transposon-insertion sequencing, along with the decreasing cost and growing accessibility of next-generation sequencing, I believe that this method will become increasingly common in the near future.

A number of bacterial species have already been subjected to transposon-insertion sequencing (Table 1.1). Microarray-based approaches to monitoring transposon mutant libraries have even been applied to eukaryotic systems (Ross-Macdonald et al., 1999), and similarly transposon-insertion sequencing can potentially be applied to any system where the creation of large-scale transposon mutant libraries is technologically feasible. Recently the Genomic Encyclopedia of Bacteria and Archea (GEBA) (D. Wu et al., 2009) has been expanding our knowledge of bacterial diversity through targeted genomic sequencing of underexplored branches of the tree of life. Applying transposon-insertion sequencing in a comparative manner across the bacterial phylogeny will provide an unprecedented view of the determinants for survival in diverse environments - the next chapter describes a study taking the first steps toward this eventual goal (Barquist, Langridge, et al., 2013). While most transposon-insertion sequencing studies to date have focused on pathogenic bacteria, these techniques could also have applications in energy production, bioremediation, and synthetic biology.

The combination of transposon-insertion sequencing with other high-throughput and computational methods is already proving to be fertile ground for enhancing our understanding of bacterial systems. For instance, by using transposon-insertion sequencing in a collection of relatively simple conditions combined with a computational pathway analysis, Opijnen and Camilli (2012) were able to provide a holistic understanding of the genetic subsystems involved in a complex process such as *S. pneumoniae* pathogenesis. In the future, methods to assay phenotype in a high-throughput manner (Bochner, 2009; Nichols et al., 2011; See also chapter **XX BIOLOG CHAPTER HERE**) may be combined with transposon-insertion sequencing to provide exhaustive simple genotype-phenotype associations with which to understand complex processes in a systems biology framework. I look forward to the adoption of these data sets by the community as an important tool for rapid hypothesis generation.

Chapter 2

A comparison of dense transposon insertion libraries in the *Salmonella* serovars Typhi and Typhimurium

This chapter is a modified version of the previously published article “A comparison of dense transposon insertion libraries in the Salmonella serovars Typhi and Typhimurium” (Barquist, Langridge, et al., 2013). This work is a result of collaboration with Gemma C. Langridge (Pathogen Genomics, Wellcome Trust Sanger Institute), who constructed the Salmonella Typhimurium transposon mutant library and contributed to a draft manuscript. In particular, portions of sections 2.3.1-3 have their genesis in Langridge (2010), though have been significantly elaborated on here.

2.1 Introduction

Salmonella enterica subspecies *enterica* serovars Typhi (*S. Typhi*) and Typhimurium (*S. Typhimurium*) are important, closely related, human pathogens with very different lifestyles. In this chapter, I describe a study comparing dense transposon insertion libraries created in these two serovars. The results of this study demonstrate that orthologous genes can have dramatically different effects on the fitness of recently diverged organisms in rich media. These differences in fitness effects are indicative of changes in the network architecture of the cell which may partially underlie the dramatically different diseases

caused by each organism and their different host ranges. Additionally, *S. Typhimurium* has served as a model organism for the discovery and functional characterization of ncRNAs. Comparing ncRNA requirements between it and a closely related serovar provides a glimpse of the functional evolution of non-coding regulatory networks.

2.1.1 The genus *Salmonella*

Salmonella is a Gram-negative, γ -proteobacterial genus within the order Enterobacterales, consisting of two species: *Salmonella enterica* and *Salmonella bongori*. Based on phylogenetic analyses of 16S and conserved amino acid sequences, it is most closely related to the genera *Escherichia*, *Shigella*, and *Citrobacter* (Paradis et al., 2005; Pham et al., 2007; D. Wu et al., 2009). Molecular clock analyses suggest that *Salmonella* and *Escherichia* shared a common ancestor between 100 and 160 million years ago (Ochman and Wilson, 1987; Doolittle et al., 1996). During the time since their divergence *Escherichia* has become established as a mammalian gut commensal, though multiple independent origins of the *Shigella* and other pathogenic phenotypes within the genus show that a disease phenotype can be developed fairly easily through the horizontal acquisition of virulence determinants and the silencing of anti-virulence loci (Kaper, Nataro, and Mobley, 2004; Prosseda et al., 2012). On the other hand, despite sharing the majority of their genomes with *Escherichia* and having broadly similar metabolic capabilities (AbuOun et al., 2009), the *Salmonellae* exist primarily as pathogens, though are possibly commensal in some reptiles (Mermin et al., 2004; Bauwens et al., 2006).

This difference in phenotype appears to be largely due to the acquisition of virulence determinants which opened new niches to ancestral *Salmonellae* (see figure 2.1). Many of these determinants are encoded on large genomic islands with sizes between \sim 6 and 140 kilobases termed *Salmonella* Pathogenicity Islands (SPIs) (Hensel, 2004). These islands encode a diverse array of virulence-related functions including secretion systems, toxins, antibiotic resistances, and lipopolysaccharide (LPS) and capsular modifications. In particular, the acquisition of SPI-1, encoding a type 3 secretion system (T3SS), and various fimbriae by the ancestral *Salmonella* likely enabled invasion of cells in the intestinal epithelium and escape from competition with other members of the gut microbiota (Bäumler, 1997). *S. bongori* appears to have only acquired a single addition SPI since its divergence from *S. enterica* and likely retains a lifestyle more similar to the

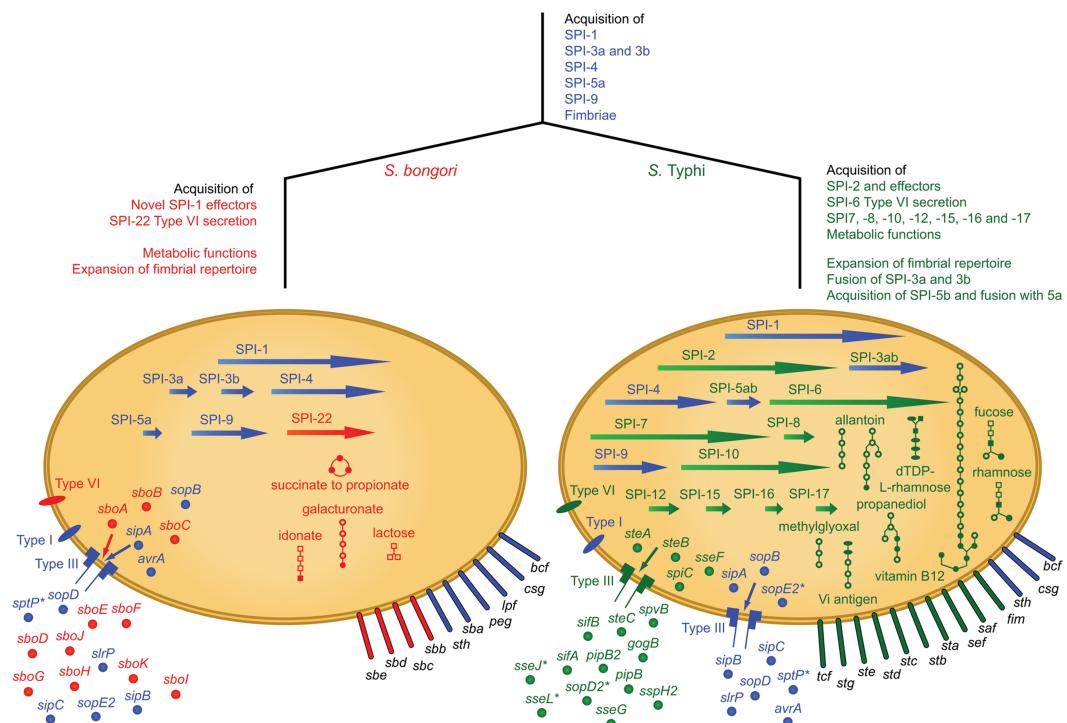


Figure 2.1: Genomic acquisitions in the evolution of the *Salmonellae*. Traits shared by the common ancestor are depicted in blue; those unique to *S. bongori* are shown in red and those unique to *S. enterica* subspecies *enterica* serovar Typhi in green. Arrows, *Salmonella* Pathogenicity Islands (SPIs); extended ovals, fimbriae; circles, effectors; small ovals and needle complexes, secretion systems. Metabolic pathways: lines, enzymatic reactions; open squares, carbohydrates; ovals, pyrimidines; open circles, other substrates; filled shapes, phosphorylated. Novel effectors acquired by *S. bongori* are secreted by the type III secretion system encoded on SPI-1. SPI-3a and 3b carry the same genes in both organisms but are fused into one island in *S. Typhi*. SPI-5a also carries the same genes in both organisms, but a further 3 kb (termed SPI-5b) has fused to SPI-5a in *S. Typhi*. *indicates a pseudogene. Reproduced from Fookes et al. (2011).

ancestral *Salmonella*, though there is evidence for additional adaptation to its niche in the reptilian gut (Fookes et al., 2011).

S. enterica meanwhile has diversified into 6 distinct subspecies: *enterica*, *salamae*, *arizonae*, *diarizonae*, *houtenae*, and *indica*. These subspecies are further divided into over 2000 serovars based on the cell-surface O, flagellar H, and capsular Vi antigens (Grimont and Weill, 2007). The acquisition of SPI-2, involved in survival inside macrophages and an enabling factor for systemic infection (Kuhle and Hensel, 2004; Abrahams and Hensel, 2006), by the ancestral *S. enterica* is thought to have been a driving force in this diversification (Bäumler, 1997). Subspecies besides *enterica* are thought to be primarily restricted to cold-blooded animals (Bäumler, 1997), though sporadic reports of zoonotic disease show these subspecies are capable of transiently infecting the mammalian gut under certain conditions (Mermin et al., 2004; Hilbert et al., 2012). However, here I will be primarily concerned with the subspecies *enterica* and its adaptation to the mammalian, and more specifically human, host.

2.1.2 Host adaptation and restriction

Bacterial adaptation to a pathogenic lifestyle is a complex process involving both the acquisition of virulence factors and gene loss through both passive decay and positive selection (Pallen and Wren, 2007; Prosseda et al., 2012). In the previous section I discussed how the acquisition of SPI-1 and -2, among other factors, have enabled *S. enterica* subspecies *enterica* to establish a niche in the mammalian gut. Access to this new niche has enabled serovars of subspecies *enterica* to explore a range of pathogenic modalities. The most common form of disease caused by *enterica* serovars is a self-limiting gastroenteritis, exemplified by the serovars Typhimurium and Enteriditidis (Santos et al., 2001). These serovars can infect a wide range of mammals and birds, but are only capable of causing serious disease in the very young (Bäumler et al., 1998), and are generally thought to exhibit a phenotype similar to the ancestral *enterica*.

A number of subspecies *enterica* serovars have adapted to causing invasive disease in specific organisms. These include Typhi and Paratyphi in humans, Dublin in cattle, Gallinarum in chickens, Abortusovis in sheep, Choleraesuis in pigs, and Abortusequi in horses. These adaptations appear to be the result of the acquisition of host-specific virulence factors (Bäumler et al., 1998). Interestingly, those serovars associated with the

most severe forms of disease appear to be most highly restricted in terms of host range. This appears to be the result of three processes: positive selection against anti-virulence loci (Pallen and Wren, 2007; Prosseda et al., 2012), and two more passive processes termed “use it or lose it” and “use it, but lose it anyway” by Moran (2004). Selection against anti-virulence loci presumably occurs during host-adaptation, and generally involved the loss of loci that provoke an antigenic response or interfere with the infective process. Once a bacterium has escaped competition in the gut microbiota and gained access to a rich intracellular niche through horizontal acquisitions, the “use it or lose it” principle leads to the loss of metabolic pathways no longer required in this environment presumably due to the lifting of selective pressure for their maintenance. The “use it, but lose it anyway” principle is a consequence of the severe bottleneck imposed by adaptation to a particular host, which will often drastically reduce the effective population size of the bacterium. This can cause fixation of inactivating mutations in potentially beneficial genes simply as an accident of the adaptive process. Together these processes may eventually prevent the bacterium from living independently of its host; particularly extreme examples are *Mycobacterium leprae* with its thousands of inactivated pseudogenes (Cole et al., 2001), *Mycoplasma* species with their highly reduced genomes (Fraser et al., 2001), and most strikingly the endosymbiont-derived mitochondria and plastid organelles (Sagan, 1967; Andersson et al., 1998). While no *Salmonella* serovars appear to have been subject to this degree of genome degradation, it is not unusual for as much as 7% of the protein-coding genes of host-restricted serovars to be inactivated (Parkhill et al., 2001; N. R. Thomson et al., 2008; Holt, Teo, et al., 2009; McClelland, Sanderson, Clifton, et al., 2004).

The serovars of *S. enterica* subspecies *enterica* exhibit a spectrum of pathogenic lifestyles, from low-pathogenicity and wide host range to high-pathogenicity and narrow host range. Recent studies examining host adaptation of Typhimurium strains to immunocompromised populations (Feasey et al., 2012; Okoro et al., 2012) demonstrate that the process of host-adaptation is both on-going and highly relevant to human health. In this study, we have used transposon-insertion sequencing to examine two recently diverged (circa 50,000 years ago (Kidgell et al., 2002)) serovars at extreme ends of this pathogenicity spectrum: Typhi and Typhimurium.

2.1.3 Serovars Typhi and Typhimurium

Salmonella enterica subspecies *enterica* serovars *Typhi* (*S. Typhi*) and *Typhimurium* (*S. Typhimurium*) are important human pathogens with distinctly different lifestyles. *S. Typhi* is host-restricted to humans and causes typhoid fever. This potentially fatal systemic illness affects at least 21 million people annually, primarily in developing countries (Crump, Luby, and Mintz, 2004; Bhutta and Threlfall, 2009; Kothari, Pruthi, and Chugh, 2008), and is capable of colonizing the gall bladder creating asymptomatic carriers; such individuals are the primary source of this human restricted infection, exemplified by the case of “Typhoid Mary” (Soper, 1939). *S. Typhimurium*, conversely, is a generalist, infecting a wide range of mammals and birds in addition to being a leading cause of foodborne gastroenteritis in human populations. Control of *S. Typhimurium* infection in livestock destined for the human food chain is of great economic importance, particularly in swine and cattle (CDC, 2009; Majowicz et al., 2010). Additionally, *S. Typhimurium* causes an invasive disease in mice, which has been used extensively as a model for pathogenicity in general and human typhoid fever specifically (Santos et al., 2001).

Despite this long history of investigation, the genomic factors that contribute to these differences in lifestyle remain unclear. Over 85% of predicted coding sequences are conserved between the two serovars in sequenced genomes of multiple strains (McClelland, Sanderson, Spieth, et al., 2001; Parkhill et al., 2001; Holt, Parkhill, et al., 2008; Deng et al., 2003). The horizontal acquisition of both plasmids and pathogenicity islands during the evolution of the *Salmonellae* is believed to have impacted upon their disease potential. A 100kb plasmid, encoding the *Salmonella* plasmid virulence (SPV) genes, is found in some *S. Typhimurium* strains and contributes significantly towards systemic infection in animal models (Gulig and Curtiss, 1987; Gulig, Danbara, et al., 1993). *S. Typhi* is known to have harbored IncHI1 plasmids conferring antibiotic resistance since the 1970s (Phan et al., 2009), and there is evidence that these strains present a higher bacterial load in the blood during human infection (Wain et al., 1998). Similar plasmids have been isolated from *S. Typhimurium* (Datta, 1962; Holt, N. R. Thomson, et al., 2007; Cain and Hall, 2012). *Salmonella* pathogenicity islands 1 and 2 are common to both serovars, and are required for invasion of epithelial cells (reviewed in Darwin and V. L. Miller (1999)) and survival inside macrophages respectively (Ochman and Groisman,

1996; Shea et al., 1996). *S. Typhi* additionally incorporates SPI-7 and SPI-10, which contain the Vi surface antigen and a number of other putative virulence factors (Pickard, Wain, et al., 2003; Seth-Smith, 2008; Townsend et al., 2001).

Acquisition of virulence determinants is not the sole explanation for the differing disease phenotypes displayed in humans by *S. Typhimurium* and *S. Typhi*; genome degradation is an important feature of the *S. Typhi* genome, in common with other host-restricted serovars such as *S. Paratyphi A* (humans) and *S. Gallinarum* (chickens). In each of these serovars, pseudogenes account for 4-7% of the genome (Parkhill et al., 2001; N. R. Thomson et al., 2008; Holt, Teo, et al., 2009; McClelland, Sanderson, Clifton, et al., 2004). Loss of function has occurred in a number of *S. Typhi* genes that have been shown to encode intestinal colonisation and persistence determinants in *S. Typhimurium* (Kingsley et al., 2003). Numerous sugar transport and degradation pathways have also been interrupted (Parkhill et al., 2001), but remain intact in *S. Typhimurium*, potentially underlying the restricted host niche occupied by *S. Typhi*.

In addition to its history as a model organisms for pathogenicity, *S. Typhimurium* has recently served as a model organism for the elucidation of non-coding RNA (ncRNA) function (Vogel, 2009a). These include cis-acting switches, such as RNA-based temperature and magnesium ion sensors (Waldminghaus et al., 2007; Cromie et al., 2006), together with a host of predicted metabolite-sensing riboswitches. Additionally, a large number of trans-acting small RNAs (sRNAs) have been identified within the *S. Typhimurium* genome (Kröger et al., 2012), some with known roles in virulence (Hebrard et al., 2012). These sRNAs generally control a regulon of mRNA transcripts through an antisense binding mechanism mediated by the protein Hfq in response to stress. The functions of these molecules have generally been explored in either *S. Typhimurium* or *E. coli*, and it is unknown how stable these functions and regulons are over evolutionary time (Richter and Backofen, 2012).

Transposon mutagenesis has previously been used to assess the requirement of particular genes for cellular viability. The advent of next-generation sequencing has allowed simultaneous identification of all transposon insertion sites within libraries of up to 1 million independent mutants (reviewed in Barquist, Boinett, and Cain (2013); see also the previous chapter), enabling us to answer the basic question of which genes are required for *in vitro* growth with extremely fine resolution. By using transposon mutant libraries of this density, which in *S. Typhi* represents on average > 80 unique insertions

per gene (Langridge et al., 2009), shorter regions of the genome can be interrogated, including ncRNAs (Christen et al., 2011). In addition, once these libraries exist, they can be screened through various selective conditions to further reveal which functions are required for growth/survival.

Using Illumina-based transposon directed insertion-site sequencing (TraDIS (Langridge et al., 2009)) with large mutant libraries of both *S. Typhimurium* and *S. Typhi*, we investigated whether these *Salmonellae* require the same protein-coding and non-coding RNA (ncRNA) gene sets for competitive growth under laboratory conditions, and whether there are differences which reflect intrinsic differences in the pathogenic niches these bacteria inhabit.

2.2 Materials and Methods

2.2.1 Strains

S. Typhimurium strain SL3261 contains a deletion relative to the parent strain, SL1344, was used to generate the large transposon mutant library. The 2166bp deletion ranges from 153bp within *aroA* (normally 1284bp) to the last 42bp of *cmk*, forming two pseudogenes and deleting the intervening gene SL0916 completely. For comparison, we utilized our previously generated *S. Typhi* Ty2 transposon library (Langridge et al., 2009).

2.2.2 Annotation

For *S. Typhimurium* strain SL3261, we used feature annotations drawn from the SL1344 genome (EMBL-Bank accession FQ312003.1), ignoring the deleted *aroA*, *ycaL*, and *cmk* genes. We re-analyzed our *S. Typhi* Ty2 transposon library with features drawn from an updated genome annotation (EMBL-Bank accession AE014613.1.) We supplemented the EMBL-Bank annotations with non-coding RNA annotations drawn from Rfam 10.1 (Burge et al., 2013), Sittka et al. (2008), Chinni et al. (2010), Raghavan, Groisman, and Ochman (2011), and Kröger et al. (2012). Selected protein-coding gene annotations were supplemented using the HMMER webserver (Finn, Clements, and Eddy, 2011) and Pfam (Punta et al., 2012).

2.2.3 Creation of *S. Typhimurium* transposon mutant library

S. Typhimurium was mutagenized using a Tn5-derived transposon as described previously (Langridge et al., 2009; a detailed protocol is available in Langridge, 2010). Briefly, the transposon was combined with the EZ-Tn5 transposase (Epicenter, Madison, USA) and electroporated into *S. Typhimurium*. Transformants were selected by plating on LB agar containing 15 µg/mL kanamycin and harvested directly from the plates following overnight incubation. A typical electroporation experiment generated a batch of between 50,000 and 150,000 individual mutants. 10 batches were pooled together to create a mutant library comprising approximately 930,000 transposon mutants.

2.2.4 DNA manipulations and sequencing

Genomic DNA was extracted from the library pool samples using tip-100g columns and the genomic DNA buffer set from Qiagen (Crawley, UK). DNA was prepared for nucleotide sequencing as described previously (Langridge et al., 2009). Prior to sequencing, a 22 cycle PCR was performed as previously described (Langridge et al., 2009). Sequencing took place on a single end Illumina flowcell using an Illumina GAIIG sequencer, for 36 cycles of sequencing, using a custom sequencing primer and 2x Hybridization Buffer (Langridge et al., 2009). The custom primer was designed such that the first 10 bp of each read was transposon sequence.

2.2.5 Sequence analysis

The Illumina FASTQ sequence files were parsed for 100% identity to the 5' 10bp of the transposon (TAAGAGACAG). Sequence reads which matched were stripped of the transposon tag and subsequently mapped to the *S. Typhimurium* SL1344 or *S. Typhi* Ty2 chromosomes using Maq version maq-0.6.8 (Li, Ruan, and Durbin, 2008). Approximately 12 million sequence reads were generated from the sequencing run which used two lanes on the Illumina flowcell. Precise insertion sites were determined using the output from the Maq mapview command, which gives the first nucleotide position to which each read mapped. The number and frequency of insertions mapping to each nucleotide in the appropriate genome was then determined.

2.2.6 Statistical analysis of required genes

The number of insertion sites for any gene is dependent upon its length, so the values were made comparable by dividing the number of insertion sites by the gene length, giving an “insertion index” for each gene. As before (Langridge et al., 2009) the distribution of insertion indices was bimodal, corresponding to the required (mode at 0) and non-required models (See Figure 2.2). We fitted gamma distributions for the two modes using the R MASS library (<http://www.r-project.org>). Log_2 -likelihood ratios (LLR) were calculated between the required and non-required models and we called a gene required if it had an LLR of less than -2, indicating it was at least 4 times more likely according to the required model than the non-required model. “Non-required” genes were assigned for an LLR of greater than 2. Genes falling between the two thresholds were considered “ambiguous” for the purpose of this analysis. This procedure lead to genes being called as required in *S. Typhimurium* when their insertion index was less than 0.020, and ambiguous between 0.020 and 0.027. The equivalent cut-offs for the *S. Typhi* library are 0.0147 and 0.0186, respectively.

We calculated a p-value for the observed number of insertion sites per gene using a Poisson approximation with rate $R = \frac{N}{G}$ where N is the number of unique insert sites (549,086) and G is the number of bases in the genome (4,878,012). The p-value for at least X consecutive bases without an insert site is $e^{(-RX)}$, giving a 5% cut-off at 27 bp and a 1% cut-off at 41 bp.

For every gene g with $n_{g,A}$ reads observed in *S. Typhi* and $n_{g,B}$ reads observed in *S. Typhimurium*, we calculated the log2 fold change ratio $S_{g,A,B} = \log_2((n_{g,A} + 100)/(n_{g,B} + 100))$. The correction of 100 reads smoothes out the high scores for genes with very low numbers of observed reads. We fitted a normal model to the mode +/- 2 sample standard deviations of the distribution of $S_{A,B}$, and calculated p-values for each gene according to the fit. We considered genes with a p-value of 0.05 or less under the normal model to be uniquely required by one serovar.

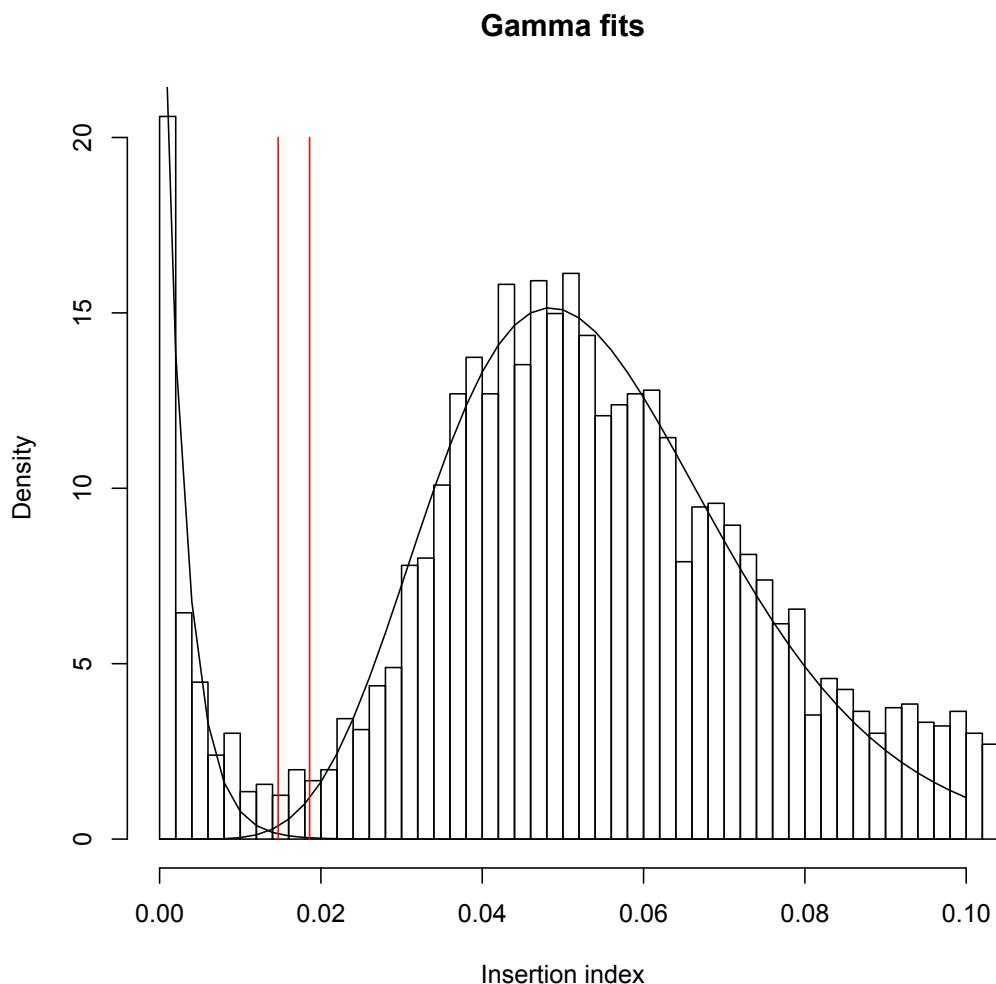


Figure 2.2: The distribution of gene-wise insertion indexes in *S. Typhi*. Bars report the density of genes with insertion indexes within each range, black lines show gamma distributions fitted to the required (left, mode at 0) and non-required (right) peaks, and red lines report associated LLR-based cut-offs for calling gene ambiguity (left) and requirement (right). The distribution of insertion indexes in *S. Typhimurium* is similar, though with a wider separation between the required and non-required peaks due to the higher insertion density attained.

2.3 Results and Discussion

2.3.1 TraDIS assay of every *Salmonella* *Typhimurium* protein-coding gene

Approximately 930,000 mutants of *S. Typhimurium* were generated using a Tn5-derived transposon. 549,086 unique insertion sites were recovered from the mutant library using short-read sequencing with transposon-specific primers. This is a substantially higher density than the 371,775 insertions recovered from *S. Typhi* previously (Langridge et al., 2009). The *S. Typhimurium* library contains an average of one insertion every 9bp, or over 100 unique inserts per gene (figure 2.3). The large number of unique insertion sites allowed every gene to be assayed; assuming random insertion across the genome, a region of 41bp without an insertion was statistically significant ($P < 0.01$). As previously noted in *S. Typhi*, the distribution of length-normalized insertions per gene is bimodal (see supplementary figure 1), with one mode at 0. We interpret genes falling in to the distribution around this mode as being required for competitive growth within a mixed population under laboratory conditions (hereafter “required”). Of these, 57 contained no insertions whatsoever and were mostly involved in core cellular processes (see table 2.1).

There was a bias in the frequency of transposon insertion towards the origin of replication. This likely occurred as the bacteria were in exponential growth phase immediately prior to transformation with the transposon. In this phase of growth, multiple replication forks would have been initiated, meaning genes closer to the origin were in greater copy number and hence more likely to be a target for insertion. We also observed a bias for transposon insertions in A+T rich regions, as was previously observed in the construction of an *S. Typhi* mutant library (Langridge et al., 2009). However, the insertion density achieved is sufficient to discriminate between required and non-required genes easily. As was first seen in *S. Typhi* (Langridge et al., 2009), we observed transposon insertions into genes upstream of required genes in the same operon, suggesting that most insertions do not have polar effects leading to the inactivation of downstream genes.

Analysis of the *S. Typhimurium* mutant library allowed us to identify 353 coding sequences required for growth under laboratory conditions, and 4,112 non-required coding sequences (see [need to put supp data in appendix](#)). We were unable to assign 65 genes to

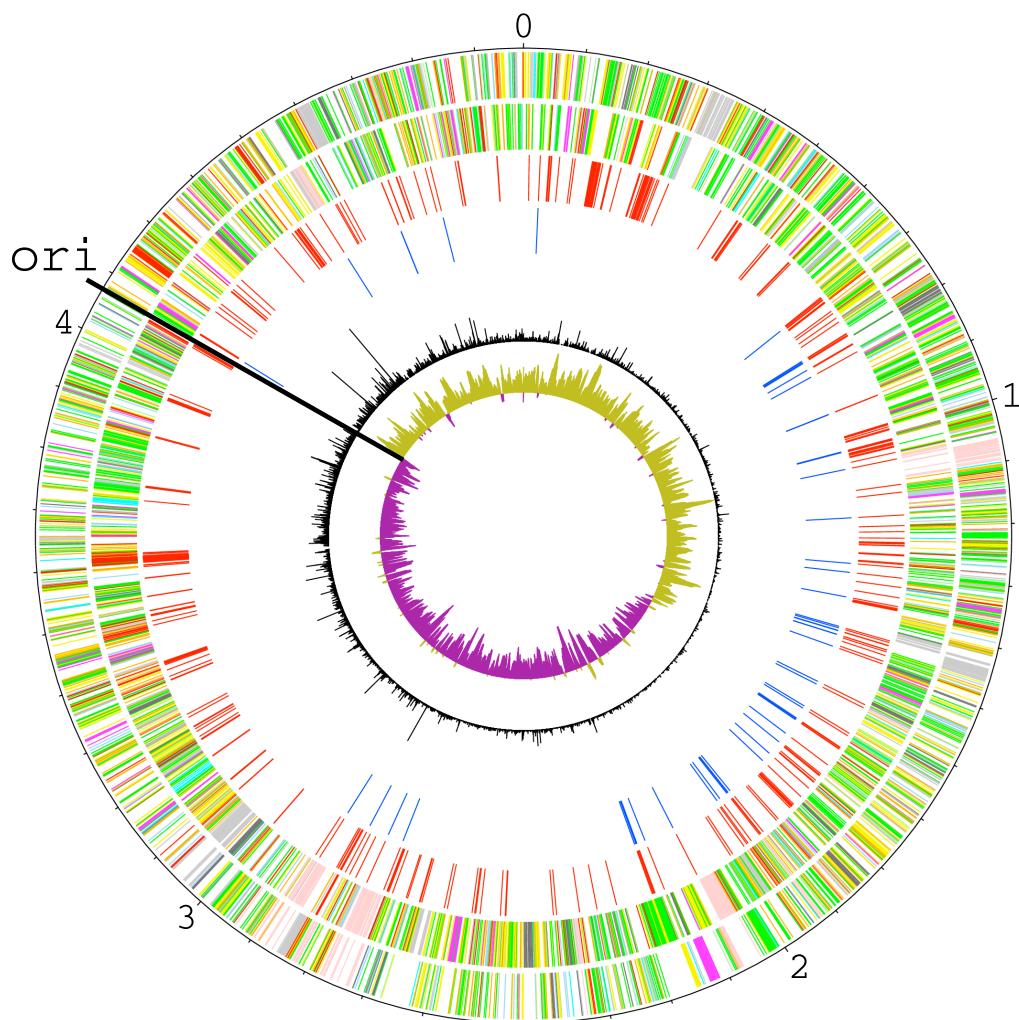


Figure 2.3: Genome-wide transposon mutagenesis of *S. Typhimurium*. A) Circular plot showing gene content, distribution of required genes, and insertion density along the *S. Typhimurium* chromosome. The outer scale is marked in megabases. Circular tracks range from 1 (outer track) to 6 (inner track). Track 1, all forward-strand genes (color-coded according to function: dark blue, pathogenicity/adaptation; black, energy metabolism; red, information transfer; dark green, membranes/surface structures; cyan, degradation of macromolecules; purple, degradation of small molecules; yellow, central/intermediary metabolism; light blue, regulators; pink, phage/IS elements; orange, conserved hypothetical; pale green, unknown function; brown, pseudogenes.); track 2, all reverse-strand genes (color-coded as for forward-strand genes); track 3, *S. Typhimurium* required genes (red); track 4, 56 genes required by *S. Typhimurium* but not by *S. Typhi* (dark blue, see also table 1); track 5, transposon insertion density; track 6, GC bias $((G-C)/(G+C))$, khaki indicates values >1 ; purple <1 .

either the required or non-required category. 60 of these genes, which we will refer to as “ambiguous”, had log-likelihood ratios (LLRs) between -2 and 2. The final 5 unassigned genes had lengths less than 60 bases, and they were removed from the analysis. All other genes contained enough insertions or were of sufficient length to generate credible LLR scores. Thus, every gene was assayed and we were able to draw conclusions for 98.7% of the coding genome in a single sequencing run (figure 2.3).

2.3.2 Cross-species comparison of genes required for growth

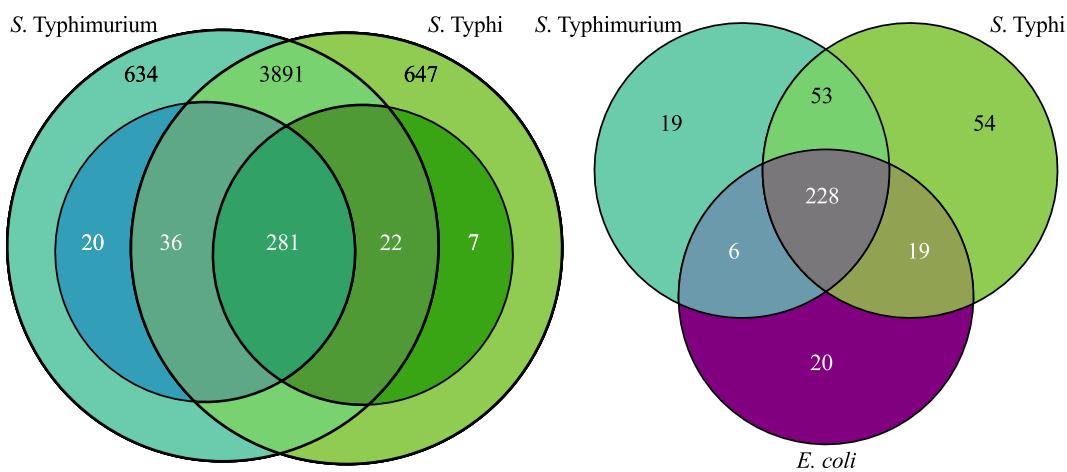


Figure 2.4: Comparison of required genes. Venn diagrams showing (A) the overlap of all genes (outer circles, light colors) and required genes (inner circles, dark colors) between *S. Typhimurium* and *S. Typhi* (excluding genes required in one serovar only which do not have significantly different read-counts). Black numbers refer to all genes, white numbers to required genes. (B) the overlap of all required genes between *S. Typhimurium* (blue), *S. Typhi* (green) and *E. coli* (purple). White numbers refer to genes with Keio essentiality scores ≥ 0.5 .

Gene essentiality has previously been assayed in *Salmonella* Typhimurium using insertion-duplication mutagenesis. Knuth et al. (2004) estimated 490 genes are essential to growth in clonal populations, though 36 of these have subsequently been successfully deleted (Santiviago, Reynolds, et al., 2009). While TrAdIS assays gene requirements after a brief period of competitive growth on rich media, we identify a smaller required set than Knuth et al. (2004) of approximately 350 genes in each serovar, closer to current estimates of approximately 300 essential genes in *E. coli* (Baba et al., 2006).

To demonstrate that TraDIS does identify genes known to have strong effects on growth, as well as to test our predictive power for determining gene essentiality, we compared our required gene sets in *S. Typhimurium* and *S. Typhi* to essential genes determined by systematic single-gene knockouts in the *Escherichia coli* K-12 Keio collection (Baba et al., 2006). We identified orthologous genes in the three data sets by best reciprocal FASTA hits exhibiting over 30% sequence identity over at least 80% of the amino acid sequence. Required orthologous genes identified in this manner share a significantly higher average percent sequence identity with their *E. coli* counterparts than expected for a random set of orthologs, at ~94% identity as compared to ~87% for all orthologous genes. In 100,000 randomly chosen gene sets of the same size as our required set we did not find a single set where the average shared identity exceeded 90%, indicating that required genes identified by TraDIS are more highly conserved at the amino acid level than other orthologous protein coding sequences.

Baba et al. (2006) have defined an essentiality score for each gene in *E. coli* based on evidence from four experimental techniques for determining gene essentiality: targeted knock-outs using λ -red mediated homologous recombination, genetic footprinting (Gerdes et al., 2003; Tong et al., 2004), large-scale chromosomal deletions (Hashimoto et al., 2005), and transposon mutagenesis (Kang et al., 2004). Scores range from -4 to 3, with negative scores indicating evidence for non-essentiality and positive scores indicating evidence for essentiality. Comparing the overlap between essential gene sets in *E. coli*, *S. Typhi*, and *S. Typhimurium*, we find a set of 228 *E. coli* genes which have a Keio essentiality score of at least 0.5 (i.e. there is evidence for gene essentiality; See Figure 2.4.) that have TraDIS-predicted required orthologs in both *S. Typhi* and *S. Typhimurium*, constituting ~85% of *E. coli* genes with evidence for essentiality indicating that gene requirements are largely conserved between these genera. Including orthologous genes that are only predicted to be essential by TraDIS in *S. Typhi* or *S. Typhimurium* raises this figure to nearly 93%. The majority of shared required genes between all three bacteria are responsible for fundamental cell processes, including cell division, transcription and translation. A number of key metabolic pathways are also represented, such as fatty acid and peptidoglycan biosynthesis (Table 2.1). A recent study in the alphaproteobacteria *Caulobacter crescentus* reported 210 shared essential genes with *E. coli*, despite *C. crescentus* sharing less than a third as many orthologous genes with *E. coli* as *Salmonella* serovars (Christen et al., 2011). This suggests the existence of a shared

Table 2.1: Core genome functions in *S. Typhimurium*. Protein-coding genes providing fundamental biological functions in *S. Typhimurium*. Genes in bold are required in *S. Typhi* (log-likelihood ratio (LLR) between required and non-required models < -2; see Methods.) * indicates genes ambiguous in *S. Typhimurium*, having a LLR between -2 and 2.

Biological Process	Sub-process	Required genes	Non-required genes
Cell division		<i>ftsALKQWYZ, minE, mukB, SL2391</i>	<i>ftsHJNX*, minCD, sdiA, cedA, sulA</i>
DNA replication	Polymerases I, II, and III	<i>dnaENQX, holAB</i>	<i>polAB, holCDE</i>
	Supercoiling	<i>gyrAB, parCE</i>	
	Primosome-associated	<i>dnaBCGT, priA, ssb</i>	<i>priB*C, rep</i>
Transcription	RNA polymerase	<i>rpoABC</i>	
	Sigma, elongation, anti- and termination factors	<i>nusBG, rpoDH, rho</i>	<i>nusA, rpoENS</i>
Translation	tRNA-synthetases	<i>alaS, argS, asnS, aspS, cysS, glnS, gltX, glyQS, hisS, ileS, leuS, lysS, metG, pheST, proS, serS, thrS, tyrS, valS</i>	<i>trpS, trpS2</i>
	Ribosome components	<i>rplBCDEFJKLMNPQRSTU VWXY, rpmABCDHI, rpsABCDEF GHijklmnPQST</i>	<i>rplAI, rpmEE2, rpmFHJJ2, rpsOR*U*V</i>
	Initiation, elongation, and peptide chain release factors	<i>fusA, infABC, prfAB, tsf, yrDC</i>	<i>efp, prfCH, selB, tuf</i>
Biosynthetic pathways			
Peptidoglycan		<i>murABCDEFGI</i>	<i>ddl, dlla</i>
Fatty acids		<i>accABCD, fabABDGHIZ</i>	

core of approximately 200 essential proteobacterial genes, with the comparatively rapid turnover of 150 to 250 non-core lineage-specific essential genes.

If we make the simplistic assumption that gene essentiality should be conserved between *E. coli* and *Salmonella*, we can use the overlap of our predictions with the Keio essential genes to provide an estimate of our TraDIS libraries accuracy for predicting that a gene will be required in a clonal population. Of the 2632 orthologous *E. coli* genes which have a Keio essentiality score of less than -0.5 (i.e. there is evidence for gene non-essentiality), only 33 are predicted to be required by TraDIS in both *Salmonella* serovars. *S. Typhi* contains the largest number of genes predicted by TraDIS to be required with *E. coli* orthologs with negative Keio essentiality scores. However, even if we assume these are all incorrect predictions of gene essentiality, this still gives a gene-wise false positive rate (FPR) of ~2.7% (81 out of 2981 orthologs) and a positive predictive value (PPV) of ~75% (247 with essentiality scores greater than or equal to 0.5 out of 328 predictions with some Keio essentiality score.) Under these same criteria the *S. Typhimurium* data set has a lower gene-wise FPR of ~1.6% (51 out of 3122 orthologs) and a higher PPV of ~82% (234 out of 285 predictions as before), as we would expect given the library's higher insertion density. In reality these FPRs and PPVs are only estimates; genes which are not essential in *E. coli* may become essential in the different

genomic context of *Salmonella* serovars and vice versa, particularly in the case of *S. Typhi* where wide-spread pseudogene formation has eliminated potentially redundant pathways (Holt, Teo, et al., 2009; McClelland, Sanderson, Clifton, et al., 2004). Additionally, TraDIS will naturally over-predict essentiality in comparison to targeted knockouts, as our library creation protocol necessarily contains a short period of competitive growth between mutants during the recovery from electro-transformation and selection. As a consequence, genes which cause major growth defects, but not necessarily a complete lack of viability in clonal populations, may be reported as ‘required.’

2.3.3 Serovar-specific genes required for growth

Many of the required genes present in only one serovar encoded phage repressors, for instance the cI proteins of Fels-2/SopE and ST35 (see tables 2.2 and 2.3). Repressors maintain the lysogenic state of prophage, preventing transcription of early lytic genes (Echols, 1971). Transposon insertions into these genes will relieve this repression and trigger the lytic cycle, resulting in cell death, and consequently mutants are not represented in the sequenced library. This again broadens the definition of ‘required’ genes; such repressors may not be required for cellular viability in the traditional sense, but once present in these particular genomes, their maintenance is required for continued viability, as long as the rest of the phage remains intact.

S. Typhimurium and *S. Typhi* both contain 8 apparent large phage-derived genomic regions (N. Thomson et al., 2004; Kropinski et al., 2007). We were able to identify required repressors in all the intact lambdoid, P2-like, and P22-like prophage in both genomes, including Gifsy-1, Gifsy-2, and Fels-2/SopE (see tables 2.2 and 2.3). With the exception of the SLP203 P22-like prophage in *S. Typhimurium*, all of these repressors lack the peptidase domain of the classical λ repressor gene cI. This implies that the default anti-repression mechanism of *Salmonella* prophage may be more similar to a trans-acting mechanism recently discovered in Gifsy phage (Lemire, Figueroa-Bossi, and Bossi, 2011) than to the λ repressor’s RecA-induced self-cleavage mechanism. We are also able to confirm that most phage remnants and fusions contained no active repressors, with the exception of the SLP281 degenerate P2-like prophage in *S. Typhimurium*. This degenerate prophage contains both intact replication and integration genes, but appears to lack tail and head proteins, suggesting it may depend on another phage for

Table 2.2: Phage elements in *S. Typhimurium*. Genomic coordinates determined from annotations in the EMBL annotation for FQ312003 and manual inspection. Repressor domains and architecture were determined using the HMMER webserver (Finn, Clements, and Eddy, 2011) and Pfam (Punta et al., 2012). Phage types were determined using repressor sequence similarity searches and information from N. Thomson et al. (2004) and Kropinski et al. (2007).

Element name	Genomic coordinates	Repressor	Repressor domain(s)	Repressor domain architecture	Predicted active?	Phage type	Required cargo
Gifsy-2 SLP105	1054795 - 1100036	SL0950	HTH_3 (PF01381)		Yes	lambdoid	N/A
N/A	1913364 - 1925490	N/A	N/A	N/A	No	remnant	SL1799
SLP203	2039803 - 2079890	SL1967	HTH_19 (PF12844) and Pep-tidase_S24 (PF00717)		Yes	P22-like	N/A
Gifsy-1 SLP272	2726717 - 2777229	SL2593	HTH_3 (PF01381)		Yes	lambdoid	SL2549
SLP281	2815382 - 2825915	SL2633	2 Phage_CI_repr (PF07022)		Yes	degenerate P2-like	N/A
Fels-2 SLP285	2855616 - 2888522	SL2708	Phage_CI_repr (PF07022)		Yes	P2-like	SL2695
SLP289	2890073 - 2900377	IsrK RNA (RF01394)	N/A	N/A	No	P4-like	N/A
SLP443	4437731 - 4459844	N/A	N/A	N/A	No	remnant	SL4132

production of viral particles. Both genomes also encode P4-like satellite prophage, which rely on ‘helper’ phage for lytic functions and utilize a complex antisense-RNA based regulation mechanism for decision pathways regarding cell fate (Briani et al., 2001) using structural homologs of the IsrK (Padalon-Brauch et al., 2008) and C4 ncRNAs (Forti et al., 2002), known as seqA and CI RNA in the P4 literature, respectively. While the mechanism of P4 lysogenic maintenance is not known, the IsrK-like ncRNAs of two potentially active P4-like prophage in *S. Typhi* are required under TraDIS. This sequence element has previously been shown to be essential for the establishment of the P4 lysogenic state (Sabbattini et al., 1995), and we predict based on our observations that it may be necessary for lysogenic maintenance as well. The fact that some lambdoid prophage in *S. Typhimurium* encode non-coding genes structurally similar to the IsrK-C4 immunity system of P4 raises the possibility that these systems may be acting as a defense mechanism of sorts, protecting the prophage from predatory satellite phage capable of co-opting its lytic genes.

In addition to repressors, 4 prophage cargo genes in *S. Typhimurium* and one in *S. Typhi* are required (See tables 2.2, 2.3, 2.4, and 2.6). The *S. Typhimurium* prophage

Table 2.3: Phage elements in *S. Typhi*. Genomic coordinates determined from N. Thomson et al. (2004) and manual inspection. Repressor domains and architecture were determined using the HMMER webserver (Finn, Clements, and Eddy, 2011) and Pfam (Punta et al., 2012). Phage types were determined using repressor sequence similarity searches and information from N. Thomson et al. (2004) and Kropinski et al. (2007).

Element name	Genomic coordinates	Repressor	Repressor domain(s)	Repressor domain architecture	Predicted active?	Phage type	Required cargo
ST15	1408790 - 1441377	N/A	N/A	N/A	No	Mu/P2 fusion	N/A
Gifsy-2	1929572 - 1972330	t1920	HTH_3 (PF01381)		Yes	lambdaoid	N/A
ST2-27	2735054 - 2745321	IsrK RNA (RF01394)	N/A	N/A	Yes	P4-like	N/A
ST27	2745477 - 2768221	N/A	N/A	N/A	No	P2/iroA fusion	N/A
ST35	3500854 - 3536047	t3402	Phage_CI_repr (PF07022)		Yes	P2-like	t3415
SopE	4457346 - 4491316	t4337	Phage_CI_repr (PF07022)		Yes	P2-like	N/A
N/A	4519423 - 4519501	IsrK RNA (RF01394)	N/A	N/A	No	remnant	N/A
ST46	4666579 - 4677433	IsrK RNA (RF01394)	N/A	N/A	Yes	P4-like	N/A

cargo genes encode a PhoPQ regulated protein, a protein predicted to be involved in natural transformation, an endodeoxyribonuclease, and a hypothetical protein. The *S. Typhi* prophage cargo gene encodes a protein containing the DNA-binding HIRAN domain (Iyer, Babu, and Aravind, 2006), believed to be involved in the repair of damaged DNA. These warrant further investigation, as they are genes that have been recently acquired and become necessary for survival in rich media.

To compare differences between requirements for orthologous genes in both serovars, we calculated log-fold read ratios to eliminate genes which were classified differently in *S. Typhi* and *S. Typhimurium* but did not have significantly different read densities (see Methods.) Even after this correction, 36 *S. Typhimurium* genes had a significantly lower frequency of transposon insertion compared to the equivalent genes in *S. Typhi* ($P < 0.05$), including four encoding hypothetical proteins (table 2.4). This indicates that these gene products play a vital role in *S. Typhimurium* but not in *S. Typhi* when grown under laboratory conditions.

Table 2.4: Genes uniquely required in *S. Typhimurium*. Genes determined to be uniquely required in *S. Typhimurium*. SL, *S. Typhimurium*; Ty, *S. Typhi*; inserts refer to the number of unique insertion sites within a gene; reads refer to the number of sequence reads over all insertions sites within a gene. †, P-value (associated with log2 read ratio) < 0.05. ‡, *sseJ* is a pseudogene in *S. Typhi*. Shaded rows indicate genes shown to be H-NS repressed in Navarre et al. (2006)

Ty inserts	Ty reads	SL inserts	SL reads	SL ID	SL gene length	Ty ID	Ty gene length	Name	Function
No orthologs in <i>S. Typhi</i>									
-	-	18	123	SL0742	1269	-	-	-	putative cation transporter
-	-	9	80	SL0830	516	-	-	-	conserved hypothetical protein
-	-	4	21	SL0831	855	-	-	-	putative electron transfer flavoprotein (beta subunit)
-	-	0	0	SL0950	323	-	-	-	predicted bacteriophage protein, potential phage repressor Gifsy-2
-	-	11	75	SL1179	789	-	-	-	envF
-	-	3	18	SL1480	249	-	-	-	lipoprotein
-	-	4	32	SL1527	264	-	-	-	antitoxin Phd.YefM, type II toxin-antitoxin system
-	-	1	3	SL1560	717	-	-	-	putative inner membrane protein
-	-	7	50	SL1601	859	-	-	-	putative membrane protein
-	-	4	36	SL1799	201	-	-	-	putative transcriptional regulator (pseudogene)
-	-	5	22	SL1830A	434	-	-	-	bacteriophage encoded pagK (phoPQ-activated protein)
-	-	3	27	SL1967	677	-	-	-	conserved hypothetical protein (pseudo-gene)
-	-	1	15	SL2045A	63	-	-	-	predicted bacteriophage protein, potential phage repressor SLP203
-	-	17	107	SL2066	900	-	-	-	short ORF
-	-	3	34	SL2549	209	-	-	-	CDP-abequose synthase
-	-	4	149	SL2593	449	-	-	-	endodeoxyribonuclease
-	-	3	7	SL2633	846	-	-	-	putative DNA-binding protein, potential phage repressor Gifsy-1 SLP72
-	-	2	21	SL2695	978	-	-	-	putative repressor protein, SLP281
-	-	5	39	SL1132	291	-	-	-	putative competence protein
-	-	5	45	SL4354A	303	-	-	-	hypothetical protein
Present in <i>S. Typhi</i> but required only in <i>S. Typhimurium</i>									
36	474	5	26	SL0032	441	t0033	306	-	putative transcriptional regulator
71	349	11	48	SL0623	642	t2232	576	lipB	lipoteichoic acid ligase B
151	3546	10	64	SL0702	897	t2156	894	-	putative glycosyl transferase
194	3007	9	61	SL0703	1134	t2155	1134	-	galactosyltransferase
231	3499	15	67	SL0706	1779	t2152	1780	-	putative glycosyltransferase, cell wall biogenesis
84	1041	2	4	SL0707	834	t2151	834	-	putative glycosyltransferase, cell wall biogenesis
49	367	14	70	SL0722	1569	t2136	1569	cycD	cytchrome d ubiquinol oxidase subunit I
74	1613	5	22	SL1069	693	t1789	693	-	putative secreted protein
20	199	1	1	SL11203	150	t1146	156	-	hypothetical protein

*Chapter 2. A comparison of dense transposon insertion libraries in the *Salmonella* serovars *Typhi* and *Typhimurium**

20	290	1	5	SL1264	315	t1209	315	-
84	384	6	26	SL1327	402	t1261	384	spiC
66	769	5	35	SL1331	270	t1265	327	sseA
36	307	2	5	SL1341	228	t1275	228	ssaH
47	407	1	3	SL1342	249	t1276	249	ssaJ
144	3197	5	14	SL1343	750	t1277	750	putative pathogenicity island protein
63	847	5	26	SL1354	267	t1288	267	putative pathogenicity island protein
73	762	4	44	SL1355	780	t1289	780	putative type III secretion protein
30	226	12	48	SL1386	693	t1322	693	putative type III secretion protein
265	3337	29	165	SL1473	1557	t1463	1557	Electron transport complex protein rnfE
85	765	6	35	SL1532	951	t1511	951	PhoPQ-activated protein
22	156	16	174	SL1561	1227	t1534 [‡]	141	putative virulence effector protein
119	1639	10	44	SL1563	762	t1536	762	Salmonella translocated effector protein (SseJ)
107	2440	5	44	SL1564	648	t1537	648	putative periplasmic amino acid-binding protein
183	1646	20	118	SL1628	1355	t1612	1364	putative ABC amino acid transporter permease
23	177	1	5	SL1659	183	t1640	183	hypothetical protein
78	617	16	104	SL1684	1014	t1664	1014	conserved hypothetical protein
37	277	4	25	SL1785	396	t1022	396	putative regulatory protein
166	2823	9	27	SL1793	915	t1016	915	conserved hypothetical protein
28	311	3	22	SL1794	159	t1015	159	inner membrane protein
23	155	1	4	SL1823	972	t0988	972	putative acyltransferase
60	402	11	58	SL2064	1002	t0786	1002	putative glycosyl transferase
87	524	7	59	SL2065	1293	t0785	1290	putative O-antigen transporter
66	559	13	74	SL2069	774	t0780	774	glucose-1-phosphate cytidylyltransferase
41	204	5	14	SL3828	1830	t3658	1830	glucosamine-fructose-6-phosphate aminotransferase
27	288	5	23	SL4250	288	t4220	288	putative GerE family regulatory protein
148	2633	16	89	SL4251	876	t4221	876	araC family regulatory protein

Present in *S. Typhi* but required
only in *S. Typhimurium*

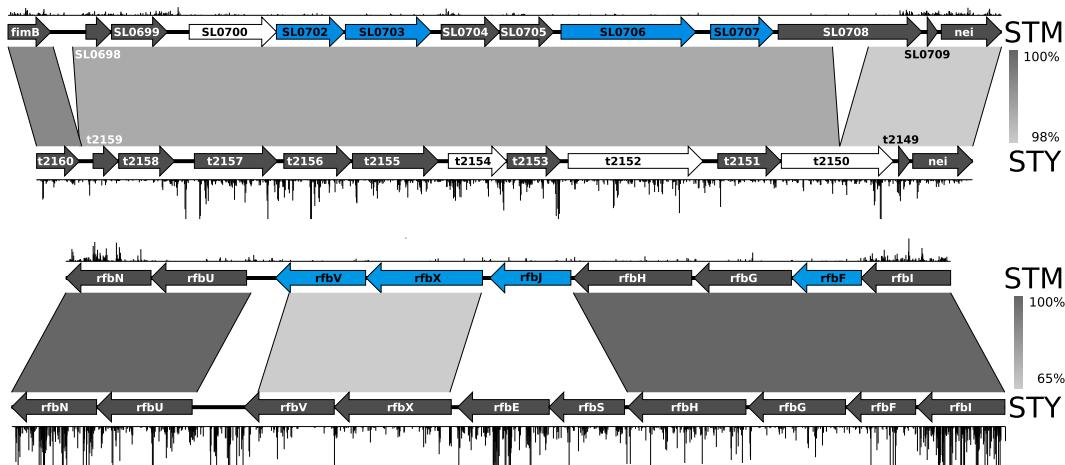


Figure 2.5: Comparison of cell surface operon structure and requirements. Diagram illustrating cell surface operons with different requirement patterns in *S. Typhimurium* and *S. Typhi*. The top figure is of an uncharacterized operon putatively involved in cell wall biogenesis, while the bottom figure shows a portion of the *rfb* operon involved in O-antigen biosynthesis. Plots along the top and bottom of each figure show insertions in *S. Typhimurium* and *S. Typhi*, respectively, with read depth on the y-axis with a maximum cut-off of 100 reads. Genes in blue are required in *S. Typhimurium*, genes in white are pseudogenes; others are in grey. Grey rectangles represent BLAST hits between orthologous genes, with percent nucleotide identity colored on the scale to the right of each figure.

A major difference between the two serovars is in the requirement for genes involved in cell wall biosynthesis (see figure 2.5). A set of four genes (SL0702, SL0703, SL0706, and SL0707) in an operonic structure putatively involved in cell wall biogenesis is required in *S. Typhimurium* but not in *S. Typhi*. The protein encoded by SL0706 is a pseudogene in *S. Typhi* (Ty2 unique ID: t2152) due to a 1bp deletion at codon 62 that causes a frameshift (Figure 4a). This operon contains an additional two pseudogenes in *S. Typhi* (t2154 and t2150), as well as a single different pseudogene (SL0700) in *S. Typhimurium*, indicating that this difference in gene requirements reflects the evolutionary adaptation of these serovars to their respective niches. Similarly, four genes (*rfbV*, *rfbX*, *rfbJ* and *rfbF*) within an O-antigen biosynthetic operon are required by *S. Typhimurium* but not *S. Typhi*. There appears to have been a shuffling of O-antigen biosynthetic genes since the divergence between the two serovars, and *rfbJ*, encoding a CDP-abequose synthase, has been lost from *S. Typhi* altogether. These broader requirements for cell wall-associated biosynthetic and transporter genes suggest that surface structure biogenesis is of greater

importance in *S. Typhimurium*.

We also identified seven genes from the shared pathogenicity island SPI-2 that appear to contain few or no transposon insertions only in *S. Typhimurium* under laboratory conditions. These genes (*spiC*, *sseA*, and *ssaHIJT*) are thought to encode components of the SPI-2 type III secretion system apparatus (T3SS) (Kuhle and Hensel, 2004). In addition, the effector genes *sseJ* and *sifB*, whose products are secreted through the SPI-2-encoded T3SS (Miao and S. I. Miller, 2000; Freeman, Ohl, and S. I. Miller, 2003), also fell into the ‘required’ category in *S. Typhimurium* alone. All of these genes display high A+T nucleotide sequence and have been previously shown (in *S. Typhimurium*) to be strongly bound by the nucleoid associated protein H-NS, encoded by *hns* (Lucchini et al., 2006; Navarre et al., 2006). Therefore, rather than being ‘required’, it is instead possible that access for the transposon was sufficiently restricted that very few insertions occurred at these sites. In further support of this hypothesis, a comparison of the binding pattern of H-NS detected in studies using *S. Typhimurium* LT2 with the TraDIS results from the SPI-2 locus indicated that high regions of H-NS enrichment correlated well with both the *ssa* genes described here and with *sseJ* (see figure 2.6). An earlier study also suggests that high-density DNA binding proteins can block Mu, Tn5, and Tn10 insertion (Manna et al., 2007); however, a genome-wide study of the effects of H-NS binding on transposition would be necessary to confirm this effect.

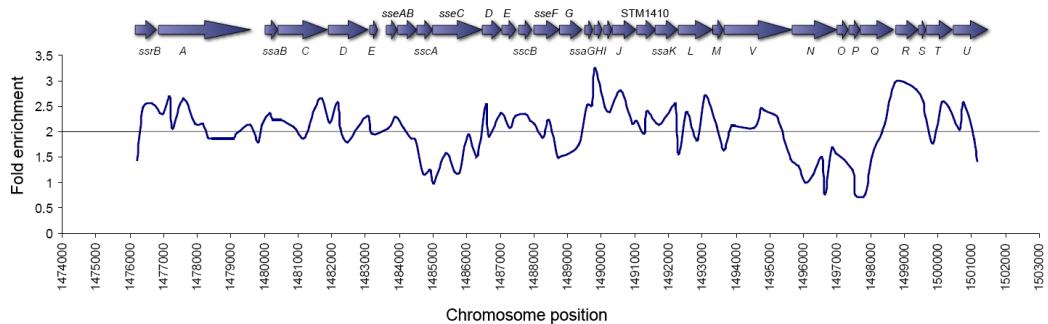


Figure 2.6: H-NS enrichment across the SPI-2 locus. Based on data from Lucchini et al. (2006) where a 2 fold enrichment of H-NS-bound DNA over a total genomic DNA control in a ChIP-on-chip experiment was taken to indicate regions of H-NS binding in *S. Typhimurium* strain LT2. Assuming these binding patterns are similar in the *S. Typhimurium* strain tested in this study, H-NS binding may have affected transposon access to genes in the SPI-2 locus.

Table 2.5: Candidate required genes affected by H-NS binding in *S. Typhimurium*. Genes identified by comparison with data from Navarre et al. (2006). Fold change values report the results of a ChIP-on-chip experiment, and indicate genes strongly bound by H-NS.

Gene	SL ID	STM ID	Fold change	Function
-	SL0830	STM0854	-16.2	conserved hypothetical protein
-	SL0831	STM0855	-33.8	putative putative electron transfer flavoprotein (beta subunit)
-	SL1069	STM1131	-13.5	putative putative secreted protein
spiC	SL1327	STM1393	-19.1	putative pathogenicity island 2 secreted effector protein
sseA	SL1331	STM1397	-46	Type three secretion system chaperone
ssaH	SL1341	STM1407	-8.8	Type three secretion system apparatus
ssaI	SL1342	STM1408	-32.4	putative putative pathogenicity island protein
ssaJ	SL1343	STM1409	-53.7	putative putative pathogenicity island lipoprotein
ssaS	SL1354	STM1420	-15.5	putative putative type III secretion protein
ssaT	SL1355	STM1421	-33.9	putative putative type III secretion protein
pqaA	SL1473	STM1544	-5.5	PhoPQ-activated protein
sifB	SL1532	STM1602	-66.8	putative putative virulence effector protein
-	SL1560	STM1630	-9.8	putative putative membrane protein
sseJ	SL1561	STM1631	-48.6	salmonella translocated effector protein (SseJ)
-	SL1563	STM1633	-91.9	putative putative periplasmic amino acid-binding protein
-	SL1564	STM1634	-22.5	putative putative ABC amino acid transporter permease
-	SL1628	STM1698	-101.4	hypothetical protein
-	SL1659	STM1728	-17.3	cytochrome b561 (cytochrome b-561)
-	SL1785	STM1856	-12.1	conserved hypothetical protein
pagO	SL1793	STM1862	-11.9	inner membrane protein (PagO)
-	SL1794	STM1864	-22.9	putative inner membrane protein

Indeed, the generation of null *S. Typhimurium* mutants in *sseJ* and *sifB*, as well as many others generated at the SPI-2 locus suggest that these genes are not truly a requirement for growth in this serovar (Freeman, Ohl, and S. I. Miller, 2003; Hensel, Shea, Baumler, et al., 1997; Hensel, Shea, Waterman, et al., 1998; Ohlson et al., 2005). While this is a reminder that the interpretation of gene requirement needs to be made with care, the effect of H-NS upon transposon insertion is not genome-wide. If this were the case, there would be an under-representation of transposon mutants in high A+T regions (known for H-NS binding), which is not what we observed. In total, only 21 required genes fall into the ‘*hns*-repressed’ category described in Navarre et al. (2006)(see table 2.5); the remainder (almost 400) contained sufficient transposon insertions to conclude they were non-required. In addition, we noted that all SPI-1 genes that encode another T3SS and are of high A+T content were also found to be non-required. This phenomenon was not observed in *S. Typhi*, possibly because the strain used harbors the pHCM1 plasmid, which encodes the H-NS-like protein *sfh* and has been shown to affect H-NS binding (Doyle et al., 2007; Dillon et al., 2010).

Twenty-two *S. Typhi* genes had a significantly lower frequency of transposon insertion compared to orthologs in *S. Typhimurium* ($P < 0.05$), indicating that they are required only in *S. Typhi* for growth under laboratory conditions (table 2.6), including the *fepBDGC* operon. This indicates a requirement for ferric (Fe(III)) rather than ferrous

(Fe(II)) iron. This can be explained by the presence of Fe(III) in the bloodstream, where *S. Typhi* can be found during typhoid fever (Wain et al., 1998). These genes function to recover the ferric chelator enterobactin from the periplasm, acting with a number of proteins known to aid the passage of this siderophore through the outer membrane (Rabsch et al., 1999). It has long been noted that *aroA* mutants of *S. Typhi*, deficient in their ability to synthesize enterobactin, exhibit severe growth defects on complex media, while similar mutants of *S. Typhimurium* grow normally under the same conditions (Edwards and Stocker, 1988), though the mechanism has not been clear. Our results suggest that this difference in growth of *aroA* mutants is caused by a requirement for iron uptake through the *fep* system in *S. Typhi*. During host adaptation, *S. Typhi* has accumulated pseudogenes in many iron transport and response systems (McClelland, Sanderson, Clifton, et al., 2004), presumably because they are not necessary for survival in the niche *S. Typhi* occupies in the human host, which may have led to this dependence on *fep* genes. In contrast, *S. Typhimurium* generally causes intestinal rather than systemic infection and is able to utilize a wider range of iron sources, including Fe(II), a soluble form of iron present under anaerobic conditions such as those found in the intestine (Tsolis et al., 1996).

Table 2.6: Genes uniquely required in *S. Typhi*. Genes determined to be uniquely required in *S. Typhi*. SL, *S. Typhimurium*; Ty, *S. Typhi*; inserts refer to the number of unique insertion sites within a gene; reads refer to the number of sequence reads over all insertions sites within a gene. †, P-value (associated with log² read ratio) < 0.05. *, the assignment of recA as a required gene has been described previously (Langridge et al., 2009), but briefly is believed to be due to the presence of the priC pseudogene in *Typhi*.

SL inserts		SL reads	SL inserts	Ty reads	Ty ID	Ty gene length	SL ID	SL gene length	Name	Function
<i>S. Typhimurium</i> in <i>S. Typhi</i>										
-	-	-	1	5	t1332	132	-	-	maY	pseudogene
-	-	-	2	32	t1920	405	-	-	-	possible repressor
-	-	-	2	12	t3157	165	-	-	-	10/Gifsy-2
-	-	-	2	12	t3166	228	-	-	-	conserved hypothetical protein
-	-	-	6	196	t3402	570	-	-	-	spurious ORF annotation overlapping the RnasP/M1 RNA
-	-	-	4	58	t3415	741	-	-	cI	repressor/protein, cs 73 prophage
-	-	-	1	6	t4531	150	-	-	-	HIRAN-domain family gene, potential DNA repair
-	-	-	-	-	-	-	-	-	-	hypothetical secreted protein
No orthologs in <i>S. Typhi</i>										
199	1792	18	59	t095	1287	SL0093	1287	surA	survival protein SurA precursor	
45	498	3	22	t0123	459	SL0119	459	yabB/mraZ	conserved hypothetical protein	
120	589	11	32	t0203	1281	SL0203	1281	hemL	glutamate-1-semialdehyde 2,1-	
123	982	2	25	t0224	1353	SL0224	1353	yaeL/rseP	aminotransferase	
67	452	1	14	t0270	576	SL2604	576	rpoE	Zinc metallopeptidase	
140	760	0	0	t0587	2286	SL2246	2286	nrdA	RNA polymerase sigma-E factor	
113	641	15	42	t2140	2802	SL0718	2802	sucA	ribonucleoside-diphosphate reductase 1 alpha chain	
116	753	13	36	t2177	1641	SL0680	1641	pgm	2-oxoglutarate dehydrogenase E1 component	
80	542	9	15	t2276	1008	SL0580	1008	fepD	phosphoglucomutase	
93	591	2	2	t2277	990	SL0579	990	fepG	ferri enterobactin transport protein	
64	508	5	6	t2278	795	SL0578	795	fepC	FepG ferric enterobactin transport ATP-	
201	1129	12	116	t2410	2355	SL0444	2355	lon	binding protein FepC	
95	518	8	20	t2730	1062	SL2809	1062	recA*	Lon protease	
135	719	16	39	t2996	1992	SL3052	1947	tktA	recA protein	
76	358	3	9	t3120	1434	SL3173	1434	rfaE	transketolase	
213	1976	6	50	t3265	1071	SL3321	1071	degS	ADP-heptose synthase	
43	448	3	10	t3326	606	SL3925	606	yigP	serine protease	
124	571	17	36	t3384	2025	SL3872	2025	rep	conserved hypothetical protein	
175	1208	6	21	t3621	2787	SL3947	2787	polA	ATP-dependent DNA helicase	
117	797	9	13	t3808	1047	SL3677	1047	waaf	DNA polymerase I	
176	1628	14	59	t4153	1080	SL4183	1080	alr	ADP-heptose-LPS heptosyltransferase II	
140	1127	10	38	t4411	951	SL4294	951	miaA	alanine racemase	
Present in <i>S. Typhimurium</i> but required only in <i>S. Typhi</i>										

2.3.4 TraDIS provides resolution sufficient to evaluate ncRNA contributions to fitness

Under a Poisson approximation to the transposon insertion process, a region of 41 (in *S. Typhimurium*) or 60 bases (in *S. Typhi*) has only a 1% probability of not containing an insertion. NcRNAs tend to be considerably shorter than their protein-coding counterparts, but this gives us sufficient resolution to assay most of the non-coding complement of the *Salmonella* genome. As a proof of principle, we performed an analysis of the best-understood class of small ncRNAs, the tRNAs. Francis Crick hypothesized that a single tRNA could recognize more than one codon through wobble recognition (Crick, 1966), where a non-canonical G-U base pair is formed between the first (wobble) position of the anticodon and the third nucleotide in the codon. As a result, some codons are covered by multiple tRNAs, while others are covered non-redundantly by a single tRNA. We expect that singleton wobble-capable tRNAs, that is wobble tRNAs which recognize a codon uniquely, will be required. In addition, we inferred the requirement for other tRNAs through the non-redundant coverage of their codons and used this to benchmark our ability to use TraDIS to reliably interrogate short genomic intervals.

The *S. Typhi* and *S. Typhimurium* genomes encode 78 and 85 (plus one pseudogene) tRNAs respectively with 40 anticodons, as identified by tRNAscan-SE (Lowe and Eddy, 1997). In *S. Typhi*, 10 out of 11 singleton wobble tRNAs are predicted to be required or ambiguous, compared to 16 tRNAs below the ambiguous LLR cut-off overall (significant enrichment at the 0.05 level, two-tailed Fishers exact test p-value: 6.4e-08.) Similarly in *S. Typhimurium*, 9 of 11 singleton wobble tRNAs are required or ambiguous compared to 15 required or ambiguous tRNAs overall, again showing a significant enrichment of required tRNAs in this subset (Fishers exact test p-value: 5.2e-07.) The one singleton wobble tRNA which is consistently not required in both serovars is the tRNA-Pro(GGG), which occurs within a 4-member codon family. It has previously been shown in *S. Typhimurium* that tRNA-Pro(UGG) can read all four proline codons in vivo due to a cmo5U34 modification to the anticodon, obviating the need for a functional tRNA-Pro(GGG) (Näsvall, P. Chen, and Björk, 2004) and making this tRNA non-required. The other non-required singleton wobble tRNA in *S. Typhimurium*, tRNA-Leu(GAG), is similarly a member of a 4-member codon family. We predict tRNA-Leu(TAG) may also be capable of recognizing all 4 leucine codons in this serovar; such a leucine “four-way

wobble” has been previously inferred in at least one other bacterial species (Osawa et al., 1992; Marck and Grosjean, 2002).

Of the 6 required non-wobble tRNAs in each serovar, four are shared. These include two non-wobble singleton tRNAs covering codons uniquely, as well as a tRNA with the ATG anticodon which is post-transcriptionally modified by the required protein MesJ/TilS to recognize the isoleucine codon ATA (Marck and Grosjean, 2002). An additional two required tRNAs in both serovars, one shared and one with a differing anticodon, contain Gln anticodons and are part of a polycistronic tRNA operon containing other required tRNAs. This operon is conserved in *E. coli* with the exception of an additional tRNA-Gln at the 3’ end that has been lost in the *Salmonella* lineage. It is possible that transposon insertions early in the operon may interfere with processing of the polycistronic transcript into mature tRNAs. Finally, we do not observe insertions in a tRNA-Met and a tRNA-Val in *S. Typhi* and *S. Typhimurium*, respectively.

Using this analysis of the tRNAs we estimate a worst-case PPV for these short molecules (~76 bases) at 81%, in line with our previous estimates for conserved protein-coding genes, and a FPR of <4%, higher than for protein-coding genes but still well within the typical tolerance of high-throughput experiments. This assumes that the “required” operonic tRNA-Glns and the serovar-specific tRNA-Met and tRNA-Val are all false positives; it is not clear that this is in fact the case.

Surveying the shared required ncRNA content of both serovars (see table 2.7), we find that the RNA components of the signal recognition particle (SRP) and RNase P, two universally conserved ncRNAs, are required as expected. The SRP is an essential component of the cellular secretion machinery, while RNase P is necessary for the maturation of tRNAs. We also find a number of required known and potential cis-regulatory molecules associated with genes required for growth under laboratory conditions in both serovars. The FMN riboswitch controls *ribB*, a 3,4-dihydroxy-2-butanone 4-phosphate synthase involved in riboflavin biosynthesis, in response to flavin mononucleotide concentrations (Winkler, Nahvi, and Breaker, 2002). Additionally, we are able to assign putative functions to a number of previously uncharacterized required non-coding transcripts through their 5’ association with required genes. SroE, a 90 nucleotide molecule discovered in an early sRNA screen (Vogel et al., 2003), is consistently located at the 5’ end of the required *hisS* gene across its phylogenetic distribution in the Enterobacteriaceae. Given this consistent association and the function of HisS as a histidyl-tRNA synthetase, we

hypothesize that this region may act in a manner similar to a T-box leader, inducing or repressing expression in response to tRNA-His levels. The *thrU* leader sequence, recently discovered in a deep-sequencing screen of *E. coli* (Raghavan, Groisman, and Ochman, 2011), appears to regulate a polycistronic operon of required singleton wobble tRNAs. Three additional required cis-regulatory elements, t44, S15, and StyR-8, are associated with required ribosomal proteins, highlighting the central role ncRNA elements play in regulating fundamental cellular processes.

Table 2.7: Candidate required ncRNAs greater than 60 nucleotides in length, excluding rRNA and tRNA. Known and putative non-coding elements classified as required or ambiguous in this screen. Required ncRNAs have a log-likelihood ratio (LLR) between required and non-required models of < -2; see Methods. * †, ncRNAs which are ambiguous (LLR between -2 and 2) in *S. Typhi*(*) or in *S. Typhimurium*(†). Hfq-binding annotations are taken from Chao et al. (2012). The downstream protein-coding genes columns report annotated CDS or ribosomal RNA start sites within 100 bases of each candidate required non-coding element on either strand, and whether these downstream sequences are also classified as required.

Name	Rfam accession	Function	Hfq-binding	Downstream protein-coding gene(s)	Downstream gene required	References
Required or ambiguous in both <i>S. Typhi</i> and <i>S. Typhimurium</i>						
SRP	RF00169	RNA component of the signal recognition particle				Rosenblad et al. (2009)
RNase P	RF00010	RNA component of RNase P	<i>ybaZ</i>	N		Frank and Pace (1998)
RFN	RF00050	FMN-sensing riboswitch controlling the <i>ribB</i> gene	<i>ribB</i>	Y		Winkler, Nahvi, and Breaker (2002)
SroE	RF00371	Putative cis-regulatory element controlling the <i>hisS</i> gene	<i>hisS</i>	Y		Vogel et al. (2003)
ThrU Leader	NA	Putative cis-regulatory element controlling the ThrU tRNA operon				Raghavan, Groisman, and Ochman (2011)
t44	RF00127	Cis-regulatory element controlling the ribosomal <i>rpsB</i> gene	<i>rpsB</i>	Y		Tjaden et al. (2002); Asseev et al. (2008); Meyer et al. (2009)
S15†	RF00114	Translational regulator of the ribosomal S15 protein	<i>rpsO</i>	Y		Benard et al. (1996)
StyR-8	NA	Putative cis-regulatory element controlling the ribosomal <i>rpmB</i> gene	<i>rpmB</i>	Y		Chinni et al. (2010)
MicA	RF00078	sRNA involved in cellular response to extracytoplasmic stress	Y	<i>luxS</i>	N	Vogel (2009b)
DsrA†	RF00014	sRNA regulator of H-NS	Y	<i>mngB</i>	N	Lease, Cusick, and Belfort (1998)
STnc10	NA	Putative sRNA		<i>nhaA</i>	N	Sittka et al. (2008)
STnc60†	NA	Putative sRNA				Sittka et al. (2008)
STnc840	NA	Verified sRNA derived from 3' UTR of the <i>fglL</i> gene	Y			Chao et al. (2012)
IS0420*†	NA	Putative ncRNA		<i>rmf</i>	N	Raghavan, Groisman, and Ochman (2011); S. Chen et al. (2002)
RGO0†	NA	Putative sRNA identified in <i>E. coli</i>				Raghavan, Groisman, and Ochman (2011)
Required or ambiguous in <i>S. Typhimurium</i> only						
rne5	RF00040	RNase E autoregulatory 5' element	<i>rne</i>		Y	Diwa et al. (2000)
RydC	RF00505	sRNA regulator of the <i>yejABEF</i> ABC transporter	Y			Antal et al. (2005)
RydB	RF00118	Putative ncRNA				Wassarman et al. (2001)
STnc510	NA	Putative sRNA		<i>pagD/pagC</i>	Y/N	Sittka et al. (2008)
STnc460†	NA	Putative sRNA				Sittka et al. (2008)
STnc170	NA	Putative sRNA		<i>SL1458</i>	N	Sittka et al. (2008)
STnc130	NA	Putative sRNA		<i>dmsA</i>	N	Sittka et al. (2008)
RseX	RF01401	sRNA regulator of OmpA and OmpC	Y			Douchin, Bohn, and Bouloc (2006)
IsrJ	RF01393	sRNA regulator of SPI-1 effector protein secretion				Sittka et al. (2008); Padalon-Brauch et al. (2008)

IsrI	RF01392	Island-encoded sRNA	Hfq-binding	Y	SL1028	Y	Sittka et al. (2008); Padalon-Brauch et al. (2008); Chao et al. (2012)
Required or ambiguous in <i>S. Typhi</i> only							
RybB	RF00110	sRNA involved in cellular response to extracytoplasmic stress		Y			Vogel (2009b)
tk5*	NA	Putative ncRNA					Raghavan, Groisman, and Ochman (2011); Rivas and Eddy (2001)
STnc750	NA	Verified sRNA		Y	<i>speB</i>	N	Kröger et al. (2012); Chao et al. (2012)
StyR-44*	RF01830	Putative multicopy (2/6 copies required in <i>S. Typhi</i>) ncRNA associated with ribosomal RNA operon			23S rRNA	N	Chinni et al. (2010)
tp2	NA	Putative ncRNA			<i>aceE</i>	N	Raghavan, Groisman, and Ochman (2011); Rivas and Eddy (2001)
RdID	RF01813	RdID RNA anti-toxin, 1/2 copies required in <i>S. Typhi</i>					Kawano et al. (2002)
STnc120*	NA	Putative sRNA					Sittka et al. (2008)
tp28*	NA	Putative ncRNA		<i>fur</i>		N	Raghavan, Groisman, and Ochman (2011); Rivas and Eddy (2001)
Phe Leader*	RF01859	Phenylalanine peptide leader sequence associated with the required <i>pheST</i> operon			<i>pheS</i>	Y	Zurawski et al. (1978)
RimP Leader	RF01770	Putative cis-regulator of the <i>rimP-nusA-infB</i> operon			<i>rimP</i>	Y	Naville and Gautheret (2010)
GlmY	RF00128	Trans-acting regulator of the <i>glmS</i> gene					Urban and Vogel (2008)

2.3.5 sRNAs required for competitive growth

Inferring functions for potential trans-acting ncRNA molecules, such as anti-sense binding small RNAs (sRNAs), from requirement patterns alone is more difficult than for cis-acting elements, as we cannot rely on adjacent genes to provide any information. It is also important to keep in mind that TraDIS assays requirements after a brief competition within a large library of mutants on permissive media. This may be particularly important when surveying the bacterial sRNAs, which are known to participate in responses to stress (Vogel, 2009a).

This is demonstrated by two sRNAs involved in the σ^E -mediated extracytoplasmic stress response, RybB and RseX, both of which can be successfully knocked out in *S. Typhimurium* (83). In *S. Typhi*, *rpoE* is required, as it also is in *E. coli* (Baba et al., 2006; De Las Penas, Connolly, and Gross, 1997). However, in *S. Typhimurium*, *rpoE* tolerates a heavy insertion load, implying that σ^E mutants are not disadvantaged in competitive growth. In *S. Typhimurium*, the sRNA RseX is required. Overexpression of RseX has previously been shown to compensate for σ^E essentiality in *E. coli* by leading

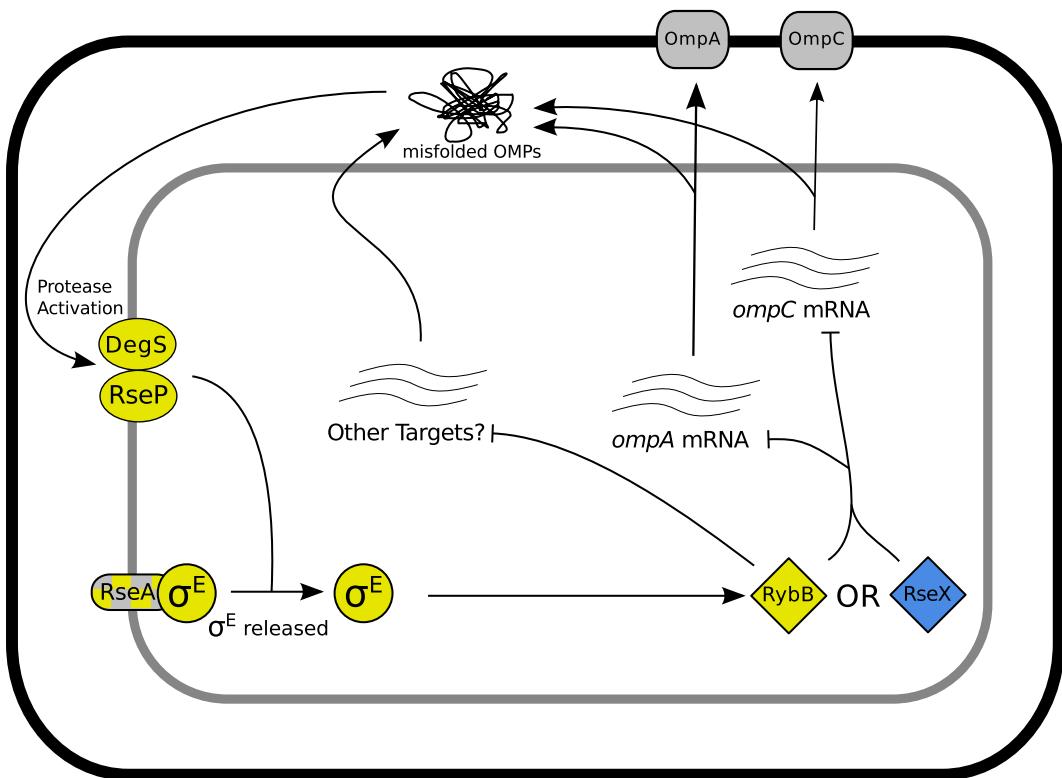


Figure 2.7: Proposed differences in sRNA utilization. Diagram illustrating inferred required sRNA regulatory networks under TraDIS. Molecules required in *S. Typhi* are highlighted in yellow and in *S. Typhimurium* are highlighted in blue. RseA, in yellow/grey check, is ambiguous in *S. Typhi*. Non-required molecules are in grey. Diamonds indicate sRNAs, circles regulatory proteins, ovals proteases, oblong shapes are membrane-anchored proteins, and rounded squares are outer membrane porins.

to the degradation of *ompA* and *ompC* transcripts (85). This suggests that RseX may also be short-circuiting the σ^E stress response network in *S. Typhimurium* (figure 2.7). To our knowledge, this is the first evidence of a native (i.e. not experimentally induced) activity of RseX.

S. Typhi on the other hand requires σ^E along with its activating proteases RseP and DegS and anchoring protein RseA, as well as the σ^E -dependent sRNA RybB, which also regulates OmpA and OmpC in *S. Typhimurium*, along with a host of other OMPs (Papenfort et al., 2006). It is unclear why the σ^E response is required in *S. Typhi* but not *S. Typhimurium*, though it may partially be due to the major differences in the cell

wall and outer membrane between the two serovars. In addition, there are significant differences in the OMP content of the *S. Typhi* and *S. Typhimurium* membranes that may be driving alternative mechanisms for coping with membrane stress. For instance, *S. Typhi* completely lacks OmpD, a major component of the *S. Typhimurium* outer membrane (Santiviago, Toro, et al., 2003) and a known target of RybB (Vogel, 2009a).

Two additional sRNAs involved in stress response are also required by both *S. Typhi* and *S. Typhimurium*. The first, MicA, is known to regulate *ompA* and the *lamB* porin-coding gene in *S. Typhimurium* (Bossi and Figueroa-Bossi, 2007), contributing to the extracytoplasmic stress response. The second, DsrA, has been shown to negatively regulate the nucleoid-forming protein H-NS and enhance translation of the stationary-phase alternative sigma factor σ^S in *E. coli* (Lease, Cusick, and Belfort, 1998), though its regulation of σ^S does not appear to be conserved in *S. Typhimurium* (Jones, Goodwill, and Elliott, 2006). Both have been previously deleted in *S. Typhimurium*, and so are not essential. H-NS knockouts have previously been shown to have severe growth defects in *S. Typhimurium* that can be rescued by compensatory mutations in either the *phoPQ* two-component system or *rpoS*, implying that the lack of H-NS is allowing normally silenced detrimental regions to be transcribed (Navarre et al., 2006). As MicA has recently been shown to negatively regulate PhoPQ expression in *E. coli* (Coornaert et al., 2010), it is tempting to speculate that MicA may be moderating the effects of DsrA-induced H-NS repression; however, it is currently unclear whether sRNA regulons are sufficiently conserved between *E. coli* and *S. enterica* to justify this hypothesis.

2.4 Conclusions

The extremely high resolution of TraDIS has allowed us to assay gene requirements in two very closely related *Salmonellae* with different host ranges. We found, under laboratory conditions, that 58 genes present in both serovars were required in only one, suggesting that identical gene products do not necessarily have the same phenotypic effects in the two different serovar backgrounds. Many of these genes occur in genomic regions or metabolic systems which contain pseudogenes and/or have undergone reorganization since the divergence of *S. Typhi* and *S. Typhimurium*, demonstrating the complementarity of TraDIS and phylogenetic analysis. These changes may in part explain differences observed in the pathogenicity and host specificity of these two serovars. In particular, *S.*

Typhimurium showed a requirement for cell surface structure biosynthesis genes; this may be partially explained by the fact that *S. Typhi* expresses the Vi-antigen which masks the cell surface, though these genes are not required for survival in our assay. *S. Typhi* on the other hand has a requirement for iron uptake through the *fep* system, which enables ferric enterobactin transport. This dependence on enterobactin suggests that *S. Typhi* is highly adapted to the iron-scarce environments it encounters during systemic infections. Furthermore, this appears to represent a single point of failure in the *S. Typhi* iron utilization pathways, and may present an attractive target for narrow-spectrum antibiotics.

Of the approximately 4500 protein coding genes present in each serovar, only about 350 were sufficiently depleted in transposon insertions to be classified as required for growth in rich media. This means that over 92% of the coding genome has sufficient insertion density to be queried in future assays. Dense transposon mutagenesis libraries have been used to assay gene requirements under conditions relevant for infection, including *S. Typhi* survival in bile (Langridge et al., 2009), *Mycobacterium tuberculosis* catabolism of cholesterol (Griffin et al., 2011), drug resistance in *Pseudomonas aeruginosa* (Gallagher, Shendure, and Manoil, 2011), and *Haemophilus influenzae* survival in the lung (Gawronski et al., 2009). We expect that parallel experiments querying gene requirements under the same conditions in both serovars examined in this study will yield further insights in to the differences in the infective process between *Typhi* and *Typhimurium*, and ultimately the pathways that underlie host-adaptation.

Both serovars possess substantial complements of horizontally-acquired DNA. We have been able to use TraDIS to assay these recently acquired sequences. In particular, weve been able to identify, on a chromosome wide scale, active prophage through the requirement for their repressors. The P4 phage utilizes an RNA-based system to make decisions regarding cell fate, and structurally similar systems are used by P1, P7, and N15 phage (Citron and Schuster, 1990; Ravin, Svarchevsky, and Deho, 1999). C4-like transcripts have been regarded as the primary repressor of lytic functions, though the IsrK-like sequence is known to be essential to the establishment of lysogeny in P4 and is transcribed in at least two phage types (Sabbattini et al., 1995; Ravin, Svarchevsky, and Deho, 1999). Our observations in *S. Typhi* suggest an important role for the IsrK-like sequence in maintenance of the lysogenic state in P4-like phage, though the mechanism remains unclear.

Recent advances in high-throughput sequencing have greatly enhanced our ability to detect novel transcripts, such as ncRNAs and short open reading frames (sORFs). In fact, our ability to identify these transcripts now far out-strips our ability to experimentally characterize these sequences. There have been previous efforts at high-throughput characterization of bacterial sRNAs and sORFs in enteric bacteria; however, these have relied on labor-intensive directed knockout libraries (Santiviago, Reynolds, et al., 2009; Hobbs, Astarita, and Storz, 2010). Here we have demonstrated that TraDIS has sufficient resolution to reliably query genomic regions as short as 60 bases, in agreement with a recent high-throughput transposon mutagenesis study in the α -proteobacteria *Caulobacter crescentus* (Christen et al., 2011). Our method has the major advantage that library construction does not rely upon genome annotation, and newly discovered elements can be surveyed with no further laboratory work.

We have been able to assign putative functions to a number of ncRNAs using TraDIS through consideration of their genomic and experimental context. In addition, ncRNA characterization generally is done in model organisms like *E. coli* or *S. Typhimurium*, and it is unclear how stable ncRNA regulatory networks are over evolutionary time. By assaying two serovars of *Salmonella* with the same method under the same conditions, we have seen hints that there may be differences in sRNA regulatory networks between *S. Typhi* and *S. Typhimurium*. In particular, we have found that under the same experimental conditions, *S. Typhi* appears to rely on the σ^E stress response pathway while *S. Typhimurium* does not; it is tempting to speculate that this difference in stress response is mediated by the observed difference in requirement for two sRNAs, RybB and RseX. We believe that this combination of high-throughput transposon mutagenesis with a careful consideration of the systems context of individual genes provides a powerful tool for the generation of functional hypotheses. We anticipate that the construction of TraDIS libraries in additional organisms, as well as the passing of these libraries through relevant experimental conditions, will provide further insights into the function of bacterial ncRNAs in addition to the protein-coding gene complement.

Published Works

Publications arising in the course of this thesis:

- Reuter S., Conner T.R., Barquist L., Walker D., Feltwell T., Harris S.R., Fookes M., Hall M.E., Fuchs T.M., Corander J., Dufour M., Ringwood T., Savin C., Bouchier C., Martin L., Miettinen M., Shubin M., Laukkonen-Ninios R., Sihvonen L.M., Siitonen A., Skurnik M., Falcão J.P., Fukushima H., Scholz H.C., Prentice M., Wren B.W., Parkhill J., Carniel E., Achtman M., McNally A., Thomson N.R. **Parallel independent evolution of pathogenicity within the genus *Yersinia*.** Manuscript under review, 2013.
- Croucher N.J., Mitchell A.M., Gould K.A., Inverarity D., Barquist L., Feltwell T., Fookes M.C., Harris S.R., Dordel J., Salter S.J., Browall S., Zemlickova H., Parkhill J., Normark S., Henriques-Normark B., Hinds J., Mitchell T.J., Bentley S.D. **Within-patient diversification and genomic stasis within a pneumococcal lineage.** Manuscript under review, 2013.
- Martin M.J., Clare S., Goulding D., Faulds-Pain A., Barquist L., Browne H., Pettit L., Lawley T.D., Dougan G., Wren B.W. **The *agr* locus regulates virulence and colonization genes in *Clostridium difficile* 027.** *Journal of Bacteriology*, 2013.
- Barquist L., Boinett C.J., Cain A.K.. **Approaches to querying bacterial genomes with transposon-insertion sequencing.** *RNA Biology*, 10(7), 2013.

- Barquist L., Langridge G.C., Turner D.J., Phan M.D., Turner A.K., Bateman A., Parkhill J., Wain J., Gardner P.P. **A comparison of dense transposon insertion libraries in the *Salmonella* serovars Typhi and Typhimurium.** *Nucleic Acids Research*, 41(8):4549-4564, 2013.
- Burge S.W., Daub J., Eberhardt R., Tate J., Barquist L., Nawrocki E.P., Eddy S.R., Gardner P.P., Bateman A. **Rfam 11.0: 10 years of RNA families.** *Nucleic Acids Research*. 41(D1):D226-D232, 2013
- Croucher N.J., Harris S.R., Barquist L., Parkhill J., Bentley S.D. **A high-resolution view of genome-wide pneumococcal transformation.** *PLoS Pathogens*, 8(6), 2012
- Westesson O., Barquist L., Holmes I. **HandAlign: Bayesian multiple sequence alignment, phylogeny, and ancestral reconstruction.** *Bioinformatics*, 28(8):1170-1171, 2012
- Gardner P.P., Barquist L., Bateman A., Nawrocki E.P., Weinberg Z. **RNIE: genome-wide prediction of bacterial intrinsic terminators.** *Nucleic Acids Research*, 39(14):5845-5852, 2011

References

- Abrahams, G. L. and M. Hensel (2006). “Manipulating cellular transport and immune responses: dynamic interactions between intracellular *Salmonella enterica* and its host cells”. *Cell Microbiol* 8.5 (May 2006), pp. 728–37. DOI: [10.1111/j.1462-5822.2006.00706.x](https://doi.org/10.1111/j.1462-5822.2006.00706.x) (see p. 21).
- AbuOun, M., P. F. Suthers, G. I. Jones, B. R. Carter, M. P. Saunders, C. D. Maranas, M. J. Woodward, and M. F. Anjum (2009). “Genome scale reconstruction of a *Salmonella* metabolic model: comparison of similarity and differences with a commensal *Escherichia coli* strain”. *J Biol Chem* 284.43 (Oct. 2009), pp. 29480–8. DOI: [10.1074/jbc.M109.005868](https://doi.org/10.1074/jbc.M109.005868) (see p. 20).
- Adey, A., H. G. Morrison, Asan, X. Xun, J. O. Kitzman, E. H. Turner, B. Stackhouse, A. P. MacKenzie, N. C. Caruccio, X. Zhang, and J. Shendure (2010). “Rapid, low-input, low-bias construction of shotgun fragment libraries by high-density in vitro transposition”. *Genome biology* 11.12 (2010), R119 (see p. 5).
- Akerley, B. J., E. J. Rubin, V. L. Novick, K. Amaya, N. Judson, and J. J. Mekalanos (2002). “A genome-scale analysis for identification of genes required for growth or survival of *Haemophilus influenzae*”. *Proc Natl Acad Sci U S A* 99.2 (Jan. 2002), pp. 966–71. DOI: [10.1073/pnas.012602299](https://doi.org/10.1073/pnas.012602299) (see p. 10).
- Andersson, S. G., A. Zomorodipour, J. O. Andersson, T. Sicheritz-Pontén, U. C. Alsmark, R. M. Podowski, A. K. Näslund, A. S. Eriksson, H. H. Winkler, and C. G. Kurland (1998). “The genome sequence of *Rickettsia prowazekii* and the origin of mitochondria”. *Nature* 396.6707 (Nov. 1998), pp. 133–40. DOI: [10.1038/24094](https://doi.org/10.1038/24094) (see p. 23).
- Antal, M., V. Bordeau, V. Douchin, and B. Felden (2005). “A small bacterial RNA regulates a putative ABC transporter”. *The Journal of biological chemistry* 280.9 (2005), pp. 7901–8 (see p. 48).
- Arnvig, K. and D. Young (2012). “Non-coding RNA and its potential role in *Mycobacterium tuberculosis* pathogenesis”. *RNA biology* 9.4 (2012), pp. 427–36 (see p. 15).
- Aseev, L. V., A. A. Levandovskaya, L. S. Tchufistova, N. V. Scaptsova, and I. V. Boni (2008). “A new regulatory circuit in ribosomal protein operons: S2-mediated

- control of the rpsB-tsf expression in vivo”. *RNA* 14.9 (2008), pp. 1882–94 (see p. 48).
- Baba, T., T. Ara, M. Hasegawa, Y. Takai, Y. Okumura, M. Baba, K. A. Datsenko, M. Tomita, B. L. Wanner, and H. Mori (2006). “Construction of Escherichia coli K-12 in-frame, single-gene knockout mutants: the Keio collection”. *Molecular systems biology* 2 (2006), p. 2006 0008 (see pp. 1, 10–12, 32, 49).
- Barquist, L., C. J. Boinett, and A. K. Cain (2013). “Approaches to querying bacterial genomes with high-throughput transposon insertion sequencing”. *RNA Biology* (2013) (see pp. 1, 25).
- Barquist, L., G. C. Langridge, D. J. Turner, M. D. Phan, A. K. Turner, A. Bateman, J. Parkhill, J. Wain, and P. P. Gardner (2013). “A comparison of dense transposon insertion libraries in the *Salmonella* serovars Typhi and Typhimurium”. *Nucleic acids research* (2013) (see pp. 3, 5, 12, 18, 19).
- Bäumler, A. J. (1997). “The record of horizontal gene transfer in *Salmonella*”. *Trends Microbiol* 5.8 (Aug. 1997), pp. 318–22. DOI: [10.1016/S0966-842X\(97\)01082-2](https://doi.org/10.1016/S0966-842X(97)01082-2) (see pp. 20, 22).
- Bäumler, A. J., R. M. Tsolis, T. A. Ficht, and L. G. Adams (1998). “Evolution of host adaptation in *Salmonella enterica*”. *Infect Immun* 66.10 (Oct. 1998), pp. 4579–87 (see p. 22).
- Bauwens, L., F. Vercammen, S. Bertrand, J.-M. Collard, and S. De Ceuster (2006). “Isolation of *Salmonella* from environmental samples collected in the reptile department of Antwerp Zoo using different selective methods”. *J Appl Microbiol* 101.2 (Aug. 2006), pp. 284–9. DOI: [10.1111/j.1365-2672.2006.02977.x](https://doi.org/10.1111/j.1365-2672.2006.02977.x) (see p. 20).
- Benard, L., C. Philippe, B. Ehresmann, C. Ehresmann, and C. Portier (1996). “Pseudoknot and translational control in the expression of the S15 ribosomal protein”. *Biochimie* 78.7 (1996), pp. 568–76 (see p. 48).
- Bentley, D. R. et al. (2008). “Accurate whole human genome sequencing using reversible terminator chemistry”. *Nature* 456.7218 (Nov. 2008), pp. 53–9. DOI: [10.1038/nature07517](https://doi.org/10.1038/nature07517) (see p. 8).
- Berardinis, V. de et al. (2008). “A complete collection of single-gene deletion mutants of *Acinetobacter baylyi* ADP1”. *Mol Syst Biol* 4 (2008), p. 174. DOI: [10.1038/msb.2008.10](https://doi.org/10.1038/msb.2008.10) (see p. 10).
- Bhutta, Z. A. and J. Threlfall (2009). “Addressing the global disease burden of typhoid fever”. *JAMA : the journal of the American Medical Association* 302.8 (2009), pp. 898–9 (see p. 23).
- Bochner, B. R. (2009). “Global phenotypic characterization of bacteria”. *FEMS microbiology reviews* 33.1 (2009), pp. 191–205 (see p. 18).

References

- Bossi, L. and N. Figueroa-Bossi (2007). “A small RNA downregulates LamB maltoporin in *Salmonella*”. *Molecular microbiology* 65.3 (2007), pp. 799–810 (see p. 51).
- Briani, F., G. Deho, F. Forti, and D. Ghisotti (2001). “The plasmid status of satellite bacteriophage P4”. *Plasmid* 45.1 (2001), pp. 1–17 (see p. 36).
- Brutinel, E. D. and J. A. Gralnick (2012). “Preferential utilization of D-lactate by *Shewanella oneidensis*”. *Applied and environmental microbiology* 78.23 (2012), pp. 8474–6 (see p. 3).
- Burge, S. W., J. Daub, R. Eberhardt, J. Tate, L. Barquist, E. P. Nawrocki, S. R. Eddy, P. P. Gardner, and A. Bateman (2013). “Rfam 11.0: 10 years of RNA families”. *Nucleic acids research* 41.D1 (2013), pp. D226–32 (see p. 26).
- Cain, A. K. and R. M. Hall (2012). “Evolution of a multiple antibiotic resistance region in IncHI1 plasmids: reshaping resistance regions in situ”. *The Journal of antimicrobial chemotherapy* 67.12 (2012), pp. 2848–53 (see p. 24).
- CDC (2009). “*Salmonella* surveillance: Annual summary”. (2009) (see p. 24).
- Chao, Y., K. Papenfort, R. Reinhart, C. M. Sharma, and J. Vogel (2012). “An atlas of Hfq-bound transcripts reveals 3’ UTRs as a genomic reservoir of regulatory small RNAs”. *The EMBO journal* 31.20 (2012), pp. 4005–19 (see pp. 48, 49).
- Chen, S., E. A. Lesnik, T. A. Hall, R. Sampath, R. H. Griffey, D. J. Ecker, and L. B. Blyn (2002). “A bioinformatics based approach to discover small RNA genes in the *Escherichia coli* genome”. *Bio Systems* 65.2-3 (2002), pp. 157–77 (see p. 48).
- Chinni, S. V., C. A. Raabe, R. Zakaria, G. Randau, C. H. Hoe, A. Zemann, J. Brosius, T. H. Tang, and T. S. Rozhdestvensky (2010). “Experimental identification and characterization of 97 novel ncRNA candidates in *Salmonella enterica* serovar Typhi”. *Nucleic acids research* 38.17 (2010), pp. 5893–908 (see pp. 26, 48, 49).
- Christen, B., E. Abeliuk, J. M. Collier, V. S. Kalogeraki, B. Passarelli, J. A. Coller, M. J. Fero, H. H. McAdams, and L. Shapiro (2011). “The essential genome of a bacterium”. *Molecular systems biology* 7 (2011), p. 528 (see pp. 3, 5, 12, 14, 25, 33, 53).
- Citron, M. and H. Schuster (1990). “The c4 repressors of bacteriophages P1 and P7 are antisense RNAs”. *Cell* 62.3 (1990), pp. 591–8 (see p. 52).
- Cole, S. T. et al. (2001). “Massive gene decay in the leprosy bacillus”. *Nature* 409.6823 (Feb. 2001), pp. 1007–11. DOI: 10.1038/35059006 (see p. 23).
- Coornaert, A., A. Lu, P. Mandin, M. Springer, S. Gottesman, and M. Guillier (2010). “MicA sRNA links the PhoP regulon to cell envelope stress”. *Molecular microbiology* 76.2 (2010), pp. 467–79 (see p. 51).
- Crick, F. H. (1966). “The genetic code. 3”. *Scientific American* 215.4 (1966), 55–60 passim (see p. 45).

- Cromie, M. J., Y. Shi, T. Latifi, and E. A. Groisman (2006). “An RNA sensor for intracellular Mg(2+)”. *Cell* 125.1 (2006), pp. 71–84 (see p. 25).
- Crump, J. A., S. P. Luby, and E. D. Mintz (2004). “The global burden of typhoid fever”. *Bulletin of the World Health Organization* 82.5 (2004), pp. 346–53 (see p. 23).
- Darwin, K. H. and V. L. Miller (1999). “InvF is required for expression of genes encoding proteins secreted by the SPI1 type III secretion apparatus in *Salmonella typhimurium*”. *Journal of bacteriology* 181.16 (1999), pp. 4949–54 (see p. 24).
- Datsenko, K. A. and B. L. Wanner (2000). “One-step inactivation of chromosomal genes in *Escherichia coli* K-12 using PCR products”. *Proceedings of the National Academy of Sciences of the United States of America* 97.12 (2000), pp. 6640–5 (see p. 11).
- Datta, N. (1962). “Transmissible drug resistance in an epidemic strain of *Salmonella typhimurium*”. *The Journal of hygiene* 60 (1962), pp. 301–10 (see p. 24).
- De Las Penas, A., L. Connolly, and C. A. Gross (1997). “SigmaE is an essential sigma factor in *Escherichia coli*”. *Journal of bacteriology* 179.21 (1997), pp. 6862–4 (see p. 49).
- DeJesus, M., Y. J. Zhang, C. M. Sassetti, E. J. Rubin, J. C. Sacchettini, and T. R. Ioerger (2013). “Bayesian Analysis of Gene Essentiality based on Sequencing of Transposon Insertion Libraries”. *Bioinformatics* (2013) (see p. 12).
- Deng, W., S. R. Liou, 3. Plunkett G., G. F. Mayhew, D. J. Rose, V. Burland, V. Kodoyianni, D. C. Schwartz, and F. R. Blattner (2003). “Comparative genomics of *Salmonella enterica* serovar Typhi strains Ty2 and CT18”. *Journal of bacteriology* 185.7 (2003), pp. 2330–7 (see p. 24).
- Dillon, S. C., A. D. Cameron, K. Hokamp, S. Lucchini, J. C. Hinton, and C. J. Dorman (2010). “Genome-wide analysis of the H-NS and Sfh regulatory networks in *Salmonella Typhimurium* identifies a plasmid-encoded transcription silencing mechanism”. *Molecular microbiology* 76.5 (2010), pp. 1250–65 (see p. 42).
- Diwa, A., A. L. Bricker, C. Jain, and J. G. Belasco (2000). “An evolutionarily conserved RNA stem-loop functions as a sensor that directs feedback regulation of RNase E gene expression”. *Genes & development* 14.10 (2000), pp. 1249–60 (see p. 48).
- Doolittle, R. F., D. F. Feng, S. Tsang, G. Cho, and E. Little (1996). “Determining divergence times of the major kingdoms of living organisms with a protein clock”. *Science* 271.5248 (Jan. 1996), pp. 470–7 (see p. 20).
- Douchin, V., C. Bohn, and P. Bouloc (2006). “Down-regulation of porins by a small RNA bypasses the essentiality of the regulated intramembrane proteolysis protease RseP in *Escherichia coli*”. *The Journal of biological chemistry* 281.18 (2006), pp. 12253–9 (see p. 48).

References

- Doyle, M., M. Fookes, A. Ivens, M. W. Mangan, J. Wain, and C. J. Dorman (2007). “An H-NS-like stealth protein aids horizontal DNA transmission in bacteria”. *Science* 315.5809 (2007), pp. 251–2 (see p. 42).
- Dziva, F., P. M. van Diemen, M. P. Stevens, A. J. Smith, and T. S. Wallis (2004). “Identification of Escherichia coli O157 : H7 genes influencing colonization of the bovine gastrointestinal tract using signature-tagged mutagenesis”. *Microbiology* 150.Pt 11 (2004), pp. 3631–45 (see p. 9).
- Echols, H. (1971). “Lysogeny: viral repression and site-specific recombination”. *Annual review of biochemistry* 40 (1971), pp. 827–54 (see p. 34).
- Eckert, S. E., F. Dziva, R. R. Chaudhuri, G. C. Langridge, D. J. Turner, D. J. Pickard, D. J. Maskell, N. R. Thomson, and M. P. Stevens (2011). “Retrospective application of transposon-directed insertion site sequencing to a library of signature-tagged mini-Tn5Km2 mutants of Escherichia coli O157:H7 screened in cattle”. *Journal of bacteriology* 193.7 (2011), pp. 1771–6 (see pp. 3, 9).
- Edwards, M. F. and B. A. Stocker (1988). “Construction of delta aroA his delta pur strains of Salmonella typhi”. *Journal of bacteriology* 170.9 (1988), pp. 3991–5 (see p. 43).
- Feasey, N. A., G. Dougan, R. A. Kingsley, R. S. Heyderman, and M. A. Gordon (2012). “Invasive non-typhoidal salmonella disease: an emerging and neglected tropical disease in Africa”. *Lancet* 379.9835 (June 2012), pp. 2489–99. DOI: 10.1016/S0140-6736(11)61752-2 (see p. 23).
- Fedurco, M., A. Romieu, S. Williams, I. Lawrence, and G. Turcatti (2006). “BTA, a novel reagent for DNA attachment on glass and efficient generation of solid-phase amplified DNA colonies”. *Nucleic Acids Res* 34.3 (2006), e22. DOI: 10.1093/nar/gnj023 (see p. 8).
- Finn, R. D., J. Clements, and S. R. Eddy (2011). “HMMER web server: interactive sequence similarity searching”. *Nucleic acids research* 39.Web Server issue (2011), W29–37 (see pp. 26, 35, 36).
- Fookes, M. et al. (2011). “Salmonella bongori provides insights into the evolution of the Salmonellae”. *PLoS Pathog* 7.8 (Aug. 2011), e1002191. DOI: 10.1371/journal.ppat.1002191 (see p. 21).
- Forti, F., I. Dragoni, F. Briani, G. Deho, and D. Ghisotti (2002). “Characterization of the small antisense CI RNA that regulates bacteriophage P4 immunity”. *Journal of molecular biology* 315.4 (2002), pp. 541–9 (see p. 36).
- Frank, D. N. and N. R. Pace (1998). “Ribonuclease P: unity and diversity in a tRNA processing ribozyme”. *Annual review of biochemistry* 67 (1998), pp. 153–80 (see p. 48).
- Fraser, C. M. et al. (2001). “Massive gene decay in the leprosy bacillus”. *Nature* 409.6823 (Feb. 2001), pp. 1007–11. DOI: 10.1038/35059006 (see p. 23).

- Freeman, J. A., M. E. Ohl, and S. I. Miller (2003). “The *Salmonella enterica* serovar *typhimurium* translocated effectors SseJ and SifB are targeted to the *Salmonella*-containing vacuole”. *Infection and immunity* 71.1 (2003), pp. 418–27 (see pp. 41, 42).
- Gallagher, L. A., J. Shendure, and C. Manoil (2011). “Genome-scale identification of resistance functions in *Pseudomonas aeruginosa* using Tn-seq”. *mBio* 2.1 (2011), e00315–10 (see pp. 3, 4, 6–8, 13, 52).
- Gawronski, J. D., S. M. Wong, G. Giannoukos, D. V. Ward, and B. J. Akerley (2009). “Tracking insertion mutants within libraries by deep sequencing and a genome-wide screen for *Haemophilus* genes required in the lung”. *Proceedings of the National Academy of Sciences of the United States of America* 106.38 (2009), pp. 16422–7 (see pp. 3–6, 13, 52).
- Gerdes, S. Y. et al. (2003). “Experimental determination and system level analysis of essential genes in *Escherichia coli* MG1655”. *Journal of bacteriology* 185.19 (2003), pp. 5673–84 (see p. 33).
- Gibson, D. G. et al. (2010). “Creation of a bacterial cell controlled by a chemically synthesized genome”. *Science* 329.5987 (July 2010), pp. 52–6. DOI: 10.1126/science.1190719 (see p. 11).
- Glass, J. I., N. Assad-Garcia, N. Alperovich, S. Yooséph, M. R. Lewis, M. Maruf, S. Hutchison C. A., H. O. Smith, and J. C. Venter (2006). “Essential genes of a minimal bacterium”. *Proceedings of the National Academy of Sciences of the United States of America* 103.2 (2006), pp. 425–30 (see p. 11).
- Goodman, A. L., N. P. McNulty, Y. Zhao, D. Leip, R. D. Mitra, C. A. Lozupone, R. Knight, and J. I. Gordon (2009). “Identifying genetic determinants needed to establish a human gut symbiont in its habitat”. *Cell host & microbe* 6.3 (2009), pp. 279–89 (see pp. 3, 4, 6, 8, 11–13, 17).
- Goodman, A. L., M. Wu, and J. I. Gordon (2011). “Identifying microbial fitness determinants by insertion sequencing using genome-wide transposon mutant libraries”. *Nature protocols* 6.12 (2011), pp. 1969–80 (see pp. 4, 6).
- Goryshin, I. Y. and W. S. Reznikoff (1998). “Tn5 in vitro transposition”. *The Journal of biological chemistry* 273.13 (1998), pp. 7367–74 (see p. 5).
- Green, B., C. Bouchier, C. Fairhead, N. L. Craig, and B. P. Cormack (2012). “Insertion site preference of Mu, Tn5, and Tn7 transposons”. *Mobile DNA* 3.1 (2012), p. 3 (see p. 5).
- Griffin, J. E., J. D. Gawronski, M. A. Dejesus, T. R. Ioerger, B. J. Akerley, and C. M. Sassetti (2011). “High-resolution phenotypic profiling defines genes essential for mycobacterial growth and cholesterol catabolism”. *PLoS pathogens* 7.9 (2011), e1002251 (see pp. 3, 9, 11–13, 52).

References

- Grimont, P. A. and F.-X. Weill (2007). “Antigenic formulae of the *Salmonella* serovars”. *WHO Collaborating Centre for Reference and Research on Salmonella, Institut Pasteur, Paris, France* (2007) (see p. 21).
- Gulig, P. A. and J. Curtiss R. (1987). “Plasmid-associated virulence of *Salmonella typhimurium*”. *Infection and immunity* 55.12 (1987), pp. 2891–901 (see p. 24).
- Gulig, P. A., H. Danbara, D. G. Guiney, A. J. Lax, F. Norel, and M. Rhen (1993). “Molecular analysis of spv virulence genes of the *Salmonella* virulence plasmids”. *Molecular microbiology* 7.6 (1993), pp. 825–30 (see p. 24).
- Hashimoto, M., T. Ichimura, H. Mizoguchi, K. Tanaka, K. Fujimitsu, K. Keyamura, T. Ote, T. Yamakawa, Y. Yamazaki, H. Mori, T. Katayama, and J. Kato (2005). “Cell size and nucleoid organization of engineered *Escherichia coli* cells with a reduced genome”. *Molecular microbiology* 55.1 (2005), pp. 137–49 (see p. 33).
- Hebrard, M., C. Kroger, S. Srikumar, A. Colgan, K. Handler, and J. Hinton (2012). “sRNAs and the virulence of *Salmonella enterica* serovar Typhimurium”. *RNA biology* 9.4 (2012) (see p. 25).
- Hensel, M., J. E. Shea, A. J. Baumler, C. Gleeson, F. Blattner, and D. W. Holden (1997). “Analysis of the boundaries of *Salmonella* pathogenicity island 2 and the corresponding chromosomal region of *Escherichia coli* K-12”. *Journal of bacteriology* 179.4 (1997), pp. 1105–11 (see p. 42).
- Hensel, M., J. E. Shea, C. Gleeson, M. D. Jones, E. Dalton, and D. W. Holden (1995). “Simultaneous identification of bacterial virulence genes by negative selection”. *Science* 269.5222 (1995), pp. 400–3 (see p. 1).
- Hensel, M., J. E. Shea, S. R. Waterman, R. Mundy, T. Nikolaus, G. Banks, A. Vazquez-Torres, C. Gleeson, F. C. Fang, and D. W. Holden (1998). “Genes encoding putative effector proteins of the type III secretion system of *Salmonella* pathogenicity island 2 are required for bacterial virulence and proliferation in macrophages”. *Molecular microbiology* 30.1 (1998), pp. 163–74 (see p. 42).
- Hensel, M. (2004). “Evolution of pathogenicity islands of *Salmonella enterica*”. *Int J Med Microbiol* 294.2-3 (Sept. 2004), pp. 95–102. doi: 10.1016/j.ijmm.2004.06.025 (see p. 20).
- Hilbert, F., F. Smulders, R. Chopra-Dewasthaly, and P. Paulsen (2012). “*Salmonellai/i_c* in the wildlife-human interface”. *Food Research International* 45.2 (2012), pp. 603–608 (see p. 22).
- Hobbs, E. C., J. L. Astarita, and G. Storz (2010). “Small RNAs and small proteins involved in resistance to cell envelope stress and acid shock in *Escherichia coli*: analysis of a bar-coded mutant collection”. *Journal of bacteriology* 192.1 (2010), pp. 59–67 (see pp. 1, 53).
- Holt, K. E., J. Parkhill, C. J. Mazzoni, P. Roumagnac, F. X. Weill, I. Goodhead, R. Rance, S. Baker, D. J. Maskell, J. Wain, C. Dolecek, M. Achtman, and G. Dougan (2008). “High-throughput sequencing provides insights into genome

- variation and evolution in *Salmonella Typhi*”. *Nature genetics* 40.8 (2008), pp. 987–93 (see p. 24).
- Holt, K. E., Y. Y. Teo, H. Li, S. Nair, G. Dougan, J. Wain, and J. Parkhill (2009). “Detecting SNPs and estimating allele frequencies in clonal bacterial populations by sequencing pooled DNA”. *Bioinformatics* 25.16 (2009), pp. 2074–5 (see pp. 23, 24, 34).
- Holt, K. E., N. R. Thomson, J. Wain, M. D. Phan, S. Nair, R. Hasan, Z. A. Bhutta, M. A. Quail, H. Norbertczak, D. Walker, G. Dougan, and J. Parkhill (2007). “Multidrug-resistant *Salmonella enterica* serovar paratyphi A harbors IncHI1 plasmids similar to those found in serovar typhi”. *Journal of bacteriology* 189.11 (2007), pp. 4257–64 (see p. 24).
- Hutchison, C. A., S. N. Peterson, S. R. Gill, R. T. Cline, O. White, C. M. Fraser, H. O. Smith, and J. C. Venter (1999). “Global transposon mutagenesis and a minimal Mycoplasma genome”. *Science* 286.5447 (1999), pp. 2165–9 (see pp. 2, 3, 11).
- Iyer, L. M., M. M. Babu, and L. Aravind (2006). “The HIRAN domain and recruitment of chromatin remodeling and repair activities to damaged DNA”. *Cell cycle* 5.7 (2006), pp. 775–82 (see p. 37).
- Jacobs, M. A., A. Alwood, I. Thaipisuttikul, D. Spencer, E. Haugen, S. Ernst, O. Will, R. Kaul, C. Raymond, R. Levy, L. Chun-Rong, D. Guenthner, D. Bovee, M. V. Olson, and C. Manoil (2003). “Comprehensive transposon mutant library of *Pseudomonas aeruginosa*”. *Proceedings of the National Academy of Sciences of the United States of America* 100.24 (2003), pp. 14339–44 (see pp. 10–12).
- Johnsborg, O., V. Eldholm, and L. S. Havarstein (2007). “Natural genetic transformation: prevalence, mechanisms and function”. *Research in microbiology* 158.10 (2007), pp. 767–78 (see p. 5).
- Jones, A. M., A. Goodwill, and T. Elliott (2006). “Limited role for the DsrA and RprA regulatory RNAs in rpoS regulation in *Salmonella enterica*”. *Journal of bacteriology* 188.14 (2006), pp. 5077–88 (see p. 51).
- Kang, Y., T. Durfee, J. D. Glasner, Y. Qiu, D. Frisch, K. M. Winterberg, and F. R. Blattner (2004). “Systematic mutagenesis of the *Escherichia coli* genome”. *Journal of bacteriology* 186.15 (2004), pp. 4921–30 (see p. 33).
- Kaper, J. B., J. P. Nataro, and H. L. Mobley (2004). “Pathogenic *Escherichia coli*”. *Nat Rev Microbiol* 2.2 (Feb. 2004), pp. 123–40. DOI: 10.1038/nrmicro818 (see p. 20).
- Kawano, M., S. Kanaya, T. Oshima, Y. Masuda, T. Ara, and H. Mori (2002). “Distribution of repetitive sequences on the leading and lagging strands of the *Escherichia coli* genome: comparative study of Long Direct Repeat (LDR) sequences”. *DNA research : an international journal for rapid publication of reports on genes and genomes* 9.1 (2002), pp. 1–10 (see p. 49).

References

- Khatiwara, A., T. Jiang, S. S. Sung, T. Dawoud, J. N. Kim, D. Bhattacharya, H. B. Kim, S. C. Ricke, and Y. M. Kwon (2012). “Genome scanning for conditionally essential genes in *Salmonella enterica* Serotype Typhimurium”. *Applied and environmental microbiology* 78.9 (2012), pp. 3098–107 (see pp. 3, 13).
- Kidgell, C., U. Reichard, J. Wain, B. Linz, M. Torpdahl, G. Dougan, and M. Achtman (2002). “*Salmonella typhi*, the causative agent of typhoid fever, is approximately 50,000 years old”. *Infect Genet Evol* 2.1 (Oct. 2002), pp. 39–45 (see p. 23).
- Kingsley, R. A., A. D. Humphries, E. H. Weening, M. R. De Zoete, S. Winter, A. Papaconstantinopoulou, G. Dougan, and A. J. Baumler (2003). “Molecular and phenotypic analysis of the CS54 island of *Salmonella enterica* serotype typhimurium: identification of intestinal colonization and persistence determinants”. *Infection and immunity* 71.2 (2003), pp. 629–40 (see p. 25).
- Klein, B. A., E. L. Tenorio, D. W. Lazinski, A. Camilli, M. J. Duncan, and L. T. Hu (2012). “Identification of essential genes of the periodontal pathogen *Porphyromonas gingivalis*”. *BMC genomics* 13 (2012), p. 578 (see p. 3).
- Knuth, K., H. Niesalla, C. J. Hueck, and T. M. Fuchs (2004). “Large-scale identification of essential *Salmonella* genes by trapping lethal insertions”. *Molecular microbiology* 51.6 (2004), pp. 1729–44 (see pp. 10, 30).
- Kobayashi, K. et al. (2003). “Essential *Bacillus subtilis* genes”. *Proc Natl Acad Sci U S A* 100.8 (Apr. 2003), pp. 4678–83. DOI: [10.1073/pnas.0730515100](https://doi.org/10.1073/pnas.0730515100) (see p. 10).
- Koonin, E. V. (2003). “Comparative genomics, minimal gene-sets and the last universal common ancestor”. *Nat Rev Microbiol* 1.2 (Nov. 2003), pp. 127–36. DOI: [10.1038/nrmicro751](https://doi.org/10.1038/nrmicro751) (see p. 10).
- Kothari, A., A. Pruthi, and T. D. Chugh (2008). “The burden of enteric fever”. *Journal of infection in developing countries* 2.4 (2008), pp. 253–9 (see p. 23).
- Kröger, C. et al. (2012). “The transcriptional landscape and small RNAs of *Salmonella enterica* serovar Typhimurium”. *Proceedings of the National Academy of Sciences of the United States of America* 109.20 (2012), E1277–86 (see pp. 25, 26, 49).
- Kropinski, A. M., A. Sulakvelidze, P. Koncza, and C. Poppe (2007). “*Salmonella* phages and prophages—genomics and practical aspects”. *Methods in molecular biology* 394 (2007), pp. 133–75 (see pp. 35, 36).
- Kuhle, V. and M. Hensel (2004). “Cellular microbiology of intracellular *Salmonella enterica*: functions of the type III secretion system encoded by *Salmonella* pathogenicity island 2”. *Cell Mol Life Sci* 61.22 (Nov. 2004), pp. 2812–26. DOI: [10.1007/s00018-004-4248-z](https://doi.org/10.1007/s00018-004-4248-z) (see pp. 21, 41).

- Lampe, D. J., M. E. Churchill, and H. M. Robertson (1996). “A purified mariner transposase is sufficient to mediate transposition in vitro”. *The EMBO journal* 15.19 (1996), pp. 5470–9 (see p. 5).
- Lampe, D. J., T. E. Grant, and H. M. Robertson (1998). “Factors affecting transposition of the Himar1 mariner transposon in vitro”. *Genetics* 149.1 (1998), pp. 179–87 (see p. 5).
- Landt, S. G., E. Abeliuk, P. T. McGrath, J. A. Lesley, H. H. McAdams, and L. Shapiro (2008). “Small non-coding RNAs in *Caulobacter crescentus*”. *Molecular microbiology* 68.3 (2008), pp. 600–14 (see p. 15).
- Langridge, G. C. (2010). “Metabolic capability in host-restricted serovars of *Salmonella enterica*”. PhD thesis. University of Cambridge, 2010 (see pp. 19, 26).
- Langridge, G. C., M. D. Phan, D. J. Turner, T. T. Perkins, L. Parts, J. Haase, I. Charles, D. J. Maskell, S. E. Peters, G. Dougan, J. Wain, J. Parkhill, and A. K. Turner (2009). “Simultaneous assay of every *Salmonella Typhi* gene using one million transposon mutants”. *Genome research* 19.12 (2009), pp. 2308–16 (see pp. 3–6, 12, 13, 25–28, 30, 44, 52).
- Lease, R. A., M. E. Cusick, and M. Belfort (1998). “Riboregulation in *Escherichia coli*: DsrA RNA acts by RNA:RNA interactions at multiple loci”. *Proceedings of the National Academy of Sciences of the United States of America* 95.21 (1998), pp. 12456–61 (see pp. 48, 51).
- Lemire, S., N. Figueroa-Bossi, and L. Bossi (2011). “Bacteriophage crosstalk: coordination of prophage induction by trans-acting antirepressors”. *PLoS genetics* 7.6 (2011), e1002149 (see p. 35).
- Li, H., J. Ruan, and R. Durbin (2008). “Mapping short DNA sequencing reads and calling variants using mapping quality scores”. *Genome research* 18.11 (2008), pp. 1851–8 (see p. 27).
- Lowe, T. M. and S. R. Eddy (1997). “tRNAscan-SE: a program for improved detection of transfer RNA genes in genomic sequence”. *Nucleic acids research* 25.5 (1997), pp. 955–64 (see p. 45).
- Lucchini, S., G. Rowley, M. D. Goldberg, D. Hurd, M. Harrison, and J. C. Hinton (2006). “H-NS mediates the silencing of laterally acquired genes in bacteria”. *PLoS pathogens* 2.8 (2006), e81 (see pp. 12, 41).
- Majowicz, S. E., J. Musto, E. Scallan, F. J. Angulo, M. Kirk, S. J. O’Brien, T. F. Jones, A. Fazil, and R. M. Hoekstra (2010). “The global burden of nontyphoidal *Salmonella* gastroenteritis”. *Clinical infectious diseases : an official publication of the Infectious Diseases Society of America* 50.6 (2010), pp. 882–9 (see p. 24).
- Mann, B., T. van Oprijnen, J. Wang, C. Obert, Y. D. Wang, R. Carter, D. J. McGoldrick, G. Ridout, A. Camilli, E. I. Tuomanen, and J. W. Rosch (2012).

References

- “Control of virulence by small RNAs in *Streptococcus pneumoniae*”. *PLoS pathogens* 8.7 (2012), e1002788 (see pp. 3, 15, 16).
- Manna, D., S. Porwollik, M. McClelland, R. Tan, and N. P. Higgins (2007). “Microarray analysis of Mu transposition in *Salmonella enterica*, serovar Typhimurium: transposon exclusion by high-density DNA binding proteins”. *Molecular microbiology* 66.2 (2007), pp. 315–28 (see pp. 12, 41).
- Marck, C. and H. Grosjean (2002). “tRNomics: analysis of tRNA genes from 50 genomes of Eukarya, Archaea, and Bacteria reveals anticodon-sparing strategies and domain-specific features”. *RNA* 8.10 (2002), pp. 1189–232 (see p. 46).
- Mazurkiewicz, P., C. M. Tang, C. Boone, and D. W. Holden (2006). “Signature-tagged mutagenesis: barcoding mutants for genome-wide screens”. *Nature reviews. Genetics* 7.12 (2006), pp. 929–39 (see p. 2).
- McClelland, M., K. E. Sanderson, S. W. Clifton, et al. (2004). “Comparison of genome degradation in Paratyphi A and Typhi, human-restricted serovars of *Salmonella enterica* that cause typhoid”. *Nature genetics* 36.12 (2004), pp. 1268–74 (see pp. 23, 24, 34, 43).
- McClelland, M., K. E. Sanderson, J. Spieth, et al. (2001). “Complete genome sequence of *Salmonella enterica* serovar Typhimurium LT2”. *Nature* 413.6858 (2001), pp. 852–6 (see p. 24).
- Mermin, J., L. Hutwagner, D. Vugia, S. Shallow, P. Daily, J. Bender, J. Koehler, R. Marcus, F. J. Angulo, and Emerging Infections Program FoodNet Working Group (2004). “Reptiles, amphibians, and human *Salmonella* infection: a population-based, case-control study”. *Clin Infect Dis* 38 Suppl 3 (Apr. 2004), S253–61. DOI: 10.1086/381594 (see pp. 20, 22).
- Meyer, M. M., T. D. Ames, D. P. Smith, Z. Weinberg, M. S. Schwalbach, S. J. Giovannoni, and R. R. Breaker (2009). “Identification of candidate structured RNAs in the marine organism ‘Candidatus Pelagibacter ubique’”. *BMC genomics* 10 (2009), p. 268 (see p. 48).
- Miao, E. A. and S. I. Miller (2000). “A conserved amino acid sequence directing intracellular type III secretion by *Salmonella typhimurium*”. *Proceedings of the National Academy of Sciences of the United States of America* 97.13 (2000), pp. 7539–44 (see p. 41).
- Moran, N. A. (2004). “Cellular microbiology of intracellular *Salmonella enterica*: functions of the type III secretion system encoded by *Salmonella* pathogenicity island 2”. *Cell Mol Life Sci* 61.22 (Nov. 2004), pp. 2812–26. DOI: 10.1007/s00018-004-4248-z (see p. 22).
- Morozova, O. and M. A. Marra (2008). “Applications of next-generation sequencing technologies in functional genomics”. *Genomics* 92.5 (Nov. 2008), pp. 255–64. DOI: 10.1016/j.ygeno.2008.07.001 (see p. 7).

- Mushegian, A. R. and E. V. Koonin (1996). “A minimal gene set for cellular life derived by comparison of complete bacterial genomes”. *Proc Natl Acad Sci U S A* 93.19 (Sept. 1996), pp. 10268–73 (see p. 10).
- Näsvall, S. J., P. Chen, and G. R. Björk (2004). “The modified wobble nucleoside uridine-5-oxyacetic acid in tRNAPro(cmo5UGG) promotes reading of all four proline codons in vivo”. *RNA* 10.10 (2004), pp. 1662–73 (see p. 45).
- Navarre, W. W., S. Porwollik, Y. Wang, M. McClelland, H. Rosen, S. J. Libby, and F. C. Fang (2006). “Selective silencing of foreign DNA with low GC content by the H-NS protein in *Salmonella*”. *Science* 313.5784 (2006), pp. 236–8 (see pp. 12, 38, 41, 42, 51).
- Naville, M. and D. Gautheret (2010). “Premature terminator analysis sheds light on a hidden world of bacterial transcriptional attenuation”. *Genome biology* 11.9 (2010), R97 (see p. 49).
- Nichols, R. J., S. Sen, Y. J. Choo, P. Beltrao, M. Zietek, R. Chaba, S. Lee, K. M. Kazmierczak, K. J. Lee, A. Wong, M. Shales, S. Lovett, M. E. Winkler, N. J. Krogan, A. Typas, and C. A. Gross (2011). “Phenotypic landscape of a bacterial cell”. *Cell* 144.1 (2011), pp. 143–56 (see p. 18).
- Ochman, H. and E. A. Groisman (1996). “Distribution of pathogenicity islands in *Salmonella* spp”. *Infection and immunity* 64.12 (1996), pp. 5410–2 (see p. 24).
- Ochman, H. and A. C. Wilson (1987). “Evolution in bacteria: evidence for a universal substitution rate in cellular genomes”. *J Mol Evol* 26.1-2 (1987), pp. 74–86 (see p. 20).
- Ohlson, M. B., K. Fluhr, C. L. Birmingham, J. H. Brumell, and S. I. Miller (2005). “SseJ deacylase activity by *Salmonella enterica* serovar Typhimurium promotes virulence in mice”. *Infection and immunity* 73.10 (2005), pp. 6249–59 (see p. 42).
- Okoro, C. K. et al. (2012). “Intracontinental spread of human invasive *Salmonella* Typhimurium pathovariants in sub-Saharan Africa”. *Nat Genet* 44.11 (Nov. 2012), pp. 1215–21. DOI: 10.1038/ng.2423 (see p. 23).
- Opijnen, T. van, K. L. Bodi, and A. Camilli (2009). “Tn-seq: high-throughput parallel sequencing for fitness and genetic interaction studies in microorganisms”. *Nature methods* 6.10 (2009), pp. 767–72 (see pp. 3–6, 8, 11, 13).
- Opijnen, T. van and A. Camilli (2012). “A fine scale phenotype-genotype virulence map of a bacterial pathogen”. *Genome research* 22.12 (2012), pp. 2541–51 (see pp. 3, 8, 13, 14, 18).
- Osawa, S., T. H. Jukes, K. Watanabe, and A. Muto (1992). “Recent evidence for evolution of the genetic code”. *Microbiological reviews* 56.1 (1992), pp. 229–64 (see p. 46).
- Padalon-Brauch, G., R. Hershberg, M. Elgrably-Weiss, K. Baruch, I. Rosenshine, H. Margalit, and S. Altuvia (2008). “Small RNAs encoded within genetic islands

References

- of *Salmonella typhimurium* show host-induced expression and role in virulence”. *Nucleic acids research* 36.6 (2008), pp. 1913–27 (see pp. 36, 48, 49).
- Pallen, M. J. and B. W. Wren (2007). “Bacterial pathogenomics”. *Nature* 449.7164 (Oct. 2007), pp. 835–42. DOI: 10.1038/nature06248 (see p. 22).
- Papenfort, K., V. Pfeiffer, F. Mika, S. Lucchini, J. C. Hinton, and J. Vogel (2006). “SigmaE-dependent small RNAs of *Salmonella* respond to membrane stress by accelerating global *omp* mRNA decay”. *Molecular microbiology* 62.6 (2006), pp. 1674–88 (see p. 50).
- Paradis, S., M. Boissinot, N. Paquette, S. D. Belanger, E. A. Martel, D. K. Boudreau, F. J. Picard, M. Ouellette, P. H. Roy, and M. G. Bergeron (2005). “Phylogeny of the Enterobacteriaceae based on genes encoding elongation factor Tu and F-ATPase beta-subunit”. *International journal of systematic and evolutionary microbiology* 55.Pt 5 (2005), pp. 2013–25 (see p. 20).
- Parkhill, J. et al. (2001). “Complete genome sequence of a multiple drug resistant *Salmonella enterica* serovar Typhi CT18”. *Nature* 413.6858 (2001), pp. 848–52 (see pp. 23–25).
- Pham, H. N., K. Ohkusu, N. Mishima, M. Noda, M. Monir Shah, X. Sun, M. Hayashi, and T. Ezaki (2007). “Phylogeny and species identification of the family Enterobacteriaceae based on dnaJ sequences”. *Diagn Microbiol Infect Dis* 58.2 (June 2007), pp. 153–61. DOI: 10.1016/j.diagmicrobio.2006.12.019 (see p. 20).
- Phan, M. D., C. Kidgell, S. Nair, K. E. Holt, A. K. Turner, J. Hinds, P. Butcher, F. J. Cooke, N. R. Thomson, R. Titball, Z. A. Bhutta, R. Hasan, G. Dougan, and J. Wain (2009). “Variation in *Salmonella enterica* serovar typhi IncHI1 plasmids during the global spread of resistant typhoid fever”. *Antimicrobial agents and chemotherapy* 53.2 (2009), pp. 716–27 (see p. 24).
- Pickard, D., R. A. Kingsley, C. Hale, K. Turner, K. Sivaraman, M. Wetter, G. Langridge, and G. Dougan (2013). “A genome-wide mutagenesis screen identifies multiple genes contributing to Vi capsular expression in *Salmonella Typhi*”. *Journal of bacteriology* (2013) (see pp. 3, 13).
- Pickard, D., J. Wain, S. Baker, A. Line, S. Chohan, M. Fookes, A. Barron, P. O. Gaora, J. A. Chabalgoity, N. Thanky, C. Scholes, N. Thomson, M. Quail, J. Parkhill, and G. Dougan (2003). “Composition, acquisition, and distribution of the Vi exopolysaccharide-encoding *Salmonella enterica* pathogenicity island SPI-7”. *Journal of bacteriology* 185.17 (2003), pp. 5055–65 (see p. 24).
- Prosseda, G., M. L. Di Martino, R. Campilongo, R. Fioravanti, G. Micheli, M. Casalino, and B. Colonna (2012). “Shedding of genes that interfere with the pathogenic lifestyle: the *Shigella* model”. *Res Microbiol* 163.6-7 (July 2012), pp. 399–406. DOI: 10.1016/j.resmic.2012.07.004 (see pp. 20, 22).

- Punta, M., P. C. Coggill, R. Y. Eberhardt, J. Mistry, J. Tate, C. Boursnell, N. Pang, K. Forslund, G. Ceric, J. Clements, A. Heger, L. Holm, E. L. Sonnhammer, S. R. Eddy, A. Bateman, and R. D. Finn (2012). “The Pfam protein families database”. *Nucleic acids research* 40.Database issue (2012), pp. D290–301 (see pp. 26, 35, 36).
- Rabsch, W., W. Voigt, R. Reissbrodt, R. M. Tsolis, and A. J. Baumler (1999). “*Salmonella typhimurium* IroN and FepA proteins mediate uptake of enterobactin but differ in their specificity for other siderophores”. *Journal of bacteriology* 181.11 (1999), pp. 3610–2 (see p. 43).
- Raghavan, R., E. A. Groisman, and H. Ochman (2011). “Genome-wide detection of novel regulatory RNAs in *E. coli*”. *Genome research* 21.9 (2011), pp. 1487–97 (see pp. 26, 47–49).
- Ravin, N. V., A. N. Svarchevsky, and G. Deho (1999). “The anti-immunity system of phage-plasmid N15: identification of the antirepressor gene and its control by a small processed RNA”. *Molecular microbiology* 34.5 (1999), pp. 980–94 (see p. 52).
- Richter, A. S. and R. Backofen (2012). “Accessibility and conservation: General features of bacterial small RNA-mRNA interactions?”: *RNA biology* 9.7 (2012) (see p. 25).
- Rivas, E. and S. R. Eddy (2001). “Noncoding RNA gene detection using comparative sequence analysis”. *BMC bioinformatics* 2 (2001), p. 8 (see p. 49).
- Rosenblad, M. A., N. Larsen, T. Samuelsson, and C. Zwieb (2009). “Kinship in the SRP RNA family”. *RNA biology* 6.5 (2009), pp. 508–16 (see p. 48).
- Ross-Macdonald, P., P. S. Coelho, T. Roemer, S. Agarwal, A. Kumar, R. Jansen, K. H. Cheung, A. Sheehan, D. Symoniatis, L. Umansky, M. Heidtman, F. K. Nelson, H. Iwasaki, K. Hager, M. Gerstein, P. Miller, G. S. Roeder, and M. Snyder (1999). “Large-scale analysis of the yeast genome by transposon tagging and gene disruption”. *Nature* 402.6760 (1999), pp. 413–8 (see p. 18).
- Rubin, E. J., B. J. Akerley, V. N. Novik, D. J. Lampe, R. N. Husson, and J. J. Mekalanos (1999). “In vivo transposition of mariner-based elements in enteric bacteria and mycobacteria”. *Proceedings of the National Academy of Sciences of the United States of America* 96.4 (1999), pp. 1645–50 (see p. 5).
- Sabbattini, P., F. Forti, D. Ghisotti, and G. Deho (1995). “Control of transcription termination by an RNA factor in bacteriophage P4 immunity: identification of the target sites”. *Journal of bacteriology* 177.6 (1995), pp. 1425–34 (see pp. 36, 52).
- Sagan, L. (1967). “On the origin of mitosing cells”. *J Theor Biol* 14.3 (Mar. 1967), pp. 255–74 (see p. 23).

References

- Sanger, F., S. Nicklen, and A. R. Coulson (1977). “DNA sequencing with chain-terminating inhibitors”. *Proc Natl Acad Sci U S A* 74.12 (Dec. 1977), pp. 5463–7 (see p. 7).
- Santiviago, C. A., M. M. Reynolds, S. Porwollik, S. H. Choi, F. Long, H. L. Andrews-Polymenis, and M. McClelland (2009). “Analysis of pools of targeted *Salmonella* deletion mutants identifies novel genes affecting fitness during competitive infection in mice”. *PLoS pathogens* 5.7 (2009), e1000477 (see pp. 30, 53).
- Santiviago, C. A., C. S. Toro, A. A. Hidalgo, P. Youderian, and G. C. Mora (2003). “Global regulation of the *Salmonella enterica* serovar *typhimurium* major porin, OmpD”. *Journal of bacteriology* 185.19 (2003), pp. 5901–5 (see p. 51).
- Santos, R. L., S. Zhang, R. M. Tsolis, R. A. Kingsley, L. G. Adams, and A. J. Baumler (2001). “Animal models of *Salmonella* infections: enteritis versus typhoid fever”. *Microbes and infection / Institut Pasteur* 3.14-15 (2001), pp. 1335–44 (see pp. 22, 24).
- Sassetti, C. M., D. H. Boyd, and E. J. Rubin (2003). “Genes required for mycobacterial growth defined by high density mutagenesis”. *Molecular microbiology* 48.1 (2003), pp. 77–84 (see p. 9).
- Seth-Smith, H. M. (2008). “SPI-7: *Salmonella*’s Vi-encoding Pathogenicity Island”. *Journal of infection in developing countries* 2.4 (2008), pp. 267–71 (see p. 24).
- Shea, J. E., M. Hensel, C. Gleeson, and D. W. Holden (1996). “Identification of a virulence locus encoding a second type III secretion system in *Salmonella typhimurium*”. *Proceedings of the National Academy of Sciences of the United States of America* 93.6 (1996), pp. 2593–7 (see p. 24).
- Shevchenko, Y., G. G. Bouffard, Y. S. Butterfield, R. W. Blakesley, J. L. Hartley, A. C. Young, M. A. Marra, S. J. Jones, J. W. Touchman, and E. D. Green (2002). “Systematic sequencing of cDNA clones using the transposon Tn5”. *Nucleic acids research* 30.11 (2002), pp. 2469–77 (see p. 5).
- Sittka, A., S. Lucchini, K. Papenfort, C. M. Sharma, K. Rolle, T. T. Binnewies, J. C. Hinton, and J. Vogel (2008). “Deep sequencing analysis of small noncoding RNA and mRNA targets of the global post-transcriptional regulator, Hfq”. *PLoS genetics* 4.8 (2008), e1000163 (see pp. 26, 48, 49).
- Soper, G. A. (1939). “The Curious Career of Typhoid Mary”. *Bulletin of the New York Academy of Medicine* 15.10 (1939), pp. 698–712 (see p. 23).
- Thomson, N. R. et al. (2008). “Comparative genome analysis of *Salmonella Enteritidis* PT4 and *Salmonella Gallinarum* 287/91 provides insights into evolutionary and host adaptation pathways”. *Genome research* 18.10 (2008), pp. 1624–37 (see pp. 23, 24).
- Thomson, N., S. Baker, D. Pickard, M. Fookes, M. Anjum, N. Hamlin, J. Wain, D. House, Z. Bhutta, K. Chan, S. Falkow, J. Parkhill, M. Woodward, A. Ivens, and G. Dougan (2004). “The role of prophage-like elements in the diversity

- of *Salmonella enterica* serovars". *Journal of molecular biology* 339.2 (2004), pp. 279–300 (see pp. 35, 36).
- Tjaden, B., R. M. Saxena, S. Stolyar, D. R. Haynor, E. Kolker, and C. Rosenow (2002). "Transcriptome analysis of *Escherichia coli* using high-density oligonucleotide probe arrays". *Nucleic acids research* 30.17 (2002), pp. 3732–8 (see p. 48).
- Toft, C. and S. G. Andersson (2010). "Evolutionary microbial genomics: insights into bacterial host adaptation". *Nature reviews. Genetics* 11.7 (2010), pp. 465–75 (see p. 14).
- Tong, X., J. W. Campbell, G. Balazsi, K. A. Kay, B. L. Wanner, S. Y. Gerdes, and Z. N. Oltvai (2004). "Genome-scale identification of conditionally essential genes in *E. coli* by DNA microarrays". *Biochemical and biophysical research communications* 322.1 (2004), pp. 347–54 (see p. 33).
- Townsend, S. M., N. E. Kramer, R. Edwards, S. Baker, N. Hamlin, M. Simmonds, K. Stevens, S. Maloy, J. Parkhill, G. Dougan, and A. J. Baumler (2001). "*Salmonella enterica* serovar Typhi possesses a unique repertoire of fimbrial gene sequences". *Infection and immunity* 69.5 (2001), pp. 2894–901 (see p. 24).
- Tsolis, R. M., A. J. Baumler, F. Heffron, and I. Stojiljkovic (1996). "Contribution of TonB- and Feo-mediated iron uptake to growth of *Salmonella typhimurium* in the mouse". *Infection and immunity* 64.11 (1996), pp. 4549–56 (see p. 43).
- Urban, J. H. and J. Vogel (2008). "Two seemingly homologous noncoding RNAs act hierarchically to activate glmS mRNA translation". *PLoS biology* 6.3 (2008), e64 (see p. 49).
- Vogel, J. (2009a). "A rough guide to the non-coding RNA world of *Salmonella*". *Molecular microbiology* 71.1 (2009), pp. 1–11 (see pp. 25, 49, 51).
- Vogel, J. (2009b). "An RNA trap helps bacteria get the most out of chitosugars". *Molecular microbiology* 73.5 (2009), pp. 737–41 (see pp. 48, 49).
- Vogel, J., V. Bartels, T. H. Tang, G. Churakov, J. G. Slagter-Jager, A. Huttenhofer, and E. G. Wagner (2003). "RNomics in *Escherichia coli* detects new sRNA species and indicates parallel transcriptional output in bacteria". *Nucleic acids research* 31.22 (2003), pp. 6435–43 (see pp. 46, 48).
- Wain, J., T. S. Diep, V. A. Ho, A. M. Walsh, T. T. Nguyen, C. M. Parry, and N. J. White (1998). "Quantitation of bacteria in blood of typhoid fever patients and relationship between counts and clinical features, transmissibility, and antibiotic resistance". *Journal of clinical microbiology* 36.6 (1998), pp. 1683–7 (see pp. 24, 43).
- Waldminghaus, T., N. Heidrich, S. Brantl, and F. Narberhaus (2007). "FourU: a novel type of RNA thermometer in *Salmonella*". *Molecular microbiology* 65.2 (2007), pp. 413–24 (see p. 25).

References

- Wassarman, K. M., F. Repoila, C. Rosenow, G. Storz, and S. Gottesman (2001). “Identification of novel small RNAs using comparative genomics and microarrays”. *Genes & development* 15.13 (2001), pp. 1637–51 (see p. 48).
- Winkler, W., A. Nahvi, and R. R. Breaker (2002). “Thiamine derivatives bind messenger RNAs directly to regulate bacterial gene expression”. *Nature* 419.6910 (2002), pp. 952–6 (see pp. 46, 48).
- Wu, D. et al. (2009). “A phylogeny-driven genomic encyclopaedia of Bacteria and Archaea”. *Nature* 462.7276 (2009), pp. 1056–60 (see pp. 18, 20).
- Zhang, Y. J., T. R. Ioerger, C. Huttenhower, J. E. Long, C. M. Sassetti, J. C. Sacchettini, and E. J. Rubin (2012). “Global assessment of genomic regions required for growth in *Mycobacterium tuberculosis*”. *PLoS pathogens* 8.9 (2012), e1002946 (see pp. 3, 8, 9, 11, 12, 14–16).
- Zomer, A., P. Burghout, H. J. Bootsma, P. W. Hermans, and S. A. van Huijum (2012). “ESSENTIALS: software for rapid analysis of high throughput transposon insertion sequencing data”. *PloS one* 7.8 (2012), e43012 (see pp. 11, 12).
- Zurawski, G., K. Brown, D. Killingly, and C. Yanofsky (1978). “Nucleotide sequence of the leader region of the phenylalanine operon of *Escherichia coli*”. *Proceedings of the National Academy of Sciences of the United States of America* 75.9 (1978), pp. 4271–5 (see p. 49).